BASIC TECHNIQUES

for Observing and Studying

Moths & Butterflies

WILLIAM D. WINTER JR.

Memoirs of the Lepidopterists' Society No. 5

Basic Techniques for
Observing and Studying

Moths & Butterflies

STUDENT COPY

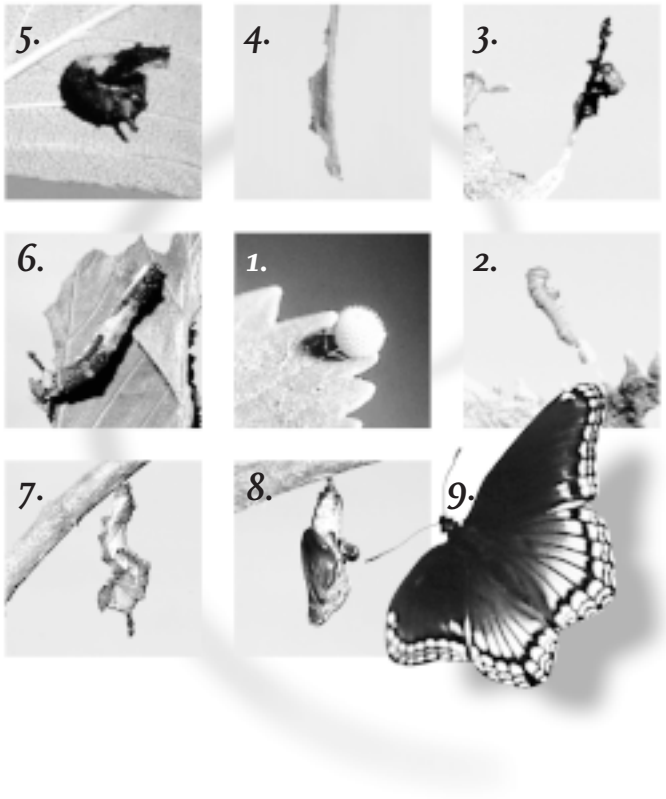
About the Cover

The cover offers a preview of a number of the subjects of the manual.

On a hawthorn tree we planted in our yard (Gardening, Chapter 5) I chanced to see (Observation, Chapter 1) a red-spotted purple, *Limenitis arthemis astyanax* (Identification, Chapter 4) deposit a single egg. Duly recorded on film (Photography, Chapter 2) as it advanced through its various developmental stages (Rearing, Chapter 6), it eventually emerged and was released in the garden where the egg had been laid.

The successive phases proceed counterclockwise from the center, following the ever-expanding Fibonacci spiral (see Glossary section), a pattern in the structure of numerous forms of plant and animal life:

(1) the egg.
(2) the first-stage larva on its "gangplank."
(3) in premolt second stage.
(4) a carefully crafted rolled-leaf hibernaculum, that some individuals make in order to rest in diapause until the next season.
(5) the middle-stage larva, mimicking a bird dropping.
(6) the fully grown larva.
(7) the prepupa, suspended and preparing to transform into a chrysalis.
(8) the completed chrysalis.
(9) the emerged adult.



Basic Techniques for Observing and Studying

Moths & Butterflies

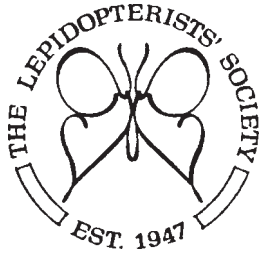
William D. Winter Jr.

The Lepidopterists' Society
Memoir Number 5, 2000

Dedication

For my wife, Jo Brewer, my companion and teacher on countless fascinating lepidopteral explorations, who has spent so many years opening the eyes of children to butterflies and the natural world.

Editor: William E. Miller
Cover Design: Seth Brewer, using photographs by the author.
Book Design and Typography: Kay McGinnis Ritter



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William D. Winter Jr.

1923-1998

William D. Winter Jr., known familiarly as Dave, was a pediatrician practicing in the Boston area. He attended Hamilton College, Clinton, New York, and received his medical degree from Harvard University.

A life-long northeasterner, he developed an early interest in Lepidoptera. That interest deepened over many years, and was shared by his wife, Jo Brewer.

Together and separately they wrote articles and books about moths and butterflies, both scientific and popular, for young people as well as adults. Their book *Butterflies and Moths: A Companion to Your Field Guide* (1986) engagingly introduces readers to the enjoyment of moths and butterflies. Long active in the Lepidopterists' Society, Dave Winter was also an early advocate of butterfly gardening.

He will be remembered for his devotion to lepidopterology as an avocation, but the methods and resources he lays out in the present book will serve all lepidopterists well whether vocational or avocational. He died after completing a draft of this book.

Jo and Dave butterfly hunting on a Hawaiian vacation.

Special Thanks

I would like to take this opportunity to thank two very special people who allowed my father's dream to become a reality. He spent the last years of his life writing but was unable to finish up before he passed away. I was left with a rough manuscript with no index and pictures that needed finishing and placement. I was fortunate enough to have found Ms. Kay Ritter by chance, and she has spent a great deal of time putting it together, converting the computer files, updating the illustrations, and designing the layout to give it the look and feel that you hold in your hand.

The true driving force throughout the entire writing of this book is Dr. William Miller from the Dept. of Entomology at the University of Minnesota St. Paul. As Editor, his help and expertise was invaluable to my father, Kay, and myself. He devoted a tremendous amount of his time towards the accuracy, content and performed the major task of indexing the complete text. Throughout the project Bill was always there with a helpful suggestion.

Again, I want to thank Kay and Bill for bringing this book to life.

Scott Winter

Table of Contents

PREFACE

In the very first issue of *The Lepidopterists' News* (1947), the one publication of The Lepidopterists' Society between 1947 and 1959, the practice was begun by the editors and several contributors of providing information on all sorts of techniques involved with the collecting, rearing, preservation, shipment, identification and many other aspects of working with the scaly-winged subjects of our impassioned interest. In 1959 the name of the *News* was changed to the *Journal of the Lepidopterists' Society,* to handle primarily scientific articles, and a new publication, the *News of the Lepidopterists' Society* was born.

Over the years several columns in our publications have been devoted to this topic: "Questions for Professor Forbes" in the early issues, and later "The Spreading Board," "Culture Corner" and "Wingtips" in the *News*. These columns, plus many individual articles contributed by members, presented many fine ideas, but they were not organized and are difficult to find, being scattered through the literature.

A few years ago I felt that all these ideas, plus the many new ones discovered or invented by lepidopterists, should be brought together in a pair of volumes: a basic techniques manual for beginning and intermediate lepidopterists, and an advanced manual aimed more at specialists in various aspects of the field. Here is the first of these volumes.

Most books on butterflies and moths have sections dealing with techniques that are addressed herein; but they tend to be limited in scope, and again, broadly scattered. The detailed treatment of many subjects, some of rather recent interest, should prove the worth of this book.

New ideas are forever gestating, and we hope that if you have comments or new approaches you will share them with your colleagues through the medium of the *News*.

The officers of the Society and I are deeply indebted to our late colleague W. D. Winter Jr. for undertaking this project and bringing it to fruition despite the incapacity wrought by Legionnaires' Disease. Dave served the Society faithfully and ably as Secretary and Editor of the *News*. I am also grateful the *Memoirs* Editor William E. Miller has completed the manuscript and seen it to print after Dave's untimely death.

Charles V. Covell Jr.
Past President, and Member, Executive Council,
The Lepidopterists' Society

FOREWORD

When I launched the Lepidopterists' Society in 1947, with Harry K. Clench and major participation by Jeanne E. Remington, it was clear that there were very many butterfly and moth collectors in North America with passionate interests in the Lepidoptera and a wide diversity of sophistication in pursuing their enthusiasms. There had already been interchanges of course, especially between very active amateurs interested in exchanges, and between the rather small array of older scientifically trained professionals who kept in touch with each others' research, notably J.H. McDunnough, the two John Comstocks, W.J. Holland, W.T.M. Forbes, and successive Barnes Collection curators (A.W. Lindsey, J. McDunnough, and F.H. Benjamin), H.G. Dyar, Carl Heinrich, August Busck, A.B. Klots, and a few others. A much more formalized association was needed and wanted, and the Society was an idea whose time had come. Equally important was a world-wide linkage of lepidopterists in all countries. Interpersonal contacts, and exchange of ideas and expertise, were a focus of the new organization.

From the beginning the newsletter featured advice on techniques of collecting, rearing, and curating specimens via many short articles. The culmination of this enterprise is in your hands, Dave Winter's superb manual. New complexities now exist, such as legal regulation of selecting and protecting endangered and threatened species, ethical aspects of collecting, and tax regulations relating to donations and sales. This manual is at the State of the Art contemporaniety. Very likely revisions will follow from time to time. I will suggest subjects for some likely new chapters in the next edition: molecular procedures for classification; a simple exposition of taxonomic reasoning, notably cladistics; procedures for measuring population size; guidelines for understanding the heredity of intra- and interspecific characters; experimental procedures for testing palatability, secondary compounds of Lepidoptera foodplants, and pollination preferences; the several parts of conserving rare species — captive breeding for reintroduction, corridor planning for dwindling species, population viability assessment for scarce species; educating the media, school children, and the public caring for natural history; and regional atlases of moths with emphasis on abundance shifts over substantial time periods.

Charles Lee Remington
Cofounder of The Lepidopterists' Society

INTRODUCTION

The observation and study of Lepidoptera (the large order of insects that includes the butterflies and moths, characterized by four membranous wings covered with small scales; *lepido*, scaly + *ptera*, wing) began, historically, with the curiosity of amateurs. The science (lepidopterology) followed, as dedicated amateurs began to devote more time to the study, shared their discoveries with others, and accumulated the knowledge that forms the basis for the education and development of professional scientists. This close interaction between avocational and scientific lepidopterists continues, so closely, in fact, that the distinction between amateur and professional is sometimes difficult to discern.

Among biologists in the broad sense, there are few who do not rate a childhood interest in the observation and collection of butterflies as a major influence in their choice of a career in natural science. For children, Lepidoptera rank with flowers and birds as "eye-openers" to the natural world.

About Collecting

Why an emphasis on collecting and preserving Lepidoptera? To work and plan effectively, biologists must know what species they are dealing with, and how they relate to other species. Collected specimens are necessary to make such determinations, and to indicate that different scientists are or are not working with the same species. Visual identification of many species of insects in the wild, especially most moths and some mimics, is unreliable or impossible. Collecting also is necessary to obtain livestock to rear, particularly with unknown or little-known species. Collecting is often necessary to estimate numbers of a species, and to monitor changes in population size from place to place and from time to time. Intelligent, reasoned, purposeful collecting can save individuals for educational and scientific purposes, before they enter the food chain as nourishment for birds and other predators.

A goal of biological science is to understand the environmental needs of each moth and butterfly species. Collected specimens form part of the "warp" of the fabric of understanding that enables us to repair or avert environmental degradation.

There are not now, nor will there be in the future, enough professional lepidopterists to do the necessary world-wide collecting. They will continue to rely on the work of properly skilled amateurs to help them in this huge task. Older experienced collectors do not live forever, nor can they reach out to as many young people as may wish to learn. It is the aim of this manual to record useful and proven techniques for studying Lepidoptera, so that those coming into the field may learn how to contribute in a competent and useful manner.

Observation and study of moths and butterflies leads to an appreciation of their intricate interrelationship with their environments, and of the critical dependence of many species on their very narrowly defined habitats. Yet knowledge of these environmental requirements is limited or unknown for many species, even in highly developed and studied countries. The multiple and continuing observations of amateurs are needed to fill in the gaps in our knowledge.

About This Book

The sequence in which material is presented here loosely follows that which many of us have followed in the development of our own interests in Lepidoptera: noticing moths and butterflies and where they occur; capturing specimens to learn more adequately what it is we are watching; learning to prepare and preserve retained specimens so they will be of future

value to ourselves and to others. In parallel with, or instead of, this sequence, many of us photograph Lepidoptera, rear them to learn of their immature stages, or enhance gardens for the benefit of butterflies and moths. Whatever your personal bent, it is hoped that you will find here techniques and pointers that will enhance your enjoyment of this fascinating and often life-long avocation.

Mine has been the pleasant task of assembling between two covers the multitudinous ideas, techniques, and maneuvers which make these activities possible. Source material includes formal and informal articles relating to Lepidoptera, dating back 50 years and more, relying particularly on techniques described in the newsletters of the various lepidopteral organizations, or retrieved from the minds of various lepidopterists, who pass them on by word of mouth. The search has not been exhaustive, nor has it always been possible to identify a published report as original, as opposed to a modification, clarification or other treatment of an earlier work. Some techniques have stood the test of use over decades or centuries; others are in their infancy and may or may not survive.

This manual includes many techniques, not because each is the only or best answer to a particular need, but because it has been helpful to one or many persons and may be right for you. The goal is not to urge uniformity, but to indicate what has been commonly found useful, while encouraging improvisation.

Obsolete or very dangerous techniques and chemical agents, described in many widely available texts, are discussed here to clarify the reasons for abandoning them. You are enjoined against using these procedures or agents; alternatives are described.

Use of this manual presumes a passing knowledge of the general structure and life cycle of Lepidoptera. This is adequately covered in the introductory chapters of the popular field guides.

Beginners, and parents and teachers seeking to assist children in learning about butterflies and moths, may understandably feel intimidated or put off by the volume of material presented here. To show how simple the entry level can be, a "minimum box," near the beginning of appropriate chapters, lists the half-dozen or fewer things that are necessary for initial participation in the subject discussed. By choosing selectively from the material in the text, it is easy to start at the surface, then dip more deeply as interests broaden and skills increase.

In general, the content of source references has been paraphrased for succinctness or clarity, or to meld related articles. In some instances published articles contained ambiguities that I tried to clarify by contacting the author(s). If no response was received, I resolved the uncertainties myself or with consultation. If this has resulted in errors, the responsibility is mine.

Citation of a reference on a particular subject means that it is treated in more detail there; it does not imply that the idea or the technique was necessarily original with that author. A deep search to determine origins did not seem germane.

Names of individual species, as much as possible, are common names backed up by Latin binomials. For higher groups, the choice has usually been anglicized family or subfamily names: for example, geometrids rather than Geometridae.

In keeping with the need to ease Americans into the metric system, measurements are presented first in metric units, then in English (in parentheses). However, one is not a meticulous three-decimal-place conversion of the other. On the contrary, the accurate figure, be it metric or English, is (1) the unit used in the source article, or (2) the size you would look for in a store or catalog in the United States. Thus you would look for a 100 mm (4") petri dish by its metric dimension, but for a 4 liter (1 gal.) jar by its English equivalent. Where standard metric substitute dimensions could be ascertained (as for some wood products), they are used instead of rounded-off conversions. Screw threads are not convertible, and only U.S. terminology is used for them.

Inclusion of brand names is for reference or identification only and does not constitute

endorsement of products.

The table of contents introduces an array of appendices on disparate subjects, referred to throughout the text.

This manual should offer each reader answers to some of the problems he or she has not yet solved. In addition, it should help to show which problems remain unresolved, and thereby indicate where one's imagination and ingenuity can usefully be directed. In a "whodunit" you must necessarily read cover to cover to get the full message. In this "how to do it," on the other hand, your needs may be concentrated in a few chapters or spread over many. *Everyone, however, could benefit from reading Chapter 10 on potential hazards of the activities of any naturalist and of lepidopterists in particular.*

Photographs and drawings are by the author, unless otherwise attributed.

The last word on techniques for studying Lepidoptera has not been written. Just as "there is more than one way to skin a cat," there is more than one way to deal with almost every facet of the study of Lepidoptera. The goal is to help you find the ways that work best for you.

About Lepidopterists

Lepidopterists are tinkerers and improvisers to an extreme. We devise our own methods and equipment for observation and rearing, generally at trivial expense. Your own solutions and inventions may well prove superior and worth sharing with the rest of us, through the pages of the *News of The Lepidopterists' Society.*

A high proportion of lepidopterists are hopelessly addicted to lepidopterology, as their spouses (many of whom share the addiction) will readily attest. Among the symptoms are an urge to share with any willing listener the marvels and details of metamorphosis, different for each species, the unbelievable strategies by which these insects strive to survive in a naturally predatory world, and the sheer beauty of form and behavior of so many.

If this manual can increase the enjoyment of Lepidoptera by amateurs, if it can increase the scope and accuracy of observations, if it can contribute to making lepidopteral activities purposeful, educational, and scientifically significant, it will meet its short-term goal. If it can also help to lead children into the ranks of future natural scientists and scientifically literate conservationists, it will prove a humble contribution to the future well-being of all of us.

ACKNOWLEDGMENTS

The box at the end of each chapter gives the names of the consultants who reviewed that chapter and made valuable recommendations. Their help was essential in preparing this manual. Their names follow, in retrobetical order:

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Seth Brewer, who created the cover, Jeff Brewer, who took many of the photographs, and Scott Winter, with his indispensable logistical assistance, made this truly a family undertaking.

I am particularly indebted to William E. Miller, Editor of the Memoirs, who has borne with me with greatest patience and understanding through this protracted endeavor.

And to the numerous unnamed others, alas, misfiled in my aging cerebral pigeon-holes, I offer my most sincere thanks.

Chapter 1.

OBSERVING MOTHS AND BUTTERFLIES

Interest in moths and butterflies most often starts with a chance glimpse of a passing large or colorful butterfly that whets the appetite for a further look. If you are already trained in following wildlife (as birders are), you have some of the necessary skills. You try to be as cautious as possible, to avoid causing alarm, and to anticipate what the creature will do next. But the most important skill is learning to understand where, when, and under what conditions butterflies or moths (and their immature stages) are active or quiescent, so that you can seek them out successfully.

Minimum needs

- *Curiosity*
- *An open mind*
- *Sooner better than later, a field guide covering your area*

1. Where and When

Since local vegetation has a strong bearing on what species of Lepidoptera may inhabit a particular place, sites with more varieties of plants are likely to contain more kinds of Lepidoptera. Highly productive sites are disturbed areas or old fields, edge zones such as roadsides, railroad verges, power-line cuts, stream banks, or small openings in forests. Monocultures, whether man-made or natural, are less productive. But no habitat seems to be devoid of Lepidoptera, if you look long enough and at the right time. Particularly in the field of the micromoths, new species continue to be discovered in the most unlikely places.

"Concentrators"

"Concentrators" are important in the search for butterflies. Patches of nectar-yielding flowers, scattered here and there in fields or along roadsides seem more productive than a broad expanse of flowery terrain. Home gardens and even city parks attract many butterflies. The most-favored nectaring plants can vary considerably from place to place, so keeping notes as to what is being visited locally can be very useful. Sap-flows, from accidental damage, or from the work of boring insects, woodpeckers, or man (such as tapping a sugar maple in early spring) are often heavily patronized. Birch sap also attracts in spring, and willow in prairie situations (Martin 1977). The

ooze on an elm tree dying of Dutch elm disease is a specific example (Peacock 1981). Wood nymphs and nymphalids frequently visit the honeydew of aphids on infested trees and shrubs (Pyle 1992). Puddling sites—moist patches of bare earth—may be visited by dozens or hundreds of (usually male) butterflies seeking salts and nitrogen-containing substances. Lepidoptera feeding at dropped fruit, rotting and fermenting on the ground, or at leakage about wineries, are often so subdued by imbibed alcohol that they are quite undisturbed by human spectators. Fecal deposits and urine, of beast or bird, are equally attractive. And many species avidly visit carrion—from a road-kill butterfly to a bloated bovine in a tropical pasture.

Feeding-sources used by butterflies can be reexamined in the evening, during the night, and just at daybreak for moths—"winter moths" in late autumn and early spring, sphinxes in their flight seasons, noctuids, especially plusiines and underwings, geometrids—any species except those that, as adults, lack functioning mouthparts (such as the saturniid giant silk moths). At a given site, such as a milkweed patch, there will often be a sequence of different species throughout the night, each with its preferred feeding time. In southern Florida, Brown (1976) noted peaks of activity at dusk, just after dark, and $2^1/_2$ hours after dark, with a reversed sequence related to dawn. Some species did not participate in the early morning periods—often true of lepidopterists also. Brown observed that insect repellents repelled sphingids, and that lights disturbed them. He recommended dark clothing, squatting to get a low line of sight at the flowers, and detecting the presence of moths by flower movement, rather than using a light. For collecting, he used a dark-colored net. More recorded and reported observations of these differences in timing would be of great interest. Nielsen (1981) identified 20 species of looper (plusiine) moths nectaring at fireweed *(Epilobium angustifolium)* and associated flowers at dusk and for the first two hours thereafter. A headlamp was very useful, with care to keep the brightest light away from the moth.

Blossoms of larval foodplants of the noctuid genus *Schinia* should be searched for resting moths by day. The handsome small moths blend cryptically with their surroundings.

Even puddling goes on at night. Members of ten families of moths have been recorded at puddles, especially in relation to the recent

presence of animals (Adler 1982).

While your interest in Lepidoptera may not lead you to collecting, you may find the Chapter 7 sections on "Baits" (Section 4) and "Lights" (Section 3) useful. Sugaring for moths is like feeding birds: it concentrates the creatures in a place of your choice. Hanging a blacklight is like hanging a bird feeder, albeit considerably more chaotic at times.

Looking around for activity at eye level is a natural tendency, as is looking down for species nectaring on ground-cover blossoms, or basking. Looking up also pays off. Many lycaenids perch on branches 10–20 feet above the ground and dash out to investigate passersby. Some perform their courtship activities about the tops of young cedars or oaks. Such butterflies appear as contrasting small dark triangles at the tip of a branch. A stick or a handful of gravel tossed up past the ends of some likely-looking branches may activate some you haven't seen. Many tropical species, and some in temperate zones, conduct most of their business in the canopy, and the down-turned eye will never notice them. In addition, some species, perhaps because of low population densities, convene near hilltops and mountain summits to locate potential mates. Such sites are worth investigating.

Geographic Range

Each species of moth and butterfly that has been studied sufficiently has a known geographic range, and these ranges are described or mapped in the more comprehensive field guides and Lepidoptera texts. Some species adhere closely to their historic ranges, some have ranges that are shrinking rapidly into a smaller area or are breaking up into isolated pockets. Others in recent decades have expanded their ranges strikingly at one or more borders. And still others, in occasional years, will make a major emigration, usually to higher latitudes or altitudes. The books can tell you what to expect, but expect to see more than that, now and then.

Habitat

Some species are quite vagile, wandering broadly and found in every county (or even every acre) within their ranges. Others, in contrast, occupy very localized habitats that provide the proper larval

foodplant and acceptable nectaring plants. Using field guides to trees, shrubs, and flowering plants greatly improves your search. Once you locate the proper habitat and foodplant, the butterfly may be easy to find. Yet every habitat that meets the classical criteria for a particular species may not be occupied.

Flight Period

Each species of moth and butterfly has its own flight period, the time of the year when the adult stage is on the wing. The field guides and texts give the specifics for each. It may be a couple of weeks once a year, or several times a year for those species producing multiple broods—and this varies with latitude and altitude. The adult life span may stretch over many months, especially if it is a species that hibernates in the adult stage.

Time of Day

Time of day is also significant. In general, butterflies are active in the brightest part of the day (but see under "Temperature," below) and moths in the dimmer or dark parts. But there are many day-flying moth species, and a few species of butterflies are crepuscular, flying at dusk or dawn, or in the case of some tropical skippers, in the hour just before dawn. Some sphinx moths nectar primarily at the crack of dawn, when few lepidopterists are out looking. Within a single moth genus, some species are most active early after dark, others before midnight, and still others in the small hours of the morning. Some individuals don't seem to know the rules—such as butterflies attracted to lights in the night, or normally nocturnal moths seen nectaring at flowers in bright daylight. So to find a particular species, you may need to know, or discover, its habits.

Temperature

Temperature controls activity. For the most part, to sustain flight, the muscles of the thorax that power the wings must be in the range of 24–44° C (75–112° F). For this to be possible, the air temperature must usually be above 13° C (55° F): the difference between air temperature and flight temperature is usually made up by absorbed heat from the sun (basking), or generated internally by rapid coordinated contractions of opposing pairs of flight muscles (vibrating). At low

temperatures, especially at high latitudes or altitudes, if a cloud obscures the sun, a flying butterfly cools to the point of alighting to wait for another opportunity to bask. On the other hand, on very hot days the combination of outside heat plus the heat generated by the muscular activity of flying can raise thoracic temperature above the danger point of 46° C (115° F). At such times some butterflies "stand on tiptoes" with the wings folded over the back to cast the thinnest shadow possible and reduce the amount of sunlight striking the body. Others rest in a shaded place during the middle of the day, restricting their flying to early morning and late afternoon (Douglas 1986). But there are many exceptions—micromoths that can spring instantly into flight at less than 4° C (40° F), and "winter moths" that can fly and forage at air temperatures down to 2° C (35° F).

Weather

For butterflies, sunny weather is clearly the most productive, unless heat is excessive. In mountainous areas, the development of heavy clouds by early afternoon can put an end to the day's activities. Increasing wind reduces the opportunity for sustained and accurate observation.

For moths, wind is again an enemy. The best observations at nectar flowers or at "sugar" bait (see Chapter 7) come on nights that are calm or nearly so. Cloudy skies are friendly, with rain being an added asset. In four weeks of observation on a white-painted veranda in Borneo, Wallace (1869) noted that the numbers of moth species and individuals coming to his lantern were greatest on clouded, rainy nights, and least when it was clear or when the moon was full, clouds or no clouds. Similar observations have been made in temperate climates, although here clouds seem to mitigate the adverse effects of a bright moon. As with all dogma, however, any collector can recall the exceptional night when the conditions were "wrong" and the number of moths flying was outstanding.

The foregoing has addressed the generalities of finding adult Lepidoptera on the wing. Many specifics, including the search for immature stages, are detailed in Chapter 6, on rearing.

2. Enhancing Vision

You will generally seek and see moths and butterflies with your

unaided eyes, but on repeated occasions you will want a closer look without having that closer look frighten the insect.

Binoculars

Binoculars are a ready solution, and the characteristics of the binoculars are vital. They should be lightweight and easily carried. Magnification of 7x, 8x, or 10x is commonly used ("10x" means an object ten feet away appears to be one foot away). Objective lenses broad enough to admit ample light are desirable (the "25" on 8x25 binoculars indicates objective lenses 25 mm in diameter). The most important characteristic of good butterflying binoculars is the near focal distance, which should be six feet or less. Binoculars with a longer near-focus leave the observer backing away from the subject in an effort to get a sharply focused view.

If you wear eyeglasses, keep two additional points in mind. You should be able to see the entire optical field without removing your eyeglasses, and the binocular eyepieces should have soft rubber rims that will not scratch your eyeglass lenses.

Be deliberate in your choice of instrument, and look at brands well known for quality. There can be variation among individual binoculars of a given model (Glassberg 1993). Make your choice in terms of function and personal fit.

Eyeglasses

Special mention of eyeglasses may seem superfluous. However, all of us, sooner or later, but usually in the fifth decade, lose the ability to focus clearly on nearby objects, and all of us who need corrective eyeglasses for ordinary good vision then graduate to bifocals. These have serious drawbacks for the lepidopterist, as there is an intermediate range where focus is poor, and within that range it is easy to overlook small butterflies, larvae and eggs. The solution lies in "variable bifocals," for which your ophthalmologist can provide the prescription. These lenses vary gradually from your distance correction at the top to your near (for reading) correction at the bottom. A correct focus for any distance is thus attainable by a slight tip of the head, up or down—a technique mastered, almost unconsciously, within a few minutes, or at most, a few hours after acquiring the lenses (Winter 1994).

A camera is a highly desirable piece of equipment for the lepidopterist. Photography is the subject of Chapter 2.

3. Behavior in the Field

Lepidoptera, with their large compound eyes, have a very broad field of vision and are particularly adept at detecting movement. Make any close approach slowly, smoothly and cautiously. While approaching from the rear may be least likely to startle, it also gives a very limited view of a butterfly perched with its wings closed. Only trial and error teaches the best angle for a particular situation. Clothing of subdued colors with minimal contrast helps. Being downwind or upwind is not significant, as Lepidoptera do not use odors to recognize humans as potential predators. While sounds generally do not seem to startle Lepidoptera (those with ears are tuned particularly to the echolocation frequencies of bats), a few species of day-flying moths have been noted to "spook" instantly at the click of a camera shutter (Winter 1991). (Possibly the click mimics that of a swallow's bill as it snaps at a flying insect.) By contrast, the glare of a camera flash often does not startle.

All land belongs to someone, and you should consider obtaining permission before entering an area in search of Lepidoptera. Specific regulations apply to the various government-owned lands, forests, and parks, and to Native American lands. Private landowners can vary greatly in their attitude towards intruders. This subject, and constraints regarding habitats of endangered species, are considered in Chapters 11 and 12.

Lepidoptera-watching is an avocation enjoyable singly or in groups, and it is particularly rewarding when shared with children. Their innate curiosity can be exercised to the fullest, and having extra pairs of eyes along increases the breadth of observation. One lepidopterist has noted that when his twins were preschoolers they spotted more lycaenids and skippers than he did—they were down closer to the action! Dogs, on the other hand, are no asset. They seem to have a penchant for dashing through the scene just as you are about to snap that perfect picture.

A quotation from Rockwell (1995) is appropriate here:

> "Over years of tromping fields and ravines . . . I've come across some things that [are very intriguing] and often quite

different from the descriptions of behavior in the few popular books that delve into the subject.... Most of these descriptions seemed to be using behavior habits [to make it easier to find and catch the animals, rather than] as an activity interesting for its own sake.

"To begin to understand where Lepidoptera fit into the overall scheme of nature, I believe one must observe behavior in its own place where the animals live, not just at the garden and roadside sites they visit. That is fun, too, but it is not entirely natural...Birders already do this, at least those looking beyond a quick peek to add to the day's, year's or lifetime's list. Whether they admit it or not, successful collectors make observations; [I appeal especially to them] to pause when they see activities not normally [seen] and simply watch and take notes for a while. Some of these observations...might even become important keys to identification. There are a number of species that seem to have greater variation in behavior...than they do in [color and pattern]."

4. Road-kills

A few people, who eschew active collecting but wish to have a collection of specimens for closer examination, do "passive collecting" — picking up road-killed specimens from highways and roadside shoulders. This is best done by bicycle, considering the distances to be covered. In this fashion one person amassed a collection of the complete roster of butterflies known for New Jersey, and even succeeded in rearing some larvae from ova produced by injured but still living females. This approach works poorly for moth collecting. Mice glean through the night and birds arise at the crack of dawn, quickly removing the night's casualties from pavement and shoulders.

Specimens so collected should be dealt with by methods covered in Chapters 7 and 8. They should be labeled as road-kills; this implies the possibility that the specimen might have been carried many miles on a radiator grille before falling off at the site where it was recovered.

5. Special Activities

Tagging programs are clarifying the migration and overwintering biology of the monarch butterfly in North America. Any interested

observer equipped with a net for capturing adults, or rearing larvae to the adult stage for tagging and release, can participate (techniques for rearing and collecting are covered in Chapters 6 and 7). The programs supply field collaborators with numbered stick-on labels bearing the request that the person finding the labeled butterfly return the label to a specified address, accompanied by a statement as to when and where the butterfly was recovered. To apply the label, you rub a patch of scales from both surfaces of the forewing at the leading (costal) edge, in towards the body, then fold the label over the edge of the wing and press the sides firmly together. If the scales are not removed down to bare the transparent wing membrane, the label will not adhere. Take care not to kink or damage the front edge of the wing. Tagging programs for monarch migration surveillance are conducted by the Insect Migration Association, Journey North, the Monarch Migration Association of North America, The Monarch Program, and Monarch Watch (see Appendix K). These organizations rely heavily on input from amateur observers and from students in elementary and secondary schools.

An annual "Fourth of July Butterfly Count" is performed in multiple localities and is open to all interested persons. Year after year population studies in defined areas yield valuable information and are an interesting educational and recreational activity. The count was initiated by the Xerxes Society and is currently coordinated by the North American Butterfly Association (see Appendix K).

6. Identification and Records

Learning to identify the moths and butterflies you observe is personally satisfying, and is essential to intelligent observation and to sharing observations with others. It takes a rather short time, with the help of the field guides and other texts, to become acquainted with the general characteristics of the different families of Lepidoptera. Then, consideration of the details of size, color, pattern, behavior, timing, and geography makes reliable field identification of most of the larger species possible. Photographs you take may help you identify additional species, while others may require close examination of a spread specimen, or even dissection by someone familiar with fine anatomical details. Netting specimens (that can subsequently be released), and using forceps and a hand lens for

examination of small features, aids in identification. Sometimes transferring a small lycaenid or skipper to a clear plastic vial simplifies examination.

Many people are personally comfortable with "more or less" naming similar or difficult species, and others are content only with a definitive answer. Chapter 4 goes into the identification process in greater detail.

Whether or not you immediately recognize the name of your observed subject, it is valuable to keep some record as to where, when, and under what circumstances you found it, a rough description in lieu of a name, and any behavioral observations of interest. Since memory blurs with time, timely notes make a difference. A pocket notebook is a partial solution, but even better is a pocket mini-recorder into which you can dictate the pertinent details while leaving your eyes free for further observation. Record-keeping is the subject of Chapter 3.

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Chapter 2.

PHOTOGRAPHY

While a camera is not essential for the observation of moths and butterflies, photography adds even more to the enjoyment of this avocation than it does to most others. The primary advantage may seem to be aesthetic—to be able to record and to share with others the beauty of the living insect engaged in its normal activities in its natural surroundings. An even greater advantage is that photographs objectively capture the details of behavior—the basking posture of an alpine butterfly, the drama of a "blue" larva being carried off into an ant nest, or the puddling conviviality of a hundred swallowtail butterflies. Commonly the same photograph achieves both goals, regardless of the intention of the photographer. Clear, well composed photographs are well worth the required efforts. Fortunately, current cameras, flashes and films have most of the solutions to technical problems "built in." Photographs are still our best means for recording behaviors, larval colorations, habitats, and many other facets of moth and butterfly lives.

Minimum needs

- *Single lens reflex camera with close-up capability*
- *ASA 100 or 200 speed color film*

1. Cameras

Almost all outdoor insect photography is done with 35 mm single lens reflex cameras. Most standard brands of cameras now come with excellent lenses, such that anyone can capture superior images suitable for publication. A high degree of automation has been developed. Exposure can be controlled by simultaneous automatic adjustment of both shutter speed and lens aperture. You can also select a specific aperture to maintain desired depth of field, retaining automatic selection of shutter speed, or you may select a specific shutter speed to stop action, retaining automatic setting of lens aperture for proper exposure. Supplementary flash can be activated and automatic metering will regulate the duration and intensity of the flash for you.

The addition of automatic focus can reduce the process to a "point

and shoot" operation. But many photographers prefer to use manual focus, since the camera often does not select the plane of focus the photographer wants in close-up or low-contrast situations. Some automatic exposure cameras have an in-focus indicator that is very helpful when you are focusing manually. An exposure compensation dial is also valuable, so that you can manually under- or overexpose a white or very dark subject to achieve a desired result.

A depth-of-field preview button allows you manually to stop down the lens aperture to determine the depth of field necessary for your subject. If you have to work with a shallow depth of field, it is critical to keep the plane of the subject parallel to the film plane to ensure sharp focus all across the image. The preview button allows you to check this positioning.

Automatic film advance simplifies taking serial shots of a "twitchy" subject or insect behavior, and automatic film rewind speeds reloading. Current cameras automatically set the film speed from bar-coded cartridges, a great convenience. A cable release socket (for tripod work), and provision for a synchronized and automatically metered off-camera flash to provide angled lighting for emphasis of surface details, are additional useful features in photographing all stages, especially immatures.

The focusing screen provided with most cameras is the "split image" type. This is unsatisfactory for close-up insect photography and in low-light situations, since one side tends to black out unless the center of your eye is precisely in the optical axis of the viewfinder. Your local camera dealer should be able to let you test one or two types of focusing screens that are more satisfactory for close work. Many photographers find a bright, clear matte screen best.

Battery type is important. The lithium batteries found in many autofocus cameras with built-in flash are very expensive and often hard to find, especially in underdeveloped countries. One battery may last through only 5–10 rolls of film. Cameras using AA batteries have a more easily replaceable power source.

2. Lenses

There are a number of desirable characteristics for lenses used to photograph moths and butterflies and their offspring. Optical quality, with control of chromatic and spherical aberration, and achromatic

coating, are basic and present in the products of most reputable lens manufacturers. If you choose lenses from the same maker as your camera body, the automatic coupling mechanisms will be compatible. Some other makers, however, produce very satisfactory compatible lenses. In addition, couplers are made that allow pairing of any two camera systems. These are readily available through the many mail order stores that advertise in photography magazines. Use of the lens hood accompanying your lens will cut down flare problems in outdoor work.

Image ratio describes how large the subject appears on your slide or negative. Many lenses used for routine photography of people and places will have a maximum image ratio of 1:4 or less, that is, the largest possible film image is $^1/_4$ the actual width of the subject. While this is adequate for large butterflies and moths, it is disappointing when used for individuals of medium size or smaller. A "macro" lens (Nikon sometimes uses the term "micro"), on the other hand, gives a 1:2 or 1:1 (life size) image ratio, and even greater magnification with auto-coupling extension tubes or bellows. These added extensions, however, result in loss of light, and the bellows is generally too clumsy for field work.

Near focus indicates the distance from film plane to subject at the highest image ratio of the lens. More important is the working distance, the space from the front of the lens to the subject. This determines how closely you must approach the butterfly to obtain the largest possible image. The longer this distance is, the less likely you are to alarm your subject. Likewise, not having to bend over so far can be a boon to an achey back. The near working distance of a 50 mm macro is around 8–10 cm (3"–4"), while that of a 100 mm macro is 20–30 cm (8"–12"), depending on the model. The 100 mm is clearly preferable, despite its greater bulk and weight. Even 250 mm macro lenses are available, but at much higher cost.

Zoom lenses have their uses, especially for taking habitat shots or photographing within butterfly houses. But a maximum image ratio of 1:4 with even a "macro-zoom" has its limitations. Most zoom lenses are not as sharp, especially up close, as single focal-length macro lenses. There is one macro-zoom with an image ratio of 1:1 to 3:1, so it is suitable only for very small subjects, at a working distance of 2.5–5 cm (1"–2").

Closer focusing and higher image ratios can also be obtained with many short telephoto and zoom lenses by using very high quality close-up lenses. These two-element lenses attach to the front of the lens and all autofocus and autoexposure functions are retained. They offer remarkably improved results over the diopter accessory lenses that have long been available. These lenses do not reduce the amount of light reaching the film, in contrast to extension tubes and bellows.

Perhaps the main questions to be answered in choosing or upgrading your camera equipment are:

(1) How sophisticated will your photographic needs (or desires) be?

(2) How much money can you spend?

(3) How much weight do you want to carry into the field?

A camera body may weigh one or two pounds, and an autofocus 100 mm macro lens can add an additional 18 ounces, more or less. The camera setup, including any necessary extension tubes and flashes, should be carried assembled and ready to shoot, or the subject will depart while you are putting your act together!

3. Film

It is possible to photograph your subjects in black and white, but most people prefer to capture the incredible beauty of moths and butterflies in color. Color film speeds now range from ASA 25 (slow) to 1000 (very fast). Slower films have finer grain and greater apparent sharpness, an advantage when you decide to publish your once-in-a-lifetime shot of never previously documented behavior. But slow films mean wider apertures, slower shutter speeds, or both, reducing depth of field and ability to stop motion. Supplementary flash will partially compensate since the flash duration is typically less than 1/1000 second.

Fast films permit using just available light but have more graininess and less appealing color values. Some of the current color films (Fuji Velvia, Kodak Elite) have very intense colors and high contrast. These films may be too contrasty when used with supplementary flash.

Color films and processing permit the choice of three different outcomes: processing for color transparencies (slide film), for prints from negatives (print film), or for prints and slides (movie print film respooled for 35 mm cameras). The last is very inexpensive, but the

quality varies widely. This film should be selected only if price is the principle consideration.

Use of print or slide film is an individual choice. Prints are much easier to view but more costly per image and bulkier to store. They are inherently less color-stable (except for Cibachrome prints from slides), but if a print is damaged, a new one can be produced from the negative. The exposure latitude for print film is also about three times greater than for color slide film and careful printing can correct for poor color balance. Slide films are very intolerant of exposure errors but are somewhat sharper than print film of the same speed. Most photo editors prefer slides to prints for publication.

Choice of paper for making color prints is important (Wilhelm 1993). Color dyes are incorporated within the paper, and the stability of the dyes determines the stability of the image. Allegedly the dyes in Agfa, Kodak, and Konica papers showed noticeable fading in the equivalent of 12–17 years of exposure to light, whereas those in recent Fuji papers showed minimal fading after the equivalent of 50 years. Ilfochrome classic papers, when properly laminated, carry a 200-year warranty against fading (commercial brochure, Holland Photo; see Appendix L). If the subject is cooperative, shoot several extra frames while bracketing the exposure. Slight movement or error in focus or exposure in one image is corrected by carefully taking additional shots. Multiple excellent originals are cheaper and better than any duplicates made later.

4. Lighting

Lighting for insect photography can range from reliance on available light only, through limited supplementary flash to allow shorter exposures and smaller apertures or to brighten overly dark contrasts in the photographic field, up to heavy use of supplementary light. The first gives "natural" pictures, and they are preferred by many but they may require long exposures and use of a tripod in the absence of bright sunlight. Aluminum reflectors provide a very effective, inexpensive method for augmenting light in the field—homemade, from a piece of cardboard about 20 x 25 cm (8" x 10") covered with lightly crumpled aluminum foil—to bounce light onto the subject.

Early morning provides one of the best opportunities to photo-

graph butterflies and caterpillars in the field using natural light. Windless conditions often present at this time allow use of a tripod, longer exposures, and fine grained film. The natural light is often diffused, softening the contrast and enhancing the color and detail of the subject. Cool morning temperatures often produce more cooperative subjects, giving more time to compose and shoot several frames.

Heavy use of flash often results in a beautifully crisp shot of a butterfly against a totally black background, a situation aesthetically displeasing to some. Examples of various proportions of natural and supplemental lighting can be found in the illustrations in *The Audubon Society Field Guide to North American Butterflies* (Pyle 1981). Experience will teach what approaches serve you best, and amateur or expert, taking a series of shots of one subject (opportunity permitting!) is the best way to assure creditable results.

The built-in flash found in most current cameras is very convenient and adds negligible weight. It can be cancelled for subjects basking in full sun on a quiet day. A butterfly perching obliquely on a gently swaying flower is photographed more sharply with the faster speed and smaller aperture that supplemental flash allows. Be sure (by doing some experimenting) that your camera's automatic metering terminates the flash quickly enough to avoid overexposures. For photographing a moth in a cryptic resting situation, or for moths in natural resting posture in a sheltered situation, use of a flash can be essential, just as it is for photographing underwing moths feeding at bait.

A camera designed to synchronize a separate flash unit with the camera via a "hot shoe," either directly or via a remote cord, provides greater versatility. The intensity and range of the flash can be much greater, and with the remote cord the light can be delivered from an angle, to emphasize the detail of surface irregularities. This is particularly useful when you work with immature stages. For natural appearance, supplementary flash should be positioned above the insect, like sunlight. The closer the flash, the brighter the light. This can overpower the existing light and yield dark backgrounds, but the brief flash duration can freeze any motion of the insect. Avoid brackets that attach a heavy flash to the filter threads of an autofocus lens. The weight can bind the autofocus mechanism. A pair of flash units supported by brackets at opposite sides of the camera can reduce

objectionable shadows and provide even greater light intensity. However, the added size and weight of such a rig may be objectionable in the field. A bracket manufactured specifically to position the flash above the lens and illuminate small objects is produced by Kirk Enterprises (Appendix L).

A ring-flash, wherein the light unit circles the end of the lens, is compact and lightweight. Because the ring source reduces shadows and highlights, its use for photographing immatures is frequently disappointing. (This effect can be countered somewhat by covering one-half of the ring light source with frosted plastic tape.) The major disadvantage of the ring is that it delivers less light to the subject, so that you may need to use lower f-stops or faster film speeds.

Whether you are using natural or artificial light, you can lighten up dim areas from any angle by using a white or foil-covered card held in the jaws of a spring clothespin. If the clothespin is attached near the top end of a dowel by a screw passed through the core of the spring, and the dowel is stuck into the ground, the card can be angled or rotated as desired. This can be particularly useful for early morning shots of resting insects.

A piece of equipment that can significantly increase the number of sharp, usable photos is a proper tripod. For field work, a sturdy tripod that permits work close to the ground while maintaining the camera in an upright position is best, and the tripod needs to work well on a variety of terrain (many photographers prefer Bogen, Gitzo, and Benbo tripods for field use.) It is essential if you wish to use natural light and exposure times of $^{1}/_{30}$th of a second or longer. Because of the dynamics of one's heartbeat, no one can maintain bedrock steadiness for exposures that long. Most young people in their prime can hand-hold a camera steadily enough to maintain focus reliably for shorter exposures or with flash, but many with more years behind us welcome a tripod, especially when working indoors or with immatures, where working rapidly is less critical. The tripod should have a central, crank-driven post for elevating and lowering the camera, and the head should both pan and tilt. A ball and socket tripod head facilitates following a moving subject. A focusing rail is invaluable and relatively inexpensive. Mounted between the tripod and the camera, the rail enables you to focus by moving the camera back or forward along its optical axis, rather than by rotating the lens

barrel. A desired image ratio on the film can thereby be assured.

5. Exposure

Most of today's cameras have very sophisticated through-the-lens (TTL) reflected-light meters, including center-weighted, spot, and multisegment. These meters have been calibrated at the factory to give proper exposure of a subject that reflects 18% of the light. Such subjects are referred to as middle-toned and are neither light nor dark, but in between. If you are using an older camera model, it is a good idea to check that your meter is properly calibrated. This can be accomplished easily by using the "f/16 sunny rule." This rule states that at f/16, proper exposure of a middle-tone subject, in bright sunlight, is at the shutter speed closest to the speed of the film being used. To calibrate your meter, take an exposure reading of a middle-tone subject, such as a gray card, at f/16 on a bright sunny day. For example, if you are using ASA 64 film, the camera's meter should read 1/60th of a second. If it does not read 1/60th, you need to adjust your film speed dial until you see that reading. With the meter properly calibrated, you can photograph any middle-tone subject and achieve a proper exposure. If the subject is not middle-tone, such as a white or very dark butterfly or moth, you will need to make an exposure adjustment to compensate for the subject's reflectance. Remember that your camera will make everything middle-tone. Without an exposure adjustment, your camera will underexpose white subjects and overexpose dark subjects. Effective exposure adjust-

Subject	Example	Adjustment
White	Cabbage butterfly	1 1/2 stops open
Very light	Bright yellow	1 stop open
Slightly brighter than middle-tone	Light green	1/2 stop open
Middle tone	Tobacco hornworm	No adjustment
Darker than middle-tone	Monarch	1/2 stop down
Very dark	Black swallowtail	1 stop down

Table 2–1. Effective exposure adjustments.

ments for handling non-middle-tone subjects are provided in Table 2-1 (M. C. Thomas, pers. comm.)

Exposure adjustments can be made either with the camera's exposure compensation dial or by manually altering the camera's shutter speed or aperture. Understanding how your camera's meter works will allow you to control how the final image will appear.

Overexposed slides are not correctable. Underexposed slides can be upgraded by as much as $1^1/_2$ f-stops in some sophisticated photo laboratories, but the cost is high.

6. Photographing in the Field

The comments on approaching butterflies in Chapter 1, Section 3, apply equally to field photography, but the photographer has to approach much more closely. It is frequently necessary to squat, kneel, or even stretch out prone on the turf (or worse), to gain the desired perspective. Your clothing will suffer, the integrity of your integument may be compromised, and in particular, your dignity may well be destroyed, if you were so foolish as to take it with you into the field! But one thing is certain—capturing a butterfly on film is a far greater accomplishment than capturing that same butterfly in a net.

Carrying the Camera

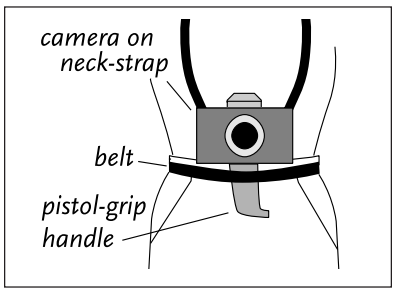

Figure 2-1. Pistol-grip for restraining camera.

Carrying presents particular challenges. A camera on a strap hanging around your neck needs additional restraint to keep it from bouncing about when you walk, or from swinging against a rock as you bend down. It is often inconvenient to use your hands for this purpose. Several very useful restraint methods also take some of the weight of the camera off your neck and shoulders. Harnesses are made with Velcro patches to hold the camera against your chest wall when not in use, or a chest patch can be applied to the front of a snug vest or jacket. A "Kuban Hitch" uses heavy but easily released rubber bands for the same purpose. If you wear a belt, a "pistol grip" (with the cable release removed) can be screwed into the tripod mount on the base of the camera, rotated 90° from its normal

orientation, and thrust inside the front of your belt (Figure 2-1). The camera is completely secure, instantly available, and most of the weight is carried on your hips instead of your shoulders. A home-made metal bracket removably attached to the base of the camera is equally useful. Continue to use the camera neck strap.

Lens Protection

Protection is necessary to prevent damaging scratches to the lens from grit-covered branches and foliage as you walk through the brush. Carrying a camera in a case adds weight and slows access. The standard snap-on lens cover is very easily lost. A tethered lens cap, swinging below the camera lens, will make an infant look at a camera, but its motion frightens butterflies away. A neutral or "skylight" filter screwed onto the front of the lens provides protection. However, the extra layer of glass between subject and film results in some slight degradation of image quality. The filter is easily removed for cleaning, and the lens cap can be added when picture taking is not imminent.

Rain and blowing dust will damage cameras and lenses, so take along an adequate tough plastic bag, or some similar means of camera protection. But avoid a totally airtight plastic bag: temperature changes can lead to precipitation on internal lens elements. Such internal fog may take more than 24 hours to dry out (and the process should not be accelerated by use of an oven!). Protection of the camera and its film (as well as spare and exposed film) from excessive heat is essential to successful photography. Beware the closed, sun-baked automobile!

7. Photographing Immature Stages

The immature stages of moths and butterflies are fascinating in their forms, their behaviors, their cryptic design and the structures they build. Once an immature is found, the challenge lies in setting up, composing, and lighting the subject to portray the desired details. Egg detail should not be overlooked—ova come in varied shapes and they demonstrate amazingly intricate surface textures that can be recorded without a scanning electron microscope!

Success is possible in the field. Upon discovering a larva, if you are addicted to rearing immatures, wait before you pluck it and pop it

Figure 2-2. Adjustable jig holding a sprig of black gum (Nyssa sylvatica).

into a box to take it home. Consider photographing its resting position, or its degree of camouflage in relation to its background, even if a full view is not possible.

To put a supporting branchlet into better position for a photograph, use a supporting jig made from two spring clothespins connected by a 20–25 cm (8"–10") piece of 2 mm (1/8") solid-core solder: attach the solder to the clothespin using a jam-fit into a 2 mm (1/8") hole near the end of a handle (as in Figure 6-20, Chapter 6, Section 9). The jig is used to stabilize one branch against another, or twist a branchlet to view the underside. An aluminum tent pole (available at sporting goods stores), with a spring clothespin inserted into the open end, pushed into the ground, can be used instead to stabilize a small branch. Flash is frequently necessary in these situations. Back at home, a tripod can be very useful.

A solid, level table with a smooth surface provides a stable base for the subject and simplifies control of composition, depth of field, and motion. A holding jig with two small alligator clamps mounted with ball joints and wing-nuts on a heavy base permits positioning the twig or leaf bearing your subject at any desired angle (Figure 2-2). This device is available as "dual helping hand" from MCM Electronics (Appendix L). Leaves or paper on which ova have been laid can be pinned to a piece of styrofoam glued to a small stick.

A similarly versatile holder can be made from a small, squat bottle filled with modeling clay, into which you thrust the supporting twig or stick. Small square ink bottles that can be positioned on their bases or any of their sides are ideal (McFarland 1988). If you find yourself uncomfortably hunched over the camera finder, place the jig on a sturdy platform built to a convenient height. The tripod should be set up on the floor, not touching the table, and the shutter tripped with a cable release to minimize vibrations. Needless to say, the work area should be sheltered from electric fans and passing breezes.

Some photographers can take superb pictures with a sturdy table and chair and rock-steady hands, elbows and forehead. If you have

this talent, then the tripod, focusing rail and cable release are redundant. The necessary flash is so brief as to minimize the effects of motion.

When you use an automatically programmed camera but focus manually (this is usually the best strategy), the focusing is done with the lens aperture wide open; it stops down at the time you shoot. The apparent depth of field is much less than it will be in the actual picture, especially if you have selected "aperture priority" with a small stop. For best results, try to orient the subject so that it is largely in a plane perpendicular to the optical axis of your camera, rather than stretched out away from you. Then try to focus so that about one-third of your subject is in front of the plane of focus, and two-thirds beyond it.

If you need an image ratio larger than that available with your macro lens and extension tubes, a bellows designed to retain the automated features of your camera system can increase the magnification ratio by a factor of three or four. The device employs a calibrated rack so a specific setting can be easily duplicated. The bellows can be mounted on the focusing rail, if it does not include one of its own. Alternatively, add a teleconverter between the camera lens and the body.

The bellows, however, is somewhat unwieldy, and is being supplanted by some of the specialized macro lenses now available, such as from Olympus and Nikon. They make full use of available light for focusing, and all automatic operations are retained.

For still further increase in the image ratio, you can use a wide-angle lens plus a "reversing ring" (Blakker 1987, Karp 1966, Shaw 1984). The ring attaches the lens to the camera backwards, and a 24 mm focal length wide-angle lens produces an image ratio of about 8:1. This sacrifices automatic aperture control, but metered flash exposure control is retained. This approach can produce very good photographs of eggs and hatchling larvae.

For the most accurate egg photography, McFarland (1971a, b) recommends a camera adapter attached to a high quality binocular zoom dissecting microscope. The eggs can be photographed on the substrate on which they were laid, or teased loose and placed in desired positions on a small square of "Perspex" (a methyl methacrylate sheet similar to Plexiglas, marketed also as "Sintra Board PVC,"

at artists' supply stores). Use a color giving desirable contrast to the subject. McFarland recommends using morning low-angle direct sunlight as the only light source, to record accurately the surface sheen and sculpturing of the eggs.

Lighting preferences vary. For synchronized metered flash, try to work in an area where light is subdued or can be reduced at the time you take the picture. A moveable drafting or gooseneck lamp of moderate intensity that can be turned on with a foot switch for focusing and turned off before shooting works well. (If you are working with black and white film this lamp can be left on during shooting.) But too much light or heat may set into motion a larva that normally rests or feeds beneath the leaf. A hand-held flash coupled to the camera by a remote cord allows you to vary the position of the flash (with or without a diffuser) to bring out surface details you wish to emphasize. Here, again, side-lighting provided by flash bounced off a white or foil-covered card can reduce unwanted shadows. This card can be conveniently held in the same solder-and-clothespin jig described above for stabilizing small branches in the field.

Always take several shots of one subject, particularly if it is rare or unlikely to be encountered again. Shoot from varied angles and with variation of the lighting angle. Film is easier to acquire than cooperative subjects. Keep notes on the conditions of each shot, so you can refine your technique.

Sunlight or skylight provides fine illumination, but it limits your hours of work and breezes will complicate your efforts. In addition, small dark larvae can quickly overheat in direct sunlight while you are composing your picture. If they cannot scurry around beneath the leaf fast enough, they can be cooked to death in a few seconds.

When you photograph immatures primarily to illustrate their external features, a contrasting background improves your picture. A good approach is to hang on the wall behind your subject a piece of poster cardboard kept flat by a thin plywood backing. The cardboard should be of a contrasting but subdued color—pale blue, medium gray, sage green. A black backing will show off white hairs on larvae, or white wing fringes. The backing should be at sufficient distance (at least 10–20 cm [4–8"]) so that its surface, and any shadows from the subject, will be out of focus. Shadows will fall outside the frame of the picture if a remote flash is placed at an angle well above or to the

side of the camera's optical axis.

Certain caterpillars do not like the limelight and immediately crawl out of sight. Larvae can be chilled in the refrigerator (not the freezer), but they warm up very rapidly and become active again when removed. Such creatures are most easily photographed in the premolt stage, on the substrate they have prepared for the purpose—do not try to move them!

Alternatively, deprive an active larva of food for a few hours, then provide it with a sprig of foodplant set up in a holder, ready for photography. You can take the subject in the act of feeding, and when it has finished you can take it in its resting posture (McFarland 1988). Simultaneously set up a number of subjects, and then work back and forth among them as they exhibit desired behavior.

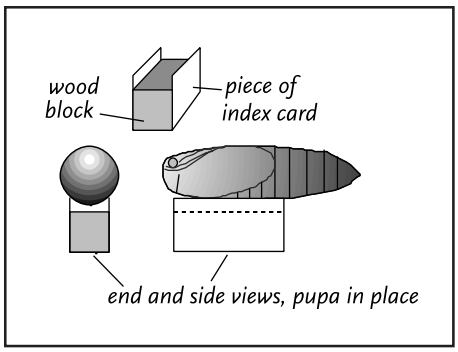

Figure 2–3. Pupa Holder.

To photograph the underside of a slug caterpillar (lycaenid, limacodid), or demonstrate the spots on the under surface of a catocaline larva, place it on a clean pane of glass or a microscope slide. A larger lens aperture, for reduced depth of field, can help to blur out any supporting structures. Be sure the light source is used from an angle that will not reflect into the lens.

If a pupa you are photographing is twitchy and rolls about, elevate it on a small home-made pupa holder (Figure 2–3). The holder consists of a small rectangular piece of balsa wood with slightly taller pieces of index card glued to opposite sides. Varied sizes can be made to accommodate different sized pupae. The pupa is placed so that the mobile abdominal segments are suspended beyond the end of the holder (N. McFarland, pers. comm.). Use a holder small enough so that it will be obscured by the pupa when photographed from above.

A small metric ruler included in photographs of immatures obviates the need to keep track of image ratios. It is an asset scientifically, but a drawback aesthetically. If you make a pair of exposures, one with the metric scale in place and another without, you will be able to know precise dimensions for the unmarked photograph.

8. Special Situations

One lepidopterist found an artful way to increase his chances of photographing sphingids feeding at deep-throated flowers (Loftin 1992). He dosed the depths of some moonflowers with a small amount of sugar solution (8–10%), using a syringe with a long needle, and the visiting hawkmoths stayed at each flower long enough for several photos.

To photograph Catocala feeding at bait, try wearing a head-lamp to free your hands for manual focusing, using an on-camera flash to take the picture. A preset small aperture reduces the need for critical focusing in the dim light. To avoid spooking the moth, cover the head-lamp lens with yellow or orange cellophane or even a thin yellow paper napkin. Cancel "preflash," if your camera has it. It may startle the moth.

Certain features of adult moths and butterflies are difficult to photograph except indoors, using captured or reared living adults. Avoid using "pinched" or dead specimens set up on vegetation to recreate a "natural" composition. The unnatural position of wings, legs, tongue, or antennae quickly expose the ruse, and many otherwise excellent publications have been marred by such bogus photographs—even to the point of portraying a luna moth nectaring at a zinnia! Yet artfully designed setups that do not take liberties with biology can be justifiably employed. Examples are ventral wing surfaces you have not been able to photograph in the field (as opposed to upper surfaces of species that never rest with wings open!), emergence sequences and moth resting postures.

When distribution surveys are being made to develop regional lists and atlases, some would-be participants are reluctant to capture and retain voucher specimens to verify the occurrence of each species. With many species, particularly larger butterflies (but much less often, moths) it may be possible to substitute a "voucher photograph" made of a captured, live specimen through an empty compact disk container from which the black holder has been removed. The specimen is placed with the wings either spread or folded over the back as the "filling" in this transparent plastic sandwich. The photograph will not be a work of art, but with full data inscribed it will be an adequate record. Try to choose an angle that does not pick up reflections. The butterfly can be released unharmed.

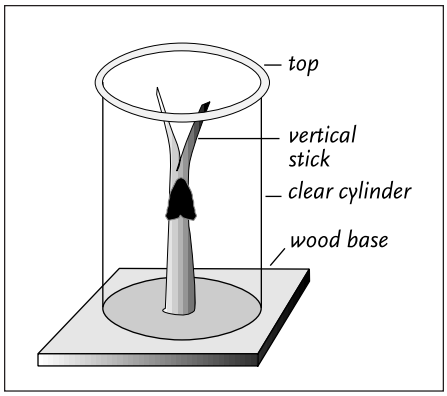

Figure 2–4. Loose-box adaptation.

The principles of lighting, backgrounds, and camera management described above for immatures apply equally here. An assortment of perching surfaces that can be positioned and supported firmly at appropriate angles are helpful: flowers, twigs with and without foliage, or chunks of bark broad enough to fill the frame of the picture. Cool surroundings and dim light are an advantage, and best of all is to work in a small, uncluttered room (does any lepidopterist have an uncluttered room?) with few things for a vagrant moth to hide behind. An overactive insect can be sedated within a small cage placed inside an iced cooler, but be sure it has time, on removal, to arrange its legs and wings into a natural posture or it will appear dead.

Dickson (1992) recommended an "insect loose-box," modified here (Figure 2–4). It is made as an open-bottomed, transparent cylinder from the plastic sheeting used for temporary storm windows, such as "Glass Clear," 10 mil thickness. The plastic is rolled into a cylinder about a foot tall, five or six inches in diameter and joined with household cement. The top, cemented on, should be cut a trifle larger than the cylinder to make it easy to grasp with one hand. The bottom is left open. A piece of wood — part of a small vertical branch — serves as a perch. This perch is cut to fit within the loose-box, but with some clearance at the sides and top, and fixed upright onto a plywood base somewhat larger than the bottom of the loose-box. Place the box over the perch on the work table and introduce the insect under the edge of the box. If the bug alights on the inside of the box, flick it off repeatedly from the outside until it comes to rest on the perch. Then focus the camera through the side of the box, remove the box, and shoot. Moths that would normally choose to rest beneath leaf litter often will not cooperate, but for others this approach is very helpful.

9. Photographing Spread Specimens

A spread specimen, or a group of spread specimens, may seem an

easy subject to photograph, but avoiding unsightly background shadows is difficult. The most beautiful results, as found in the *Moths of North America* series (Hodges 1971), involve use of a backlighted, glass-supported wax pinning surface, multiangled overhead lights, and a long working distance so that the outermost specimens do not seem to be leaning away from the center. Excessive backlighting results in translucent, unnatural images. Such a setup is hardly within reach of the amateur.

A single specimen can be pinned in the top of a pedestal made from a 15 cm (6") piece of drinking straw mounted vertically in a wooden base. Plug the top of the straw with modeling clay (Figure 2–5). Background shadows will be negligible, if a fairly wide aperture (for shallow depth of field) is employed.

To photograph the underside of a spread specimen, past advice has often been to remove and reverse the pin to show the underside. This operation is difficult and can damage the specimen. A safer and simpler approach is to take a hexagonal or square nut for an 8 mm ($5/16$") machine screw, pack the hole with modeling clay, and imbed the head of the pin, upside down, in the center of the clay (the data label should be temporarily removed). This makes a sufficiently stable base for small to medium specimens, and the nut is obscured by the specimen's wings. Larger nuts can be used for heavier specimens (Winter 1980). Or try the pedestal described above. A ring-flash is very useful when you are photographing pinned specimens; shadows are diminished, and demonstration of surface texture is rarely necessary.

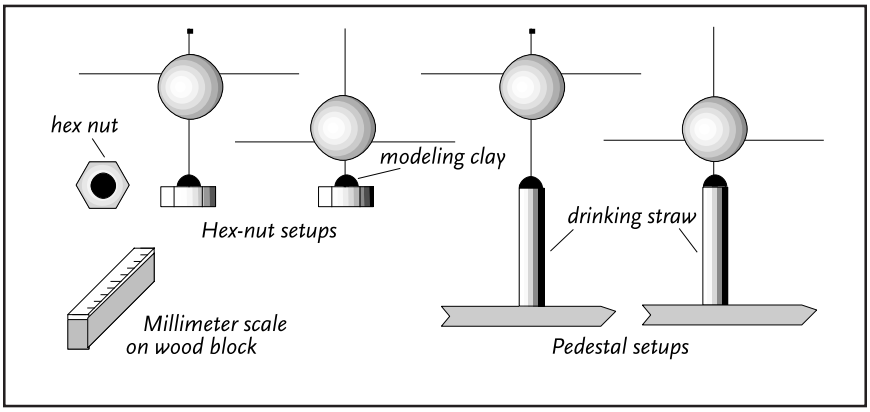

Figure 2–5. Aids for photographing single spread specimens.

To photograph a specimen alongside a millimeter scale for accurate size reference, cut a narrow strip, including the numbers, from a thin plastic ruler, and cement it to a small wood block the same size as the scale strip (Figure 2–5). The thickness of the block should equal the height of the specimen's wing plane above the supporting base.

Ultraviolet photography, to depict the ultraviolet-reflecting areas of butterfly wings, is beyond the scope of this manual.

10. Videography

"Where the 35 mm camera may excel with image quality, it is the camcorder that captures the quality of life." (Ebner 1993). Cinematography to record the motion and actions of butterflies and moths on film is, for the amateur, an expensive and almost archaic technique. Increasingly sophisticated and decreasingly expensive camcorders make videography the preferred medium for recording the behavior of Lepidoptera in all their developmental stages. The introduction of a small color videoscreen as the viewfinder for the camcorder has simplified monitoring the scene as you record it, as well as enabling you to keep track of your subject if it bolts. You do not have your eye "glued" to the back of the camera. Video light requirements are far lower than for photography on film. Current camcorders will focus from infinity down to a few centimeters, producing image ratios several times natural size. The tapes can be viewed on the finder screen or your regular TV screen, and copied to standard videotape format for viewing via an ordinary VCR. Four video formats are currently available. Their characteristics are compared in Table 2–2.

Features vary with the product. A zoom lens is standard, usually up to 8x (the limit for hand holding). Power zoom is best for steadiness, and wireless remote control is ideal for tripod work with immatures and emergences. A color viewfinder simplifies locating the subject, which can be obscure in a black and white finder. Automatic focus is standard, but manual override is essential. Automatic power shutoff conserves battery power. You will need three or four fully charged rechargeable batteries for a day's work. Also available are automatic date and time recording (these mar a picture and are best not used—keep a notebook), fade to black and white, audio and video input/output jacks, image stabilization, and digital effects.

Camcorder Format	Weight	Picture detail	Playback	Capacity
Std VHS	6–10 lbs	Good	Std VHS VCR	2 hours
Super VHS	≈ 6 lbs	Much better	Via camcorder thru TV, or special Super VHS VCR	30 mins
8 mm	≈ 3 lbs	Slightly better than standard	Via camcorder thru TV, or special 8 mm VCR	30 mins to 2 hrs
Hi 8	≈ 3 lbs	Best	Via camcorder thru TV, or special Hi 8 VCR	30 mins to 2 hrs

Table 2–2. Comparison of video formats.

Cables for connecting to a TV set are usually included. Accessory lenses screw onto the front lens of the camera for close-ups of ova, detailed anatomy, and even wing scaling.

Videomaker Magazine (P. O. Box 469026, Escondido, CA 92046) gives information on brands and equipment, and articles on videography. Because of the cost of a camcorder, it is wise to begin by borrowing or renting one for a weekend or two to become familiar with the equipment. You can then make a prudent choice.

Do not compromise on tape quality. Use only brand name high grade videocasettes, such as Fuji, Maxell, Scotch, Sony, TDK and others.

Tape outdoors whenever possible. Bright sunshine generally produces the most natural video color. However, during cloudy spells pierids and other brightly colored forms can be taped with less chance of burnout or excessive glare. Many butterflies are easiest to photograph before 10 am or after 3 pm, when nectaring is more leisurely and lighting is softer. Avoid taping on excessively windy days. Try to zoom slowly from wide angle to close up, until the subject fills more than half the screen, if possible—this can call for an approach to no more than two feet. Focus manually: it is faster and more accurate. The principles of approaching a moth or butterfly (Chapter 1, Section 3) apply equally to videography.

With practice it is possible to carry the camcorder on a tripod and set it in place in just a few seconds; this avoids camera motion. A monopod is somewhat faster but also less steady. For very close work a tripod or tabletop pod is essential.

Know your camera. Its operation must be learned and practiced so that in the field, where timing is critical, recording is automatic and instinctive. Take your Lepidoptera shots and your family shots on separate tapes. Once a tape is fully recorded, break off the plastic tab on the cassette to prevent accidental erasure. Log your shots as you take them, each subject, location, and date. Be sure to include the counter numbers for easy retrieval of a particular shot.

Camcording subjects are infinite: the hatching of eggs and the elevation of larval tubercles and other fleshy projections; the molting process; details of ambulation; chrysalis formation, "seat-belt" construction and the legerdemain of withdrawing the cremaster from the larval skin and thrusting the hooks into the silk pad; shelter and cocoon construction; emergence, splicing the two halves of the tongue together, expanding the wings; courtship, assembling, mating, oviposition, etc. Slow-motion recording of some of these events allows portrayal of processes that occur too rapidly for the naked eye to appreciate.

There are some disadvantages to video recording. Video image quality does not yet match photographic standards, but advancing digital technology will certainly improve this. Editing requires special equipment and new skills to be effective. Transferring from the original field source to a first or second generation tape for editing, titling, voicing, etc., results in substantially reduced image quality. Despite these drawbacks, and the cost of the equipment, there is no more satisfying way to record behavior in action.

11. Data and Record Keeping

Memory can be a fleeting thing, and immediately labeling any collection of photographs enhances their value and significance. Record keeping will be covered in detail in Chapter 3, but any slide or print of Lepidoptera should bear the date and the locality where the picture was taken and the name of the photographer. These elements should be written in ink, since they are unequivocal. The name of the insect should also be included, initially in pencil if identity is uncer-

tain. Also indicate the roll of negatives from which a print was made, to facilitate making future reprints.

A photo log, completed when shots are taken and listing every frame, is essential when you keep photographic records of immature stages, especially when you rear unknown larvae. Remember to record the image ratio, so that actual size of a larva can be calculated. The log should indicate also the rearing lot-number of each subject (see Chapter 6 on rearing), so it will be possible to link accurately this year's photographs with next year's emergences—the emerged adult may be your only means of identifying the immatures collected in the wild as eggs, larvae or pupae.

12. Organizing a Photograph Collection

As you develop skill photographing Lepidoptera, consider how you wish to organize your collection of slides and prints so that any particular one or group can be located easily. Images can be organized geographically, or chronologically, or by general subjects such as behaviors (nectaring, basking, mating, etc.), life cycles, camouflage, or predation, etc., but perhaps the best approach is to organize them in numerical order following a taxonomic checklist of Lepidoptera. They will then be sequenced in the manner of most museum or extensive private collections of specimens or photographs. For Lepidoptera of the U.S. and Canada, *Check List of Lepidoptera of America North of Mexico* (Hodges 1983) is the standard, covering both butterflies and moths. For butterflies alone, *A Catalogue/ Checklist of the Butterflies of America North of Mexico* (Miller & Brown 1981) with its supplement (Ferris 1989), is appropriate. Unfortunately, such lists are not available for all areas.

An excellent retrieval system can be set up with a database using a home computer. The photographs are indexed initially by "MONA No." or "M&B No." and then subindexed by geography, date, general subjects, and so on. If you wish to retrieve a series of photographs on parasitoids and predators, or on cryptic forms and coloration, the computer can provide a list of slides by their MONA numbers.

There are many types of slide-viewers used to sort a group of slides. Handheld viewers are slow and clumsy. Backlighted tabletop viewers allow examination of a few dozen slides at a time, but restacking the slides to return them to the file is tedious. An exceed-

ingly useful device is the "Slide-a-Lite." With a sweep of your hand from left to right you spread out a stack of thirteen slides (a third of a roll) on a light-bar for examination. Another sweep of your hand, right to left, restacks the slides in their original order for return to the file.

To facilitate examining individual slides without projecting them, use an inexpensive loupe mounted on eyeglasses or on a forehead frame.

Slides are returned from processors in various boxes and plastic sleeves that are not particularly suitable for permanent filing. One good system uses drawers designed to hold parallel rows of slides arranged in such order as you find most useful (see "NegaFile Systems," Appendix L). These provide 200 cm ($78^1/_2$ "row-inches") of storage space per drawer (1400–1570 slides, depending on whether the slides are plastic or cardboard mounted). There is enough "headroom" (a total inside depth of about 6.5 cm [$2^1/_2$"]) to accommodate small index tabs to indicate the beginning of a number series, a family or a genus. A moveable backing block (or you can use a block of foam) keeps the slides upright in an incompletely filled row. The drawer is enclosed in a case that protects the slides from light and dust.

When estimating your storage needs, calculate on the basis of 18 plastic mounted or 20 cardboard mounted slides per row-inch.

A plastic "Keep Box" (hardware stores) with headroom for index tabs, provides 76 cm (30") of row (for 540–600 slides) and is dustproof but not lightproof.

Two useful cardboard boxes (Mason Box Co., Appendix L) hold 8 cm ($3\ ^3/_4$ row-inches: 60–65 slides) and 22 cm ($8\ ^3/_4$ row-inches: 160–175 slides), respectively. The headroom is zero, so you will need to make flexible index tabs.

For easy access, some photographers store their best images in three-ring binders using plastic pages that display 20 slides at once. Use only polypropylene or polyethylene "archival quality" pages (see "Century" and "Print File," Appendix L). Absolutely avoid polyvinyl chloride (PVC) pages, which over time can seriously damage slides or negatives. These display pages can be viewed on light tables made to handle two or more sheets at once. You can also use such pages to submit photos for publication.

Prints can be damaged (early fading or staining) by albums made from high acid materials and sometimes by self-stick pages. The 3M self-stick album is allegedly satisfactory. Negatives are best stored at moderate temperatures and low humidity—not in a cellar or attic (Wilhelm 1993).

Some of us find that not all our photographs are perfect, but developing a ruthless attitude towards suboptimal shots can be difficult. A good rule of thumb is: if you have nothing better to illustrate this species, this developmental stage, this particular behavior, keep it until you have taken a better one—then retire the inferior shot to a "cull file" or preferably to the wastebasket. A corollary to this is: keep a list of shots that need to be upgraded, so that you can keep them in mind each time you go out in the field or decide whether or not to rear or photograph a species again.

In the course of time, you may accumulate a considerable portfolio of pictures of flowers from which your subject has just departed. These are not without value, as they can be used to demonstrate the anatomical details of utilized nectar flowers that might otherwise have been obscured by the butterfly!

Whatever your main interest in the observation and study of Lepidoptera may be, give serious consideration to enriching it with photography. With outstanding cameras available at reasonable cost, even a beginner can capture superb images of natural history and behavior on film, thereby making significant contributions to the science. While this chapter has placed considerable emphasis on techniques, these are merely adjunct to your main mission. Insect macrophotography is primarily based on your being there, ready and willing to record what you see, even getting down and getting dirty, or lying belly-up for some shot that no one else has had the imagination or patience to capture.

Reviewed and augmented by
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Noel McFarland, Michael C. Thomas,
and David L. Wagner

For further practical information on cameras and photographing Lepidoptera, consult:

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Chapter 3.

RECORDS

Perhaps the easiest thing to gloss over, defer, or omit entirely is keeping field records of your activities with Lepidoptera. It can be tempting to keep a "life list" but go no further. Such a list without pertinent and significant notes is like an index or a table of contents with no book attached. Many lepidopterists can look at a photograph or a specimen and its data label and immediately recall much of the circumstances when it was taken—but only for a time. Not only do field records make a very satisfying personal diary, but they are an indispensable resource when you wish to share information with others, whether informally or in print. It is extremely frustrating to begin to write about a particular experience and discover that the details have become blurred or mixed with other times and places. The result is an anecdotal account that may or may not contain fragments of fact. Also, timely field notes are the backbone of unassailable data on photographs or on the pin labels of collected specimens.

Minimum needs

- *Spiral-bound pocket notebook and pencil*

1. Field Records

How and where should field notes be made? They should be made when the observations are made, not deferred to a later time or date, and should contain the elements of information included in Figure 3–1. A spiral bound shirt-pocket notebook is a convenient medium for field notes. They can be quite cryptic and abbreviated, but should be updated whenever there is a break in activity or before moving to a new site. They should then be "fleshed out" later in the day, by transcribing them into a permanent logbook or other chronological record, perhaps on a computer.

Another reliable method uses "temporary trip reports" (Figure 3–1) held on a small clipboard on the front seat of your vehicle. A separate sheet is made out as you leave each site visited, and the information is transferred that night into a permanent bound field notebook.

You can use a pocket minirecorder for dictating notes as you walk. These notes also must be transcribed later, since relocating a particu-

lar item on a tape can be quite tedious. They easily fall victim to procrastination or accidental erasure, however. A developing technology, the all-digital recorder, makes it possible to record voice notes, then transfer them electronically to your computer. It is very compact, and very expensive; but in time the price may come down, so it sounds promising for field notes. Some observers use a lap top computer in the field to record data directly.

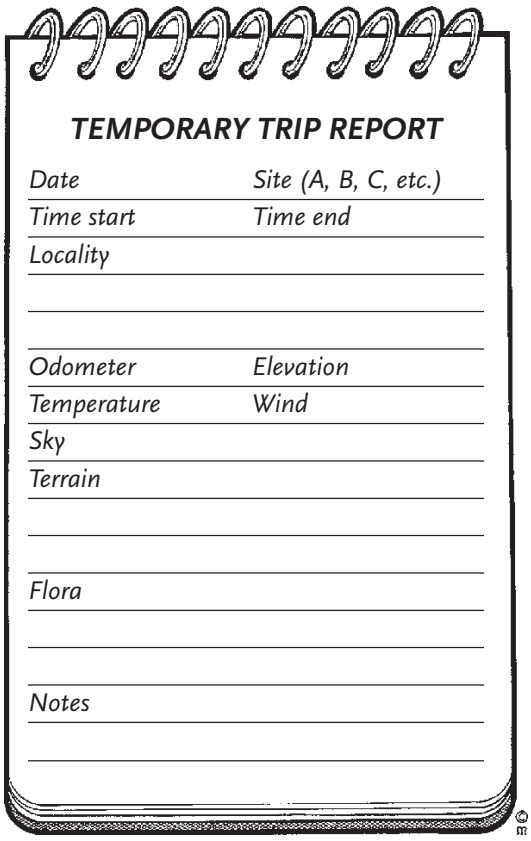

Figure 3–1. Temporary trip report.

If you store your field and rearing notes in a computer, be sure to make the usual backup copies. Saving paper copies is also prudent. There is concern that within only a decade or two it will be difficult to find software capable of reading your current disks!

What should be included in field notes? Detailed location, time, and date are basic, and weather observations—sun, wind, temperature—are significant. Location should be stated in terms that can be identified on standard current maps refined as to approximate direction and distance from the center of a municipality, a route intersection, or intersection of a highway and a named watercourse, for example (but see Chapter 8–3 concerning impermanence of highway numbers). Altitude (in meters) should be included in mountainous areas. A note as to habitat, in varied terrain, can be helpful—"upland pasture" or "edge of marsh," etc. Comments as to behavior or activity, and the times of day or night that particular species were active, are also of interest. Date format should be unambiguous, so that September 5th is not mistaken for May 9th. A form such as "5 Sep 94" or "5 IX 94" is

unmistakable (but the use of four digits for the year is better). The month should never be stated in Arabic numerals.

Because prints and slides may not be available for labeling for days or weeks after they are taken, it is vital that all the necessary information be recorded promptly and accurately (including camera settings and film types, if your photographs are likely to be placed in contests or in publications). The source and circumstances of collection of livestock that may be reared far away from their points of origin must also be recorded to avoid later uncertainties. And in the case of preserved specimens that may not be pinned and spread for many months after a collecting trip, this information is essential for the production of proper pin labels. Temporary labels are acceptable for temporary purposes, such as on field-pinned specimens; location and date are the minimum data for such labels.

Paper record systems, long in use, include a logbook for the chronological, geographic and physical details of your field experiences. A bound lined ledger is far more useful, and more durable, than a shoebox filled with miscellaneous notes scribbled on the backs of envelopes. The log is often augmented by card files grouping observations by single species, by behaviors (such as patrolling, hilltopping, carrion feeding, or defensive maneuvers), and by subjects such as parasitoids, predation, cryptic coloration, etc. Grey (1964), who spent his life studying variation and subspeciation among *Speyeria,* devised a detailed but relatively uncomplicated filing-card system for correlating his field observations. His sequence of primary subdivisions was "Species," "States and Provinces," and "Counties." Under counties, separate cards gave topographical sites, such as a drainage system on a particular mountain or range, for a particular day's collecting, along with numbers, sex ratios, other *Speyeria* species at the same site, altitude, ecological details, and comments on variation within a species on that occasion. Clues strategically located at the corners of the cards allowed for quick manual sorting for any desired parameter.

While many will continue to devise card files suited to their own personal pursuits, it is likely that more will be advancing to computers for compiling and correlating observations from their records.

The long-term goal is to develop a coherent and chronological system of record-keeping, organized so that you are able to retrieve

easily the particular bits of information you want, and so that others can make sense of your material.

2. Computerized Records

If you have discovered the acrobatics that a home computer can perform for you in filing and retrieving your observations and records, you are likely to choose this medium over paper.

Nothing will be said here about specific brands of computers or particular software—such statements are out of date before they are printed. In general, you will want a computer that you can understand and be using in minutes, not weeks; that responds rapidly to your commands; that has more megabytes of storage and RAM than you can dream of needing; and that communicates rapidly with a speedy and legible printer. Software should include a versatile word-processing program and a database program that can handle several thousand records in each file. ("Record," as used in this sense, means the observation of a single species or individual at a particular time and place.) The caveat about making timely backup copies of all your work on disk cannot be overemphasized, and it is handy to print paper copies of rearing results or of slide or specimen inventories to share with others who do not have a compatible computer or appropriate software available to them.

The logbook can be kept as a simple word-processor document, in which a desired subject can be located quickly by using the "find" utility. Various databases accommodate file card functions, with instant retrieval possible. Inventories can be displayed or printed out by species, date, geography, or whatever other parameters you may define at the time of input. Careful planning as to how you set up your record-keeping will enable the computer to serve you best. Too much complexity can mean slow responses. Too little foresight can result in inefficient retrieval.

Computer log entries should be on a day-to-day basis, in order to avoid loss of details. Card-file and database entries can be day-to-day, or can be deferred (particularly in the case of inventories) to the "off season," when there may be more time to verify determinations and generally "put your house in order."

Your database can be used as a resource for creating a "want/need" list—species you have not yet reared, those for which you need better

photographs or additional specimens, or those that you have thus far failed to discover. Coupled with this can be a prospective calendar of when and where to look for your desiderata. If you are planning a vacation or a business trip, you would add the special opportunities presented by the places you visit. Names of knowledgeable locals to consult can be found in the membership directory of the Lepidopterists' Society. Lepidopterists are noted for their willingness to help others with similar interests. If moths are your interest, it is worthwhile to incorporate in your calendar the dates of new and full moons. An appointment-book desk accessory, a word-processor document or a database file can accommodate these lists and can organize them chronologically and taxonomically.

Inventories deserve special mention. You may have a friend or correspondent inquire when or whether a particular species or genus occurs in areas you have studied, or what assortment of species you have recorded in a particular area or time. A properly indexed computerized inventory allows you to print out, in just a few minutes, exactly the information requested. There is no need to pore over drawers of slides or endless boxes of specimens. Indicating the destination of specimens you have transferred to others is also worthwhile, should someone wish to examine that material.

An inventory is also an asset at the time of transfer of a collection of Lepidoptera to an institution or another individual (see Chapter 13, Disposition of Collections). For this purpose following the nomenclature and taxonomic order of a standard list, such as that edited by Hodges (1983) for North America, simplifies the process. In addition to the usual what-where-when-by whom, it is good to include an indication that you do or do not have collection permit documentation in those instances where it is appropriate. Such inventories should not be initiated late in life, but should be ongoing, year after year.

Records of rearing experiments are covered under rearing (Chapter 6 and Appendix A). Day-to-day recording is essential for accurate reporting of your findings. You may eventually learn that the information you record is already well known. On the other hand, if it turns out to be new information and you have not kept records, it is an opportunity lost.

The information to be recorded on photographs has already been discussed in Chapter 2. Rearing records are covered in Appendix A.

Records for pin labels of preserved specimens are detailed in Chapter 8.

"The evil that men do lives after them.
The good is oft interred with their bones."

The unrecorded and unreported discoveries of too many excellent amateur lepidopterists have been and are continuing to be similarly interred.

Reviewed and augmented by
Mogens C. Nielsen, Jerry A. Powell,
Floyd W. Preston, and Ernest H. Williams

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Chapter 4.

IDENTIFICATION

Most of us are more satisfied when we can name the species we see, photograph, or collect. Identifications are made by comparison—with illustrations in field guides and other books, or with preserved specimens—or by consultation with colleagues with broader experience. You should be able to identify most butterflies and most medium and large moths from heavily studied areas of the world with the approaches described here, but there are many genera of medium and smaller moths for which illustrations are inadequate or unavailable, or comparison of external features uncertain. For these, dissection of genitalia gives the only reliable answer. This can be done years or decades later from a specimen.

Minimum needs

- *Field guides covering your area*

1. Books on Butterflies and Moths

Many excellent books are now available for identification of butterflies. Some are true field guides, illustrated so as to point out each species' distinctive features and designed to be pocketed in the field. The information about each species is limited but includes such things as distribution, flight periods, foodplants, habitat, distinction from similar species, etc. Other books are major texts covering details of behavior, life histories, variation in markings, and extensive consideration of subspecies. Illustrations may be color paintings of spread specimens, or photographs of live individuals taken in the wild. Many give a representative sampling of immature forms, but extensive field guides to larvae are still in the future. Scope of the various books may cover a single state, a broad but cohesive region (such as the Rocky Mountains), or part or all of a continent. In most books the illustrations are arranged by families in formal taxonomic order. In a few they are arranged by color and "look-alikes." A surprising number avoid skippers.

There are far fewer books on moths. Some cover selected members of many families (Holland 1903, Covell 1984). Others provide in-depth coverage of a single family, subfamily, or even a single genus

(Hodges 1971 and later fascicles of *The Moths of America North of Mexico* series). Appendix J provides extensive annotated lists of useful books for identifying Lepidoptera.

2. Basics of Classification

Scientific classification utilizes features of form and structure. Lepidoptera are distinguished from other insect orders by the presence of overlapping scales on the wings, most butterflies from most moths by the presence of knobbed or clubbed antennae (a few moths are exceptions), and skippers from other butterflies by the presence of hooked tips beyond the clubs. Families of Lepidoptera are scientifically determined by the status of various features such as setal (bristle) patterns and other structures of the larvae, or the mouth parts, wing venation and anatomy of genitalia of the adults.

Skippers and butterflies comprise about six percent of the Lepidoptera species in any given region of the United States. The rest are moths, which vary in size from the giant silkmoths down to midgets that can walk through the mesh of a window screen. As your interest broadens to include some of the more difficult genera (such as *Eupithecia, Euxoa, Hydriomena)* and the genera of smaller moths, you and your advisers will be using increasingly sophisticated methods to identify your subjects.

Within a species there may be great uniformity of appearance, or there may be regular polymorphism, with differences in appearance by season or by sex. An individual may show an excess of black (melanism), or absence of expected color (albinism). Sometimes normal patterns are discernible despite color differences; or the patterns themselves may vary. Such variations may appear on a random basis in some species or with predictable frequency in others. An individual may be part female, part male (a gynandromorph). Even genitalia can show considerable variation within a species. Recognizing these variants can often be quite difficult, so when in doubt, seek more experienced help. Above all, avoid becoming trapped in an extreme attitude: "If it doesn't conform, it must be a new species," or "If it doesn't conform, ignore it."

Structure

The lepidopteran body, in all stages but the egg, consists of three

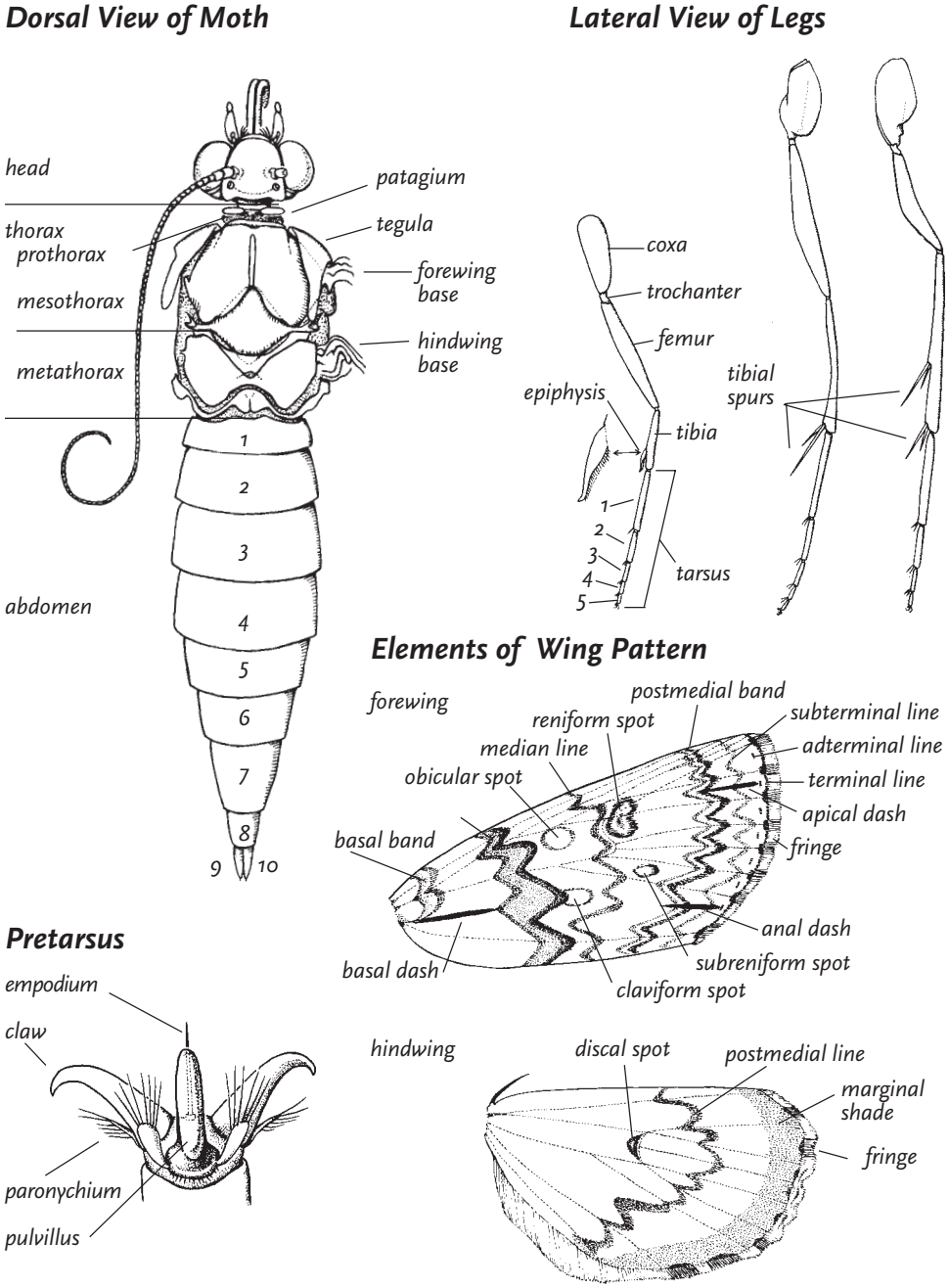


Figure 4–1. Anatomic features, continued. Drawings by Elaine R. (Snyder) Hodges from The Moths of America North of Mexico, *fasc. 21 (Sphingoidea), reproduced courtesy of the artist and the Wedge Entomological Research Foundation.*

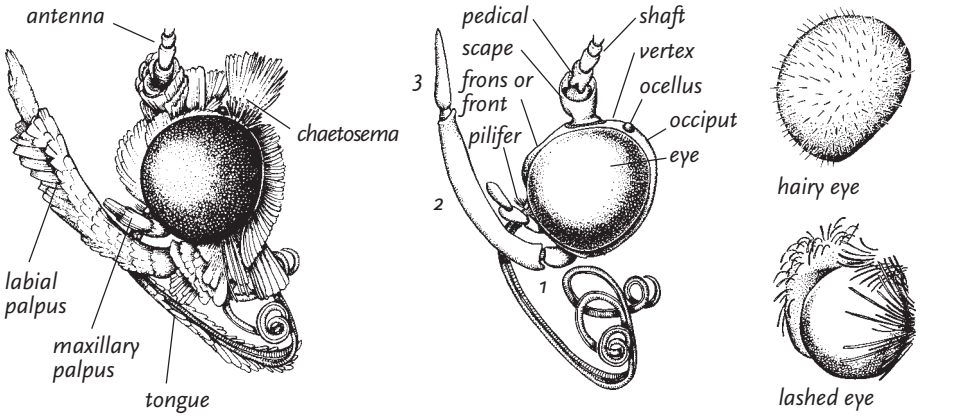
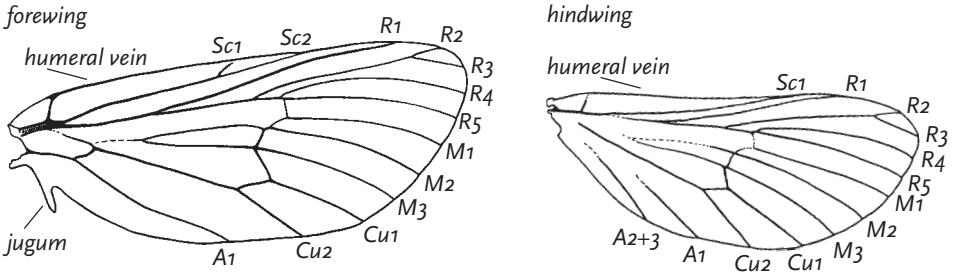


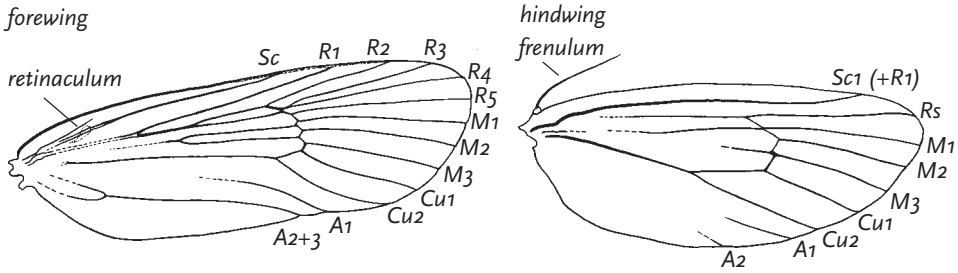
Figure 4–2. Anatomic features. Drawings by Elaine R. (Snyder) Hodges from The Moths of America North of Mexico, *fasc. 21 (*Sphingoidea*), reproduced courtesy of the artist and the Wedge Entomological Research Foundation.*

Complete Venation

Primitive Moth



Specialized Moth



Major Areas of Wings

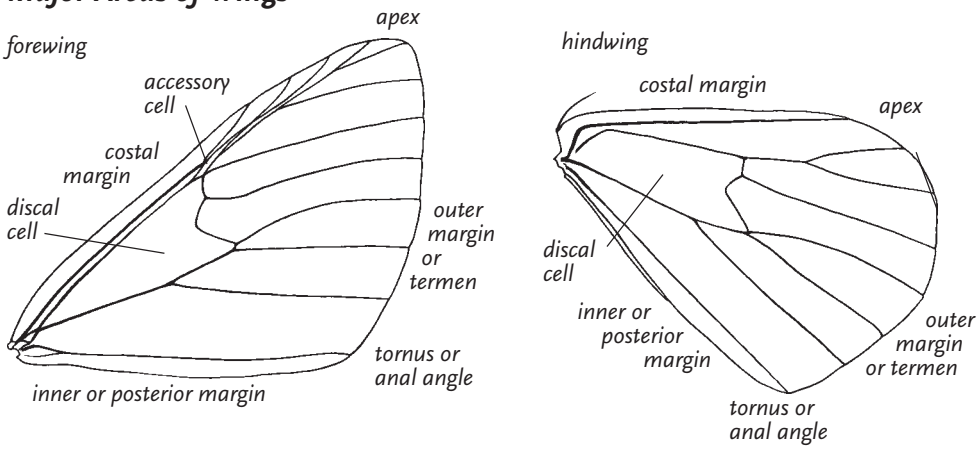


Figure 4–3. Anatomic features, continued. Drawings by Elaine R. (Snyder) Hodges from The Moths of America North of Mexico, *fasc. 21 (*Sphingoidea*), reproduced courtesy of the artist and the Wedge Entomological Research Foundation.*

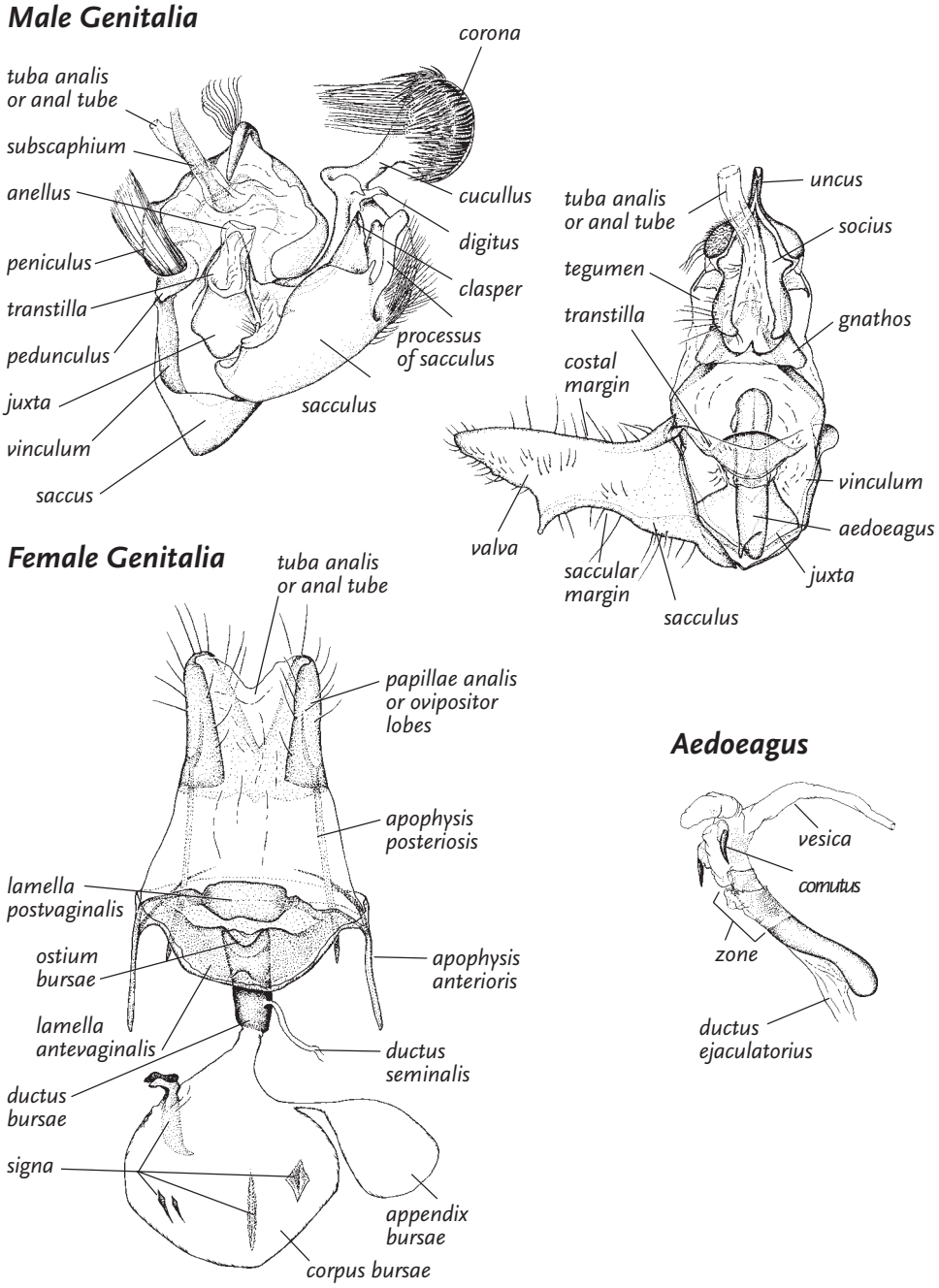


Figure 4–4. Anatomic features, continued. Drawings by Elaine R. (Snyder) Hodges from The Moths of America North of Mexico, *fasc. 21 (*Sphingoidea*), reproduced courtesy of the artist and the Wedge Entomological Research Foundation.*

regions (Figure 4–1): head, thorax (three segments), and abdomen (ten segments). Structures on the head include paired antennae which may be hairlike (filiform), saw-toothed (serrate), comblike (pectinate), feathery (plumose or pinnate), and clubbed, knobbed or hooked (Figure 4–2). Male and female antennae may differ significantly, with those of the male being more elaborate. One function of the antennae is to detect the presence of chemicals (pheromones) emitted by the opposite sex. A pair of large, compound eyes can have a diagnostic shape or placement (because of a wide or narrow forehead or "front"). The eyes may have "lashes" (overhanging scales) or even "hairy eyeballs" (fine bristles protruding from between the facets of the hadenine or pantheine noctuid compound eye, or in *Glaucopsyche* blues). Compound eyes detect motion, form, and a wide range of color, as well as ultraviolet. A pair of ocelli (minute single-faceted eyes) is often present, but commonly obscured by the scales and hairs (vestiture) covering the head. These distinguish only light vs. darkness. The labial palpi (palps) are two short, segmented appendages arising at the sides of the mouth. They are often helpful in identification. The tongue (proboscis), double-barreled and held coiled beneath the head, is an extensible sipping-straw through which the insect feeds.

The thorax bears three pairs of legs and two pairs of wings. The segments of the legs are named with terms used in the lower limbs of vertebrates: coxa, trochanter, femur, tibia, and tarsus. The legs may be decorated with scales and hairs, and armed with spines, spurs and combs. The last tarsal segment bears the tarsal claws. Chemical sensors on the tarsi aid in identifying the appropriate foodplant for ovipositing.

The wings are attached to the sides of the thorax, with the forewings generally larger and overlapping the hindwings. In some primitive moths a small lobe (the jugum) projecting from the posterior edge of the forewing engages the front edge of the hindwing and helps to keep the wings together in flight. This structure is absent in more recently evolved families, and many of these moths have, instead, a frenulum—one or more bristles on the front edge of the hindwing that fit into a small retaining structure (retinaculum) on the under-surface of the forewing. In most moths there is a single bristle in males, but three in females. The structure and placement of the

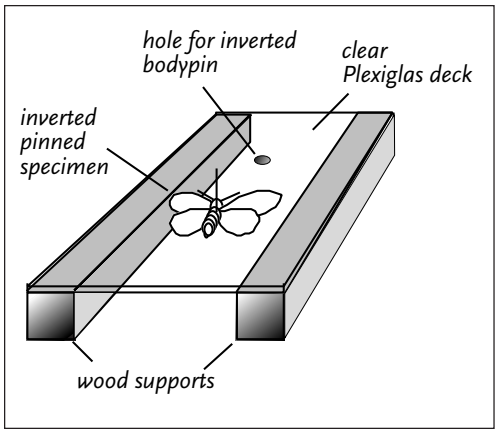


Figure 4–3. Stage for examining wing veins. (Redrawn from Rings et al. 1992.)

bristles and retinaculum is helpful in classifying some moths.

The wing veins are laid out in a pattern that is traceable, with variations, throughout the entire order Lepidoptera. The basic vein patterns are shown in Figure 4–3. The terminology uses words from the vertebrate skeleton.

The costa is the leading edge of a wing. A subcostal vein (Sc) is the foremost vein in each wing. Parts of the radial veins (as many as five, R1–5), form the anterior margin of a roughly triangular veinless area called the discal cell. There are three medial veins (M1–3), and the absence of the proximal part of at least M2 contributes to the veinless area of the cell. The medial veins also form the cell's outer boundary. There are two cubital veins (Cu1–2); Cu1 defines the posterior margin of the cell. Of the three anal veins (A1–3), A1 is usually absent and A3 may be.

In the hindwing the subcostal and first radial veins are usually fused into a single vein (Sc + R1), and R2–5 are fused and renamed Rs. In many butterfly families (and in lasiocampid moths) there is, at the base of the hindwing, a humeral vein that strengthens that corner of the wing and keeps it from dislocating above the forewing.

Venation patterns for all the families can be found in Borror et al. (1989). Common (1990), and Scoble (1992) are the most up to date resources.

Wing venation can be studied easily only in spread specimens, most clearly on the under surface. Veins can be made more visible by carefully brushing scales from the under surface of the wing, or, without damaging the specimen, by wetting the wings with ethyl alcohol or rubbing alcohol so that they become translucent. Rings et al. (1992) recommended use of a homemade supporting stage (Figure 4–3): drill a small hole in the center of a 10 cm (4") square of clear Plexiglas and support opposite edges on blocks of wood about 2 x 2 x 10 cm ($^3/_4$ x $^3/_4$ x 4"). If you drill two holes you can do side-by-side comparisons. Place the specimen upside down on the stage, with the

head of the pin through the hole, then wet the wings with the solvent to visualize the veins. Let the solvent evaporate before removing the specimen, to avoid scale loss. Without using the solvent, this is a convenient support for examining the frenulum and details of the anatomy of the legs and palpi. *Caution:* antennae that were not properly spread and project above the plane of the wings are likely to be damaged if the specimen is inverted on the stage.

Androconial patches are groups of specialized scales on the wings of males of some butterfly species. They can be broad areas on some satyrids; small forewing discal area patches on lycaenids; tight forewing patches on "branded skippers;" in an overlapping costal fold on the forewing of some pyrgine skippers; and an oval black bulge on vein Cu2 of the hind wing of danaines—these can be of considerable help in classification. Other groups, especially pierids, may have patterns formed by scales which reflect ultraviolet light, invisible to our eyes. The techniques for demonstrating these UV-reflectant areas are not covered in this book.

The noctuids and related families have paired organs of hearing, the tympanal organ, on the last segment of the thorax. A taut membrane transmits sound waves to two or three nerve cells which are more sensitive to changes in loudness of a sound than to its specific wave length, but they can detect the supersonic echolocation frequencies of foraging bats. The location of the tympanum and the direction in which the tympanic membrane faces is a distinction between families. In geometrids and pyralids these organs are on the first abdominal segment.

The abdomen is divided into 10 segments that may be readily distinguished or may be obscured by scales. Tufts of scales on the dorsal surface of certain abdominal segments or fringes on terminal segments can be distinctive identifying characters, as are rows of spines on the edges of segments.

The abdomen houses the digestive tract and the organs of reproduction. The genitalia in the female arise from segments 8 and 9, and in the male from segments 9 and 10. A small spiracular opening on each side of all abdominal segments except the latter-most leads to the tracheae, treelike branching hollow tubes which conduct oxygen to the internal organs and body fluids.

The abdomens of many caterpillars bear pairs of fleshy walking

appendages (prolegs). In most families these are present on segments 3 through 6, with a 5th pair on the last segment. These are fleshy, rather than hard-surfaced like the thoracic true legs, and usually bear tiny hooks, the crochets, that assist them in maintaining a tight grip on the foodplant when walking or resting. The true legs, while necessary for walking, are also used for manipulating the foodplant while feeding. The only larvae readily confused with those of Lepidoptera are those of sawflies (Hymenoptera) with prolegs on abdominal segments 2 through 8, that often rest with the last few segments curled around on themselves. The prolegs of sawflies have no crochets.

In some families certain body parts are nonfunctional (the wings in the female gypsy moth and other lymantriids, many high-altitude and island genera, and in both sexes of the commercial silk moth); modified (the forelegs in nymphalids—hence the term "brush footed" butterflies—danaines, satyrids and male lycaenids); or rudimentary or absent (the proboscis in the saturniids, lymantriids, cossids, oenochromine geometrids and many winter moths; or the wings in some female geometrids, bagworm moths, and tussock moths). In the case of wingless females, identification may be easiest from a pair taken while copulating, with the male identifiable; by caging a freshly emerged female to see what males she attracts; or by rearing ova to an identifiable larval stage or to mature males. The possibility of using these approaches should be kept in mind while the specimen in hand is alive.

Details of the anatomy of the genitalia of Lepidoptera (Figure 4-4) are extremely useful in defining and distinguishing among species, genera, and other taxa. Genitalia are generally examined on prepared specimens (Chapter 8). Preparing genitalia is the subject of Chapter 9.

It follows from the above discussion that some species of Lepidoptera can be identified solely on the basis of field observations, and that taking good photographs can increase your opportunities for identifying many more. Yet there will remain many species which can be identified only by collecting and carefully examining them. The level at which you pursue and enjoy your lepidopteral activities is a personal choice.

3. Names

Just as our own names and the names of other people are essential in our interactions with society, so the names of the Lepidoptera we see are essential in any search for further information about them, and in communicating with colleagues. Every species that has been formally described has a scientific name, and some species have common names. There are good arguments as to which is better for everyday use, and which involves less ambiguity.

Common names for a particular species vary from country to country, and in a large country like the U.S.A., one name may apply to different species in different areas, or one species may have different names in different areas. The Lepidopterists' Society and the Xerces Society compiled a butterfly common names list for America north of Mexico, plus Hawaii (Miller 1992). The North American Butterfly Association produced a considerably revised list (NABA 1995); a list for Mexico is expected. Many of the larger and commoner moth species have long had common names, and in England common names have been much more widely used for smaller moths. Some of the British names have been adopted when the same species occurs in North America, but that practice has been irregular. The author of the recent eastern moth field guide (Covell 1984) was required by the publisher to provide common names for all species (exceptions were allowed for some of the "microlepidoptera"). Additional lists are the Entomological Society of America list of common names of insects (Sutherland 1989) and the Quebec list correlating French, Latin, and English names (Anonymous 1964).

Scientific names of Lepidoptera, as of all organisms, are Latinized binomials, with a capitalized generic name and a lower case species name, always with both names underlined or italicized. This, in some writings, is followed by the name of a person, the author who originally described and named the species. The year of the description is often appended. If the species is now placed in a genus different from that in which it was originally described, the author's name is enclosed in parentheses.

Scientific names have the advantage that they are in a single language accepted the world over. One would think that therefore they would be stable. Such is often not the case. Detailed anatomical examination and immature stages not previously known often reveal

relationships indicating that a species belongs in a different genus. Examination of old literature may show that an earlier author had already described the species under a different name or sex, and the earlier name usually is given priority. The generic name *Eubaphe,* for example, that long resided in Arctiidae, was found to have been used earlier in Geometridae and was restored to that family, while the species it previously embraced were consigned to the arctiid genus *Holomelina.*

Strict rules govern the assignment, publication, and use of scientific names. These can be found in the *International Code of Zoological Nomenclature,* 3rd ed., 1985, published by the International Trust for Zoological Nomenclature, London, England.

Does this matter for the amateur? As you leaf through the butterfly books in an effort to make an identification, you may see that in *The Butterfly Book* (Holland 1898) the bronze copper is called *Chrysophanus thoe.* In the Opler and Malikul (1992) field guide it is called *Lycaena hyllus*. In between it was listed in the checklist of Miller and Brown (1981) as *Hyllolycaena hyllus*. Comparable examples are found among the moths. As you go from one book to another, you may be able to recognise a particular species only by its picture. Fortunately, most species have less checkered pasts. The recent propensity of some publishers to economize on pages by indexing species only as subheadings under the genus can make it very difficult to look up a species that has a changed generic name.

The concept of subspecies is confusing. A subspecies can be defined as "a subdivision of a species, usually a geographic race. The different subspecies of a species are usually not sharply differentiated and intergrade with one another and are capable of interbreeding." (Borror et al. 1989). To be scientifically acceptable *(International Code of Zoological Nomenclature)* and have legal standing (Endangered Species Act), a subspecies must be named. Subspecies are actually arbitrary designations, since the worker applying a name to a segregated population chooses which characteristics of the population to include and which to ignore. Subspeciation occurs most commonly where there is geographic isolation of areas of suitable habitat from one another, as is common in the mountains of the American West. Because the appearance of a subspecies may be variable and not absolutely distinctive, some people suggest that the amateur can

most easily assign a specimen to a subspecies on the basis of where it came from, rather than how it looks.

4. Identification from Books

As you strive to become familiar with the various families of moths and butterflies, it is good to settle down with a field guide or other text for your area and study the introductory material. This provides an understanding of the anatomy and metamorphosis of Lepidoptera, and familiarity with the terminology used to describe wing shapes and patterns. Some of the guides include end-papers depicting the general forms and relative sizes of the different families. Browse through the illustrations, and you can quickly learn to recognize a butterfly as swallowtail, sulphur, or satyr, or a moth as sphinx, geometer or plume moth, and so on. Get a feel for absolute and relative sizes: large for swallowtails and the monarch; medium large for fritillaries and some angle-wings; medium for many whites, sulphurs, and satyrs; medium small for some crescents; small for most blues and coppers—not to mention the far greater ranges of size and silhouette found in moths. Notice information on resting positions—wings over back, wings outspread, forewings up with hindwings spread, head down or head up; behavior while feeding—quiet, fluttering, hovering; and flight character—gliding, steady, zig-zag, frenetic dashing. All these details will help you to focus your search. The existence of mimicry, where a species gains an advantage by resembling a different unpalatable species, complicates the process of identification but creates additional interest. For extensive consideration of mimicry, see Wickler (1968).

When comparison with an illustration seems successful, check the text information as to habitat, range, and flight period. If these do not coincide with your observations, be suspicious and try again. But a poor "fit" is not absolutely impossible. Some species wander widely now and then, or are carried long distances by the wind. An occasional individual may fail to maintain diapause and emerge out of its usual season. A specimen found fluttering or dead in a parking lot may have traveled hundreds of miles on a radiator grill (and this possibility should be dutifully recorded). A larva which pupates on a vehicle or a piece of freight may emerge in another country or another continent. But these "off base" possibilities should not be accepted

immediately at face value—better to consult with someone more expert about them. The vast number of your records will be of individuals flying in a reasonable place at a reasonable time, and with the exception of unusual aberrants and hybrids, you should be able to find the butterflies illustrated. Even aberrations can usually be related to normally colored specimens without too much stretch of the imagination.

Moths are more of a challenge—there are about twenty times as many families and ten times as many species of moths as there are of butterflies—but this is no reason to be intimidated. The major families of "macro" moths are easily recognized—saturniids, sphingids, arctiids, notodonts, geometrids, and noctuids—as are many of the smaller families and many of the "micro" moths. As you try to identify smaller or less showy species of moths, you may not be able to find illustrations and may need to resort to verbal descriptions in texts. This calls for familiarity with the terminology of wing patterns and shapes, and an understanding of the often unusual terms for describing colors, some of which are not in dictionaries! And here the "zebra dilemma" can raise its head: what appears to be a white wing with black patterning may be described in the books as a black wing with white patterning.

Books which rely heavily on verbal descriptions often provide keys to help in identifying a species. These give you pairs of "either-or" choices based on anatomical features. Each choice may lead to another pair of choices until the final identification is reached. If the key covers a large number of species, and you are uncertain as to whether that palp is "porrect" or "appressed", your search may grind to a halt. But it can be fun to try, and it frequently pays off. Running a known species backwards through a key can be a useful training exercise. Useful keys are found in Common (1990), Scoble (1992) and Forbes (1923, 1948, 1954, 1960). Being able to place a moth in its proper family or subfamily greatly simplifies identification. The various fascicles of *The Moths of America North of Mexico* (1971 and later) also provide such keys. There are also good keys for North American families in Borror et al. (1989) and for the world in Brues et al. (1954), although the latter are somewhat outdated.

For every moth or butterfly with a scientific name there is a publication containing the original description of that species. This can

prove useful in confirming an identification (although older descriptions often have little detail), for its historical interest, or when you are writing up an article reporting your observations on the insect. Locating this description can be a challenge, but a rational procedure to follow was outlined by Donahue and Donahue (1989). This is reproduced in Appendix C.

5. Comparison with Other Collections

When you have exhausted your own resouces and skills in your effort to identify a species (or before reaching the point of exhaustion), it is time to seek out and consult someone of greater experience. A more experienced lepidopterist can often guide you in identifying a species that puzzles you. If your specimen happens to be atypical, he or she may be able to point out a more typical example. Sessions of this sort can increase your skill and confidence in identifying species, so that eventually you in turn may be consulted.

If you do not happen to know anyone nearby who is a more accomplished lepidopterist, helpful people can often be located through the membership directory of The Lepidopterists' Society. The listings of the interests of each member can give a clue as to who might be helpful. If they are unable personally to assist you, they may be able to tell you where in the area there are photographers, or reference or synoptic collections accessible to the public, so that you may seek out other material for comparison. Lepidopterists are commonly happy to share their knowledge with you. Access to museum collections is discussed in Chapter 11. Remember, however, that a collector's concepts of species often are only 90% correct and that even museum collections may have shortcomings in accuracy or in complete coverage of all the species in a given area. The most valuable adviser recognizes the limitations of his knowledge and admits it.

6. Help from Specialists

At times your informal adviser will also be stumped, and you will want to consider seeking professional help, usually from a specialist at a museum collection. First, you should narrow down the field so that you know at least which family or subfamily you are dealing with, if not the genus. Photographs should be good quality, and specimens

should be in good condition and well spread—out of focus slides and badly worn or rubbed specimens may not be worth pursuing—and all must have full data (see Chapters 3 and 8). But do not simply knock on the door with a box in your hand and expect to be welcomed. A preliminary contact, by mail or phone, is advisable, stating the problem and how far you have narrowed it down, and asking whether help can be offered. An introduction from a colleague who is already acquainted with the museum personnel can help to open the door.

Some experts will help you out of sheer kindness. If you are a collector, it is appropriate to offer some of your extra specimens to the museum collection—photographers should offer copies of slides. And some will assist in return for specified monetary compensation, either to them or to their institutions. It is well to clarify such conditions in advance.

In well studied families, such as geometrids and noctuids, finding a new species would be rare in the eastern U.S., but in the west many remain to be discovered, and in some of the families of micro moths 50–90% of the species are undescribed. Opportunities abound for valuable contributions by amateurs, and professionals are ready and willing to advise and assist serious amateur lepidopterists.

All this is not as difficult as it may sound, and if you become deeply involved with Lepidoptera, either on a personal basis or as a participant in faunal surveys, knowing someone at a good museum collection can be a great help.

7. Identification of Immatures

Eggs (ova) come in numerous shapes and forms, often distinctive for particular families. They may be spherical, hemispheric, shaped like a disk, a dumbell, a loaf, a spindle. The exterior surface may be smooth or intricately sculptured, textured or ribbed, glossy or matte, bare or adorned with hairs. They may be laid on end, on edge, or lying down. They may be deposited singly, stacked with several on top of one another, lined up in a row, in a single-layered patch or several layers deep, between buds, within blossoms, on a leaf tip, a leaf edge, on the upper surface, the under surface, in a cuff around a stem of carefully selected diameter, on bark, bare or covered with body scales from the female, within crevices in bark. While they are most commonly laid on the larval foodplant, in some instances they

are laid on debris in the immediate vicinity, or the female may eject them as she flies over an area where the foodplant is growing. Each species employs its own combination of characteristics and behaviors. Books often give details of egg form and placement in their species descriptions, but for many species details on placement in the wild are still unknown. General comments about the ova of specific groups are found in many of the more detailed texts, but there is no overall key for identifying ova. One German-language work (Döring 1955) deals solely with butterfly eggs and has over 2500 drawings.

Identification of viable eggs is not necessarily difficult, however. Those found by chance in the wild are usually on the proper foodplant and can often be reared out to a recognizable stage or to an identifiable adult. Those collected by following an ovipositing identifiable female are thereby known. And those laid in captivity by captured females are likewise identifiable (but their foodplants may remain unknown if not recorded in the books). The difficult eggs are those found within live traps, or on walls about outdoor lights, with no identifiable female or foodplant. Rearing or identifying these eggs can be a major accomplishment, and very edifying.

Larvae must be looked at in terms of appearance and behavior—what do they look like, how do they move about and feed, and what, if anything, do they build?

The head may be smooth, granular, hairy, spined, patterned or plain, distinct from the body (as the "door-knob" heads of skipper larvae) or buried out of sight beneath the anterior thorax (as in lycaenids). It may blend into the cryptic design or the body, or contrast with it.

The thorax and abdomen may be of one color, or striped, banded, spotted, dotted, or decorated in myriad intricate patterns and colors. The surface may be smooth, granular, or furnished with warts, tubercles, fleshy filaments, horns, branching spines, stinging spines, hairs, fringes, in various combinations of colors, lengths and distribution. The assortment is diagnostic for each species, particularly as to physical structures, but in some species there may be several recognized color variations. Changes in color and form can vary strikingly from one instar to the next, making it seem that someone switched your caterpillars overnight. These changes are consistent for the species. But a feature that is characteristic of a particular family, such

as the anal horn on sphingids, may also occur in an unrelated species (the notodont *Pheosia rimosa*, for example).

Prolegs are useful in grouping larvae. The grasping structures on the prolegs, the crochets, are important for grouping species into families. Crochets are often visible with a hand lens. Five pairs of prolegs, on abdominal segments 3 through 6, and on segment 10, are basic, and the caterpillar ambulates by moving the pairs forward one or more at a time, from back to front. Geometrids, in contrast, usually have the first three pairs of prolegs absent. When they move, they grip with the thoracic legs, then release the prolegs and move the hind end of the body forward in a looping pattern—hence the common names "loopers," "measuring worms," or "inch worms." Yet all loopers are not geometrids. Plusiine noctuids have lost the prolegs on abdominal segments 3 and 4 and progress in a similar "semilooper" fashion. Catocaline noctuids, including the underwings, retain all the prolegs, but, especially when young, they race about in a very active looping fashion. In some notodonts the anal claspers are modified into long filaments. These are not used for walking but for making the profile of the larva less wormlike. In the lycaenid butterflies and the limacodid moths the prolegs are barely identifiable and the larva glides along like a slug. Leaf miners may have few or no prolegs.

Feeding behaviors vary. Some larvae feed on the under surface of the leaf, some on the upper. Some mine between the two surfaces, leaving characteristic burrows. Some chew neatly at the edge of the leaf, wasting nothing, while others chop and drop large fragments as they feed. Many are borers within stems, roots, pods, or even hard wood. Some feed on dead organic matter, and a few are carnivorous on aphids, ant larvae, or other caterpillars. Some rest right where they feed, while others retire to a place where they can hide or blend with their surroundings. Feeding habits, if recorded in the reference books, can help you identify larvae.

Most caterpillars can produce silk, and those that construct shelters by pulling and sewing leaves together thereby give clues to their identity. These features are sometimes included in species descriptions.

A useful shortcut when you are trying to identify a larva is to start with the foodplant. Some texts (such as those of Forbes 1923, 1948,

1954, 1960) include an index of foodplants, and if you are certain what your caterpillar is feeding on, then a small or medium number of possibilities is presented. The verbal description of one of those species may very well match the creature at hand. The two-volume index by Tietz (1972) is designed solely to relate "macrolepidoptera" larvae to foodplants. The Lepidoptera as well as the plants are indexed by common names and by species names. The insect species index includes all the references (up to 1950) to life history descriptions (with a notation as to what stages are illustrated), and a list of all the recorded foodplants. The author of the volumes was unable to check the validity of the information in the references, and some statements have subsequently been found to be invalid.

Pupae are distinctive to varying degrees, both in their configuration and in the place they are formed. Many butterfly pupae are naked and suspended from the posterior end. Papilionid and pierid chrysalids have in addition a silk "seat-belt" about the middle. Lycaenids pupate bare or among a few bits of leaf sewed together on the ground. Moths are more variable. Some pupate within shelters or galleries they make on or in the foodplant, and some in carefully constructed silk cocoons secured to twigs. Others attach to the enshrouding leaf and fall with it to the ground in autumn, or spin their cocoons among leaves on the ground. Hairy larvae may incorporate the body hairs in the cocoon (arctiids, lymantriids). A great many moth larvae pupate near or beneath the surface of the soil, and these are occasionally dug up during gardening.

The features of the adult insect are partly outlined in the pupal shell, particularly the eyes, tongue, antennae, legs, and wings. The tongue case in some sphingid pupae stands out from the rest of the body, like a jug handle. The length of these structures, relative to one another, can often give general or specific clues to identity.

Having made appropriate observations on an immature stage, how can you best make use of them for identification? Many of the books mentioned in Appendix J give selected illustrations of ova, larvae, or pupae. Especially see old volumes by Packard, and by Barnes and McDunnough, if you can find them (Appendix J). Others include keys for tracking down identities, and many are detailed in their species descriptions. The Peterson guide to caterpillars (Wright 1993) has very useful material, but the number of species is limited. Tuskes

et al. (1996) shows last instars of all U.S. and Canadian silk moths. Stehr's *Immature Insects* (1987) is comprehensive and detailed, but it seldom reaches down to the species level and has none of the conveniences of a field guide. McCabe's *Atlas of Adirondack Caterpillars* (1991) gives many black-and-white illustrations and is particularly valuable for its list of 435 references picturing larvae. *The Owlet Moths of Ohio* (Rings et al. 1992) depicts many representative northeastern North American noctuid larvae. Crumb (1956) shows many noctuid larvae and Godfrey (1972) many hadenine noctuid larvae. *Eastern Forest Caterpillars* Wagner et al. (1995) gives examples of over 50 larvae representative of many groups and is excellent for general orientation. *Caterpillars of Britain and Europe* (Carter & Hargreaves 1986) is another excellent resource.

In the long run, it is a matter of gradually picking up the differences among the various groups, then looking for specific details in the references available to you. The fact that rearing may be the only solution is not a bad thing, unless a parasitoid thwarts your efforts. Photographing as you go, and then seeking a consultant, can offer salvation if the larva dies before maturing. Photographs are of little help with micros, however.

8. Sexing

Determination of the sex of adults is of interest in dimorphic species—those in which the appearances of males and females differ. It is of importance in all species if your goal is to obtain ova for rearing or if needed for taxonomic study. In moths where the antennae differ between the sexes, those of the male are heavier and more elaborate. Sometimes visible areas of androconial scales (described in Section 2) give an easy clue, as do the expandable tufts of hairs on the legs of some male noctuids, especially deltoids. The contour of the abdomen as viewed from above or below can be indicative: sides parallel in the male, convex in the female, and the female abdomen overall heavier. Visible claspers at the end of the male abdomen are an obvious difference in many species. Gentle squeezing near the end of the abdomen with forceps will often evert the claspers and make them more visible, or make the female's ovipositor protrude. The sex differences in the frenulum in moths have been noted in Section 2. In nymphalid butterflies where the abdominal differences

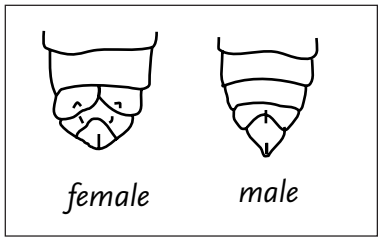


Figure 4–4. Sexing pupae. (Redrawn from Guilbot 1982)

may be obscure, the male forewing apex is often more "drawn out" than that of the female.

Pupae can be sexed in several different ways. If you have several dozen pupae of a single species and a balance with which you can take accurate weights, the heaviest will usually be females, and the lightest, males. You can do this with intact cocoons, provided that you remove any supporting sticks first. This method is useful in trying to select females for breeding stock. In moths where the sexes have markedly different sized antennae and the pupae are naked, such differences are often apparent on the pupal shell. It is a poor idea to cut cocoons open to ascertain this, however; it can reduce their survival, or give crippled wings. The configuration of the genital orifice on the under surface of the abdomen is a particularly useful distinction (Figure 4–4). In the male there is usually a narrow linear depression flanked by two rounded prominences, oval or triangular. The female generally shows two midline grooves, on the eighth and ninth segments; in addition, the anterior margins of these segments may be distorted, as if the midline had been pulled forward and inward (Guilbot 1982). A moderately strong lens and oblique lighting are helpful for distinguishing these differences on small pupae. Female *Limenitis* pupae show a ventral midline longitudinal slot on the eighth abdominal segment; the male lacks this slot (Kean & Platt 1973).

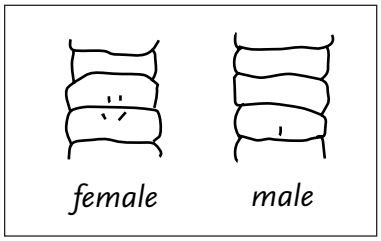


Figure 4–5. Sexing larvae. (Redrawn from Underwood 1994)

The sex of larvae can sometimes be inferred, but it is difficult to ascertain precisely. In a group of larvae that are all healthy, those that feed longest and become heaviest are more likely to be females. In some lymantriids females have an extra (6th) instar. You can sex some kinds of living larvae by examining the underside of the eighth and ninth abdominal segments with a dissecting microscope (Underwood 1994). Under direct lighting, rather than transillumination, the female shows a pair of tiny pits straddling the midline near

the border of each of these segments. The male shows a single midline pit on the ninth segment only (Figure 4–5). The following quotation explains how to restrain the larva being examined (D. L. A. Underwood, pers. comm.): "I have found that what works in holding live caterpillars varies with the species. Large *Papilio zelicaon* larvae were easily held back side down with the fingers. Smaller instars often spin enough silk to adhere upside down on a glass slide, which works well, or they may be carefully squeezed between a glass surface and another hard surface long enough to see the telltale pits. I'd recommend sexing dead larvae until one gets good at recognizing the pits; the training will save a lot of time and frustration when working with squirmy live ones. Chilling is a good idea, and you probably could use a pair of light forceps laid gently on them so that one prong lies on each side of the abdominal segments containing the sex-specific pits."

Kean & Platt (1973) indicated that in fifth instar female *Limenitis* there is on the under surface of the eighth segment, anterior to the genital pore (ninth segment), a pair of longitudinally elongated translucent spots, appearing dark when sidelighted. In the male this area is blank. Testes may be visible dorsally in a translucent-skinned larva when transilluminated.

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Chapter 5.

GARDENING FOR LEPIDOPTERA

The chief goal of gardening for butterflies is to entice them to a site where you can readily observe and enjoy them. Another is to increase the numbers of butterflies in an area. A third, perhaps better classified as a wish (but frequently expressed in correspondence to The Lepidopterists' Society) is to reverse the decline of a threatened species. A further goal is to maintain, over a period of many years, a garden that will serve as an indicator of the environmental health of the surrounding community. We shall reconsider later whether these goals can be realized.

Minimum needs

- *Soil, seeds, showers, sun*
- *A tillable patch of soil*
- *Some wind protection*
- *Nectaring plants, chosen for your area*

If, as you read this, you come to feel that it is all on a scale far beyond what you can provide or take care of — despair not. Read to become familiar with the principles involved, then consider gardening at a level that fits your resources — spatial, financial, energetic. Even in urban settings (where butterflies do indeed occur) the nectaring plants you provide in a window box or a tub, or tuck into a corner of a neighborhood vegetable plot will catch the attention of some of the more widely ranging species. Attracting a few of these butterflies in such a situation can give as much satisfaction as a list of 30 species in a suburban back yard. In a schoolyard, a sunny area that can be dedicated to a garden can be planned and have basic soil preparation performed as a unit, then be marked off in separate plots for individual classes to manage. Each class may care for only a few types of plants, but together they can make a very effective garden. As each group's efforts are seen to complement the others, sociology is learned, as well as biology.

The butterflies attracted to a garden are local species that, in their normal wanderings, come upon your garden, find a good assortment of nectaring plants, and stay a while to feed. The presence of larval foodplants increases the attractiveness of the site, and the use of the foodplants is increased by nearby availability of good nectar sources.

When starting a garden for butterflies, it is a good idea to study your local surroundings to see what species are flying at various

seasons. At the same time, notice and record what plants they use most commonly for nectaring. This survey can be done in local gardens, plant nurseries and parks, in disturbed areas and empty lots, along roadsides, on power-line and railroad rights of way, in old fields, etc. Take inventory of the plants already at the garden site and adjacent areas. The presence of a nearby larval foodplant species may make it unnecessary to add it to your garden.

Field guides and texts (Appendix J) will provide information as to flight periods, habits, habitat requirements, and larval foodplants for each species. Learn which species roam widely and use a number of larval foodplants (the "generalists"), and which have but a single larval foodplant with specialized habitat requirements. You can then use this information to shape your decisions on plant selection and planting.

If any of the specialized larval foodplants will thrive in your garden, try them. The butterflies may well come by and take advantage of them. On the other hand, it is not reasonable to plant "for" a particular species, if that species does not occur locally.

1. Physical Considerations

The space, slopes, exposures, and soil available to you are pretty much fixed. Few people can afford to make major changes, so it is only when you are selecting a new home that you can put these considerations high on your list.

Top priority for butterflies is sun, and having good sun on some part or other of your garden throughout the day is a great asset. Shelter from wind also favors butterflies. Planning windbreaks where their shadows will not be detrimental can take some foresight. Butterflies appreciate exposed bare patches where they can soak up the sun, as well as shrubby or weedy areas for nighttime roosting. For your own enjoyment, consider having some of the garden where it can be readily viewed from your desk, dining table, or kitchen sink.

If you plan to photograph butterflies in your garden, planting areas should be relatively narrow—perhaps no more than 1.2 m (4') wide—and accessible from both front and back. This makes it possible to select vantage points for best lighting, easier focusing, and avoiding your own shadow. It also simplifies care of the garden. If the planting must be wider, then strategically placed flagstones or patio blocks

will enable you to work within the broader areas. Consider a crescent shape for visibility and access. Siting the garden near a line of trees or tall shrubs gives an accessible "flight path," especially for swallowtails.

2. Garden Planning and Plant Placement

In winter, at seed catalog time, garden planning can be very expansive, but the realities of approaching spring can dictate major readjustment. Plans for making any of your lot into better gardens must consider the time and funds available for the project, as well as the resources necessary for maintenance. You should also decide ahead of time if your lot will be used primarily as butterfly habitat, primarily to meet your own aesthetic needs, or be somewhere in between. It is also worth considering whether there are any community regulations or expectations—whether your front lawn *must* be an impeccably manicured monoculture, or whether a carefree meadow mowed once a year is really perfectly acceptable—and, for that matter, whether you yourself are comfortable with certain parts of your yard left ragged and "underkempt." A butterfly garden is not a fixed combination of absolutes, but a mixture of such useful elements as you are able and willing to accommodate.

Take the time to make a manageable, long-term plan that covers gradual incorporation of the features you wish to include. Start with areas that need least alteration and can become attractive soonest, then move on to areas needing more extensive change. Also consider the possibilities for reducing the scope of your activities without leaving behind an unsightly mess, should your interest or abilities diminish in the future. Planning is time well spent. It is easier to erase with the end of a pencil than with a shovel.

Advance planning can also reduce the amount of maintenance. Keep invasive plants in check by surrounding them with beds of annuals that are dug up each year. Bordering beds with mowed turf helps to keep plants in bounds. Edging placed around flower beds protects them from grass encroachment and simplifies trimming with the lawn mower. In some situations, potted plants set into the ground can be used to provide a succession of bloom at a particular spot.

Figure 5–1 provides examples of many of the features that contrib-

ute to an effective garden. This one is for an idealized north temperate situation. Compass orientation is not rigid, and direction of prevailing fair weather winds will affect your local details. The broad undesignated area in the left bed can be used for biennials that keep moving themselves about, and for some of the coarser perennials. Carrots or broadleaf parsley can be put into any vacant triangles in the bed on the right.

Soils and drainage can be altered by standard gardening principles—adding humus, sand, altering gradients, using raised beds. That will not be gone into here. But the soil available to you affects the choice of plants that can thrive with ordinary care. Available rainfall and supplemental watering are other factors. Slow surface watering via soaker hoses is most economical, does not disturb nectaring butterflies, and does not foster fungus diseases. Group your plants according to their water requirements. Many natives are drought tolerant or seasonal in their need for water, while nursery-cultivated plants may require more regular watering.

Fertilizing is basic, and one dose of organic fertilizer worked into the soil early is much more satisfactory, and less labor and expense, than repeated doses of soluble chemical fertilizers. A bit of hen manure turned into an annual bed a few weeks before planting time can give phenomenal results. However, testing the soil for nutrients and the pH level before planting provides a good starting point. You will then know what kind of soil you have, so that it can be amended if necessary. It is a "good farming" practice.

Choice of plants should follow a few general principles. They should be suitable for your hardiness zone (many seed catalogues and gardening books show zone maps), and if plants are listed as requiring some kind of "winter protection" in your zone, decide whether you will be able to provide it. This may call for location in a sheltered corner on the south side of a house, or special mulching. Buddleia, a five-star choice among butterflies, is a case in point. It is useful even where winter-kill of the above-ground portion is routine. But in general, if a plant will require a lot of pampering to grow in your area, it is probably not worth the time and expense, and it may also be a disappointment as a nectar-producer. "Let me say to you and to myself in one breath, cultivate the tree which you have found to bear fruit in your soil" (*Thoreau*).

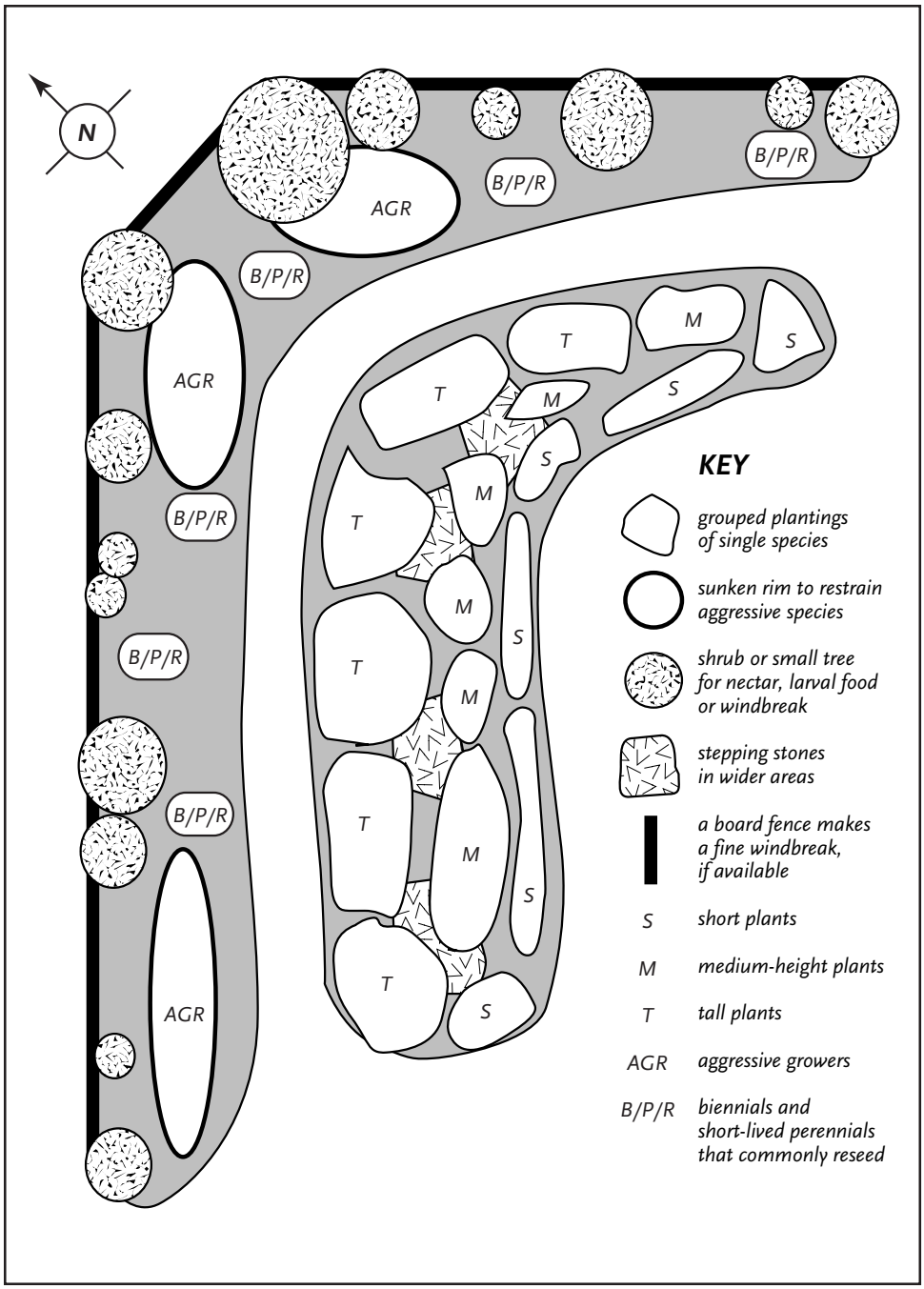

Figure 5-1. Garden planning.

Local wild plants have already proven their hardiness. If they are annual or biennial, however, they must be allowed to go to seed, and their offspring may come up all over the garden. As the garden is being prepared and rejuvenated in the spring, pot these seedlings temporarily and set them aside. After bed preparation, transplant them to reestablish or enlarge a grouping of their own kind. Seedlings of any desirable cultivated plants are handled similarly. Nursery cultivars of native plants (such as the scarlet monkey flower) can add color variety but it may take trial-and-error to discover whether butterflies patronize them.

Look for dual-use plants: screening plus larval food (wild black cherry, red cedar, black locust, hackberry, willow, pipevine); screening plus nectaring (lilac, flame vine, ninebark, unpruned privet, mockorange); nectar plus larval food (milkweeds, buttonbush, sunflowers, thistle); ground cover plus larval food (plantain, violets, pansies, Johnny-jump-ups, parsley, carrots, baby tears).

Perennials vs. annuals: perennials in the long run require more labor, but they are worth the extra effort. The earlier bloomers provide nectar before many annuals are ready; a mature plant can reach a stature most annuals cannot attain; and many of the perennials are outstanding nectaring plants. But they are harder to weed and cultivate; they often have more demanding light, water, pH and fertilizer requirements; and despite the name, they are not "live-forevers" but can die out after a few years—hence the value of recognizing seedlings and adding them periodically to the patch. The ability of some perennials to spread via lateral roots may be an asset or a disaster, something best learned from someone else's experience!

Annuals, on the other hand, give quick results the first year and often have an extended blooming period if dead flowers are removed. Some are tropical in origin and must be replanted each year, while others self-sow reliably and become permanent inhabitants. A few biennials are very useful; some of these (such as dame's rocket) may survive and rebloom an extra year or two and in the meantime self-sow and remain established.

Tender perennials: there are a few tender perennials that can be handled as tub-plants in the north if you have a place for them indoors in the winter. Among these are Pentas, Lantana, and tropical milkweed (*Asclepias curassavica*).

How should plants be placed? Massing groups of one species of flower together—rather than scattering or alternating individual sorts—is important for visual show and for the impact of fragrance in attracting butterflies readily. Butterflies tend to concentrate on a species that they find is producing nectar well, and a blooming group of plants close together gives the insect greater reward for less effort, motivation to remain at the site, and no doubt a clearer target to return to at another time. This grouping also makes photography easier—the subject is intensely preoccupied. In contrast, a scattering of single plants leads to rapid movement from one to another and less satisfaction for both the diner and the observer.

Proper staging of plants—shorter in front, taller in back—goes without saying. In the northern hemisphere, the "back" should be any side but south, to minimize shading problems. This staging gives easy access for both the butterflies and the photographer, and allows the butterfly to make a quick escape if it feels threatened. Short nectaring plants surrounded by taller vegetation are poorly patronized. When properly placed, the tall plants shelter but—at least for two-thirds of the day—do not shade the shorter ones. Rational use of fertilizer is important (except where it is inappropriate for a certain plant species). Well-fed annuals can easily overwhelm starved perennials. Plant heights should be chosen to fill appropriately the available niches in your garden.

You can even double the use of space by planting a foodplant among nectar plants—dill among tall, leggy painted sunflowers; carrots beneath blooming bachelor buttons; a mixed ground-cover of dandelions and violets in a partly shady area.

If you are planting a totally bare, new lot, or contemplating major changes to a planted lot, consider each plant in terms of its desirability and utility—what is positive, and what is useful or clearly negative. Will a potential shade tree eventually shade out your garden, without providing larval food for desirable local species? A different location or a different species may be a better choice. For screening along lot lines can you use larval foodplant trees? Can you plan an area to be left weedy to accommodate nettles, thistles, common milkweed and other useful plants that are too unrestrained to be planned into flower beds? Such questions are best considered early in the planning process.

If you are planning a garden to be enjoyed by the public, handicapped accessibility should be included.

3. Nectar-plant Selection

The quality and quantity of nectar production is of primary importance. If good nectar isn't there, the flower won't serve your customers. Among our favorite and showiest flowers, some are almost never utilized—roses, tulips, gladioli—and even many cultivars of otherwise attractive garden species may be totally ignored. "Doubles" may retain their fragrance and attactiveness to us, but the butterflies frequently prefer single blossoms. Self-sown "volunteer" offspring of ignored fancy cultivars may revert to what many people call "ugly magenta" and attract butterflies mightily. Self-sown petunias that have reverted to their more primitive color and small size are attractants superior to their robust and ruffled cousins. Some plants that are winners in one part of the country may grow nicely in other parts but show no attraction. It is for this reason that observation, to see what attracts in your area, can be most worthwhile.

The full facts of nectar production are not known, but it has been determined that, depending on the plant species, it can be affected by soil character, available soil moisture, ambient humidity and temperature, time of day, and immediately antecedent weather conditions (Ajilvsgi 1991). Changes in one or more of these parameters can raise or lower production rate, nectar composition and sugar concentration, but not enough is known to suggest how we might alter these conditions to the butterflies' advantage. The most useful plants for the butterfly gardener are those that are least fussy about soil conditions *(Abelia, Buddleia, Lantana, Rudbeckia, Sedum),* and native plants grown in soil as nearly as possible like that in which they grew in the wild.

Flower color has received considerable attention, but no solid conclusions are available. Depending on which book you read, the best colors are purple and yellow, or mauve, pink, and white, or red and orange, or blue.... Some authors (such as Rothschild & Farrell 1983) speculate on color preferences of particular species of English butterflies. The real answer for each of us probably lies in local observation and accumulated personal experience.

Flower shape is significant. Florets with nectaries only a few

millimeters from the surface can be enjoyed even by small butterflies with short tongues, whereas deep tubular flowers can require a tongue several centimeters long to exploit them. A swallowtail can enjoy the offerings of a day lily, but a lycaenid usually cannot. A flower with multiple nectar-producing florets provides a copious meal for minimal energy expenditure. A flower stem that can support the weight of a large butterfly puts the diner at ease.

Undoubtedly the most important consideration in nectar plant selection is to have productive flowers available in sequence throughout the butterfly flight season. Choose plant species for their blooming times, from early spring to frost. Within a species, note strains that are early or late bloomers and propagate these from seeds or root stock divisions to extend the species' season. Perennial phlox and joe-pye weed, as two examples, can be stretched from early July into September by judicious selection. Most of the butterfly gardening books and most seed and nursery catalogues indicate the blooming periods of the recommended plants. Interpret these in terms of your latitude and hardiness zone.

Statements in some of the butterfly gardening books indicate that a certain plant is "attractive to" a particular butterfly species, yet in some instances the plant's blooming time does not coincide with the butterfly's flight period, at least in some parts of the country. Look at such statements critically, in terms of your area, before accepting them.

4. Larval Foodplant Selection

Inclusion of larval foodplants calls for many of the considerations addressed above. Can the plant thrive on your site with only routine care? Are its growth characteristics compatible with your general planning—is there a place to put it? Will it meet the needs of butterfly species present in your area? Can it serve additional purposes—nectar source, screening, shelter?

Some excellent larval foodplants—thistles, the broadly spreading common milkweed, nettles—are quite unsuited to culture in flower beds, even though the first two are also superb nectaring plants. They need space for their spreading or haphazard growth habits where the nettles' and thistles' spiny defenses will not be a problem.

A given species of butterfly may often utilize different larval

foodplants in different parts of the country. Your field guide can tell you whether you may be able to include in your garden a foodplant for the species in which you are interested.

Providing nectar plants at least helps to support butterflies during the reproductive period of their lives, whether or not they choose also to use the larval foodplants you provide. However, foodplants in proximity to reliable nectar sources seem to be best utilized (Rothschild & Farrell 1983). The combination of both types of plants can both attract and produce more butterflies.

5. Fields and Meadows

If space, terrain, and soil characteristics allow it, maintaining an open grassy area as a field or meadow is very productive. Its grasses will support the local satyrids and hesperiine skippers, and other herbs growing among them will support sulphurs, blues, and coppers. Flowers provide additional nectaring sites. This is an ideal location for thistles, milkweed, and dogbane, as well as clovers, asters, and sheep sorrel. The floral mix will depend upon the area you are in.

Converting a lawn or old field into a self sustaining meadow involves eliminating alien species and replacing them with native grasses and forbs (Stein 1995a, b). The first stage involves successive hand-spraying with glyphosphate (available as the biodegradable herbicide "Roundup" in garden stores) alternated with raking to remove dead vegetation; tilling is unnecessary. Seed selection comes next, avoiding the many available seed mixes (often regionally designated) that include undesired alien species. This is not xenophobia. The alien species in question are often short-lived but grow rapidly and shade out developing seedlings of desired native long-lived perennials. Hand-sowing follows, in spring or autumn according to instructions accompanying the seed. It is also useful to collect seed of local native plants to include in the autumn sowing, or to start in a seedbed for subsequent setting out. You should obtain permission of landowners before collecting, and you should be aware of and comply with any laws relating to protected species. Robinson (1995, 1996) describes methods for collecting and starting wild plant seeds.

Growth the first season is disappointing, sometimes not detectable for some species. Lingering weed seeds will germinate, but can be controlled by mowing at 10–15 cm (4–6") before they have opportu-

nity to seed. Avoid watering and fertilizing; such treatment favors the weeds.

The second summer is equally discouraging; the weeds are back, needing mowing, and some alien perennials may need to be hand-pulled. But by the third summer your introductions will be well established and take over the meadow. Specific plants, if not introduced as seed, can be planted the third spring (e.g., butterfly weed).

Subsequent care usually involves controlled sectional burning, detailed in the second part of Stein's article (Stein 1995b). State and local burning permits require advance planning.

It is useful to set out foodplant trees and shrubs about the periphery of a meadow, if they are not already present.

6. Mowing and Pruning

A meadow that is not cared for in some regular fashion will fall victim to succession and soon become an "old field" and then a wood, at least in areas where forest is the natural climax vegetation. If this is not your goal, then some planning is in order. While an old field is excellent butterfly habitat, it can be difficult to keep it from progressing rapidly to the next stage of complete overgrowth.

Regular mowing is an effective way to control invading shrubs, hardwoods, and conifers, but timing is controversial. Tall coarse perennials can be prevented from flowering by mowing several times a season, and grasses and smaller plants can thereby be favored, but this procedure may curtail the flowering and seeding of the smaller plants. Rotary mowing while larvae are feeding and growing destroys livestock. Sickle-bar mowing in late September, after most flowering and larval feeding are finished, is much less damaging to forbs and Lepidoptera. Mowing alternate halves every second year is even more "humane" and still gives adequate control of invaders. Some people recommend removing the mowed cuttings after about a week (allowing lingering caterpillars time to seek shelter lower down), but this removes potentially recyclable nutrients from the field. Cuttings left lying do not seem to impede next year's growth significantly.

If your field has a generous stand of common milkweed, mowing part of it in early summer before blooming will encourage a fresh growth of tender shoots later in the summer, a boon to later generations of monarch butterflies.

To avoid trampling your meadow unnecessarily, you may wish to maintain a few random paths, the width of your lawn mower, to give access to the interior without disturbing the vegetation. A similar and wider path maintained about the periphery, between the field and any adjacent scrub or woods, will help to keep the edge vegetation from taking over the field and can also serve as a fire break.

If you have chosen to include a scattering of foodplant shrubs or trees in or around the edges of a field or your yard, alternate-year pruning is very useful. If you have young duplicates of foodplant tree species, cut one back to a foot or so from the ground every second year, during its dormant period (and continue to do this biennially). The resulting stump sprouts will produce vigorous young growth, of a height attractive to the butterflies and convenient for you. Cherries, locusts, hawthornes, elms, willows, oak saplings or scrub oaks—many species respond readily to such treatment.

Similar pruning should be applied to nettles. After *Vanessa atalanta* or *Nymphalis milberti* has pupated in early summer, cut back half the nettle patch so there will be fresh new growth for a later brood. This can significantly increase your success in local rearing. The same treatment can be used for tropical milkweed, in the ground or in containers, if it is not continually producing fresh growth of its own accord.

Our resident woodchuck demonstrated to us that judicious cutting back in early summer can make New England asters bloom well on shorter stems, with less damage from wind and rain in the autumn. This can also apply to joe-pye weed if you have strains that like to grow ten feet tall. It is important to experiment with only part of your stock so that you can learn the best time for cutting back.

The foregoing should not imply that a woodchuck is an asset. The beasts seem to prefer to eat the best nectaring plants, and they consume the monarch larvae along with the milkweed. Deciding how to reconcile woodchucks (or other varmints) and butterfly gardens can be a very important issue. It is possible to live-trap and remove woodchucks to wild land, but in some states this is illegal. If you feel inclined to distance yourself permanently from a particular woodchuck and know where its burrow is, a carbon disulfide "woodchuck bomb" (hardware stores) tossed down the chuck hole at dusk is usually effective (directions on package).

To reiterate, removing faded flowers from annuals before they begin seed production can greatly prolong their blooming periods.

7. Plant Lists

There is no need to repeat here the many good regional plant lists in the gardening books listed in Appendix J. Others are found in Wexler (1994), Swengel (1995), and lists can be obtained from Butterfly World in Florida (Appendix K). Use such lists for a start, and select species that are suitable for planting on your site. The principles already described should enable you to make good choices.

Information from many observers throughout the United States indicates that there are occasional surprises with regard to particular plants. A plant excellent in most places may attract through only a part of its growing season in one area, and not at all in another. Perennials outstandingly successful in the North may be hard to maintain farther south because of inadequate cold to induce dormancy. In some narrow zones along the Pacific coast, a spring bloomer on one side of a mountain barrier may become an autumn bloomer a few miles away on the opposite side. A butterfly strongly attracted to a particular nectar plant in one area may ignore it in another.

Two confusing pairs of plant names deserve special mention. European books cite "valerian" as an excellent nectar plant, referring to *Valeriana officinalis,* a native of northern Eurasia. The "valerian" marketed in United States seed catalogs is *Centranthus ruber,* native to the Mediterranean area. The former, also referred to as "common valerian" and "cherry-pie," is a naturalized escape in the Northeast, heavily patronized by butterflies and moths. The latter, also called "red valerian" and "Jupiter's beard" is utilized well on the Pacific coast but poorly in the East, as compared with *V. officinalis.*

The other confusing pair is *Centaurea* spp. (knap-weeds, star thistle, cornflowers: composite family) vs. *Centaurium pulchellum* (centaury: gentian family). This latter is a seldom mentioned freely self-seeding annual, naturalized from Europe to our Northeast, and visited heavily from early to midsummer.

8. Plant Sources

Seeds for cultivating annuals can be obtained through catalogs,

some of which now highlight varieties considered useful as butterfly nectar sources. Local garden centers also offer good seedlings. Many seed companies and mail order nurseries also handle perennials, both as seeds and as plants, although the latter often reach the buyer in rather stressed condition. Culture directions are regularly provided and should be followed. Local nurseries are a better source of perennials, as their stock is heavier, healthier, and tolerates transplanting better. This is particularly true for shrubs and trees.

If you have friends willing to share perennials by division or cuttings, or will give you seeds or volunteer seedlings, this is an excellent source of stock. The frequently superior qualities of these volunteers have already been mentioned. Keep a mental list of friends raising the larval foodplants you use. If you are rearing caterpillars that consume all your available fodder, these friends may be able to help you out with a "care package."

Native wild plants need special mention. In many jurisdictions there are specific regulations prohibiting digging of some or all wild plants, or even taking cuttings or collecting seeds. Local garden clubs should be able to acquaint you with current restrictions or required permits. Some nurseries are licensed to propagate wild plants and seeds. You should be sure that stock and seeds that you buy have been legally produced. Beware of bargain-priced "nursery grown" plants. They have usually been dug from the wild, grown briefly in a nursery, then potted up for sale. They may or may not survive. "Nursery propagated" plants are just that, have received extensive care and attention from the nurseryman, and they are accordingly more expensive. But they are highly likely to survive and prosper, especially if they have been reared in soil comparable to yours. By purchasing stock from such sources you are not encouraging destruction of native stocks (Ellis 1991).

Some states have regulations concerning import of all or certain plants into, or transport through, the state. Details of such regulations must be obtained locally. As indicated already, plants grown in other parts of the country may not find your soil conditions compatible and may not produce attractive nectar.

In addition, you should be aware of restricted plant species. In some states, the growing of particular species of weedy, invasive plants such as thistles and milkweed is illegal. Specific details should

be available from your county extension service.

9. Accessory Nutrition and Amenities

In addition to nectar, many butterflies derive part of their nutrition from other sources, and for some species nectar is of minor or no importance. The heliconiine butterflies can actually ingest and digest pollen, often from a narrow list of plant species, but this is a minor subject for temperate zone gardeners.

Fallen fermenting fruit appeals to many butterflies and moths (not to mention hornets). The tipplers may become temporarily unable to fly, and hence very cooperative for the photographer.

Sap-runs are very attractive, whether from branches broken by winter ice or winds, or from bark damage by burrowing insects. If you have access to a sugar maple *(Acer saccharum)*, tapping it in late winter and letting the sap flow down the bark will provide an excellent feeding station for hibernators, both moth and butterfly.

Excreta of various sorts are widely visited. Some lycaenids imbibe "honeydew" excreted by aphids and other sucking insects. Bird droppings are patronized by lycaenids and skippers. Feces of other vertebrates have a devoted following among many families of butterflies and moths.

Carrion is exploited avidly by many families. Size and character are unimportant—anything from a whole cow down to the smallest shrew, snake, or toad. Even road-kill butterflies are feasted on by their relatives.

This leads back to the subject of neatness. If you are willing to tolerate an occasional dead body (preferably of the smaller sort), a fecal deposit here or there, or the wind-falls from a fruit tree, your "butterfly pub" will be significantly enhanced.

Butterflies, males in particular, seem to have a need to obtain extra salts and amino acids, and they are often seen in the wild congregating on and drinking from damp sand. You may wish to create a puddle or seep in your garden, to meet this need (Ajilvsgi 1991). Using scrap 1-inch boards, make a frame of any desired shape, perhaps 2 feet across, and line it with heavy plastic stapled to the inside. Place the frame in a depression in the ground, in an open sunny area, so that the surrounding earth comes to or just above the top of the frame. This should then be nearly filled with coarse sand,

into which you mix table salt or rock salt, on the order of half a cup to a gallon of sand. Provide enough water to keep it constantly damp, but not so much as to allow overflow and leaching of the salt into the surrounding garden. Addition of a bit of manure or urine will enrich the mix.

Butterflies welcome areas where they can bask in the sunlight to raise their internal temperatures and make flying possible. Providing a smooth open patch of rock or soil in a sunny spot in your garden is worthwhile. The main requirements are shelter from wind and absence of nearby vegetation that would obstruct a quick departure. They are most used early and late in the day.

Butterfly feeders have their advocates, but they receive mixed reviews. They seem unattractive in the presence of good nectaring flowers, but are allegedly utilized during "blank periods" when nothing else is in bloom, if they are placed in a patch of well-patronized plants that have just finished blooming. Commercially available feeders are generally made to resemble large bright flowers and contain a reservoir to hold a sugar solution. Another type of feeder is a saucer of sugar solution raised on a pedestal. A yellow and red "nylon scrubber" draws the solution up out of the saucer and also serves as a perch for a feeding butterfly. These feeders definitely are utilized in commercial butterfly houses, but their efficacy out-of-doors is less certain. Chapter 6, on rearing, considers various sugar concentrations in detail.

There are homemade feeders that do work. One is a stake with wedges of watermelon impaled on it. A peeled overripe banana placed in a cut-off pantyhose leg and suspended from a branch will also attract fruit-feeding moths and butterflies (and hornets).

For years the name "butterfly house" has been applied to glassed-in, climate-controlled structures in which free-flying butterflies are exhibited for public viewing (see Chapter 11). Now the term is also being used for wooden boxes, perhaps considered analogous to birdhouses by under-informed catalog copywriters. The item being offered is usually a hibernation box, a hollow wooden box with slots in one side, in which hibernating adult butterflies are supposed to spend the winter. Snetsinger (1997), in a central Pennsylvania area with four resident hibernating species, monitored 40 hibernating boxes during two seasons and recorded zero use. Efforts of several Lepidop-

terists' Society members to find positive reports of butterflies using such boxes have failed. At best, they would be used only by butterflies naturally capable of hibernating in a particular climate. They would not provide effective shelter for nonhibernators, unable to survive low temperatures, despite the implications of some advertising text and illustrations.

Heal (1973) describes a hibernation box, but does not assess efficacy. It is a narrow box 1.2 m tall with a number of slots in front measuring 14 x 80 mm. It is made of unplaned lumber with a hinged top, no wood preservatives, and the "inside partly lined with bark, fastened to a wall with the bottom about 1.7 m above ground."

The best hibernating site that you can provide for adults, apart from a rickety outbuilding, is a woodpile left undisturbed. That woodpiles are used has been demonstrated again and again when a nymphalid has flown free in the living room, from wood brought indoors for the fireplace. Since hibernating butterflies tend to return to their chosen crevices, releasing such individuals out of doors in winter will probably doom them, if the ambient temperature is less than 60° F. They can, however, be placed in a screened cage (for rodent protection) in an unheated garage or cellar entry, sprayed with water occasionally, and released when the weather warms enough in spring.

10. Pest Control

Control of "pests," whether insect or plant, has long been almost a reflex activity on the part of gardeners. "If it crawls, squash (or spray) it!" "If it's a weed, yank (or spray) it!" But, to paraphrase an old aphorism, "one man's weed is a butterfly's wildflower," and that thing that crawls may be a moth or butterfly trying to make good.

In general, plants growing in a habitat for which they have evolved, with sun and moisture to meet their needs, will be healthier and less susceptible to attack by damaging insects. Fertilizer provided slowly and continuously from composts and mulches, rather than from quick-fix sprays, gives more robust, less tender plants. Handpicking of specifically unwanted bugs into a can of water with a surface layer of oil is a safe approach. Biological control of aphids by lacewings and lady bugs can be very useful, but the praying mantis, so often recommended for general garden insect control, also consumes butterflies

and cannot be welcomed. The various Hymenoptera and Diptera parasitoids that feed within Lepidoptera in their early stages we cannot control, other than by destroying their lepidopteran hosts, but moths and butterflies have managed to stay in balance with these for eons, without our interference.

Use of pyrethrin and rotenone insecticides is occasionally justifiable to save threatened individual plants. While these are potent and nonselective, with cautious control, use of weak solutions, and washing off the plants as soon as the miscreants are dead, unwanted damage can be avoided. BT *(Bacillus thuringiensis)* should not be used—it is too broadly infectious to Lepidoptera larvae.

If you have tub-plants (not currently being used for rearing) that are infested with aphids, red spider, whitefly or mealybugs, a problem particularly when they are brought indoors for the winter, there is a simple remedy. Place a cup with a tablespoon or two of ethyl acetate on the soil of the container, cover the plant with an inverted plastic bag, and tape it tightly about the sides of the container. Place the whole thing on a porch overnight (above freezing), then unwrap and bring in the next day. This is usually very effective.

Tub-plants currently in use for rearing, such as tropical milkweed (*Asclepias curassavica*) or passion vine (*Passiflora* spp.), may acquire aphid colonies tended by ants. These ants may strive to remove your larvae from the plant. To control this, set the tub on a couple of bricks in a large plastic basin. Keep several inches of water in the tub at all times, and keep the plant free of contact with any adjacent vegetation or walls.

Not all ants are detrimental. Many lycaenid species depend on ants for protection from small spiders and other predators of larvae. Chapter 6 deals extensively with the protection of larvae being reared outdoors.

Using herbicides to remove "weeds" from your lawn can rob you of opportunities. Small flowers growing in the turf are patronized avidly by many sulphurs and skippers, and some species may pay attention only to such flowers. Neatness does not pay.

Are birds an enemy of the butterfly gardener? Arguably "yes," because many birds are literally made of Lepidoptera. But birds and butterflies have coexisted far longer than people have been around to upset any equation. It seems to be possible to tolerate birds, or even

feed and post houses for them, and still have a satisfying number of butterflies.

11. *Adding, Introducing, and Releasing Lepidoptera*

Beginning gardeners often wish to stock their gardens with butterflies. As already mentioned, butterfly gardens stock themselves from their environs, without our help. It is also clear that most butterflies acquired outside, brought home, and released will exit the premises rapidly and eventually settle down, address unknown.

The advice to release reared butterflies into sheltered shrubbery after dark, so that they will "feel at home" the next morning when they resume activity, has apparently not yet been demonstrated to be valid. Newman (1967), in England, advocated acclimatizing reared butterflies in a simple mesh-covered outdoor frame cage, perhaps 1.2 m (4') square and tall enough to be placed over some growing nectar plants—enough space to allow the inmates to take short flights. A few leafy branches were placed atop the cage for shelter on hot days, and the mesh and flowers were watered with a fine spray several times a day to prevent dehydration. Additional food was made available in the form of dilute sugar syrup. After three or four days the cage was partly opened and the butterflies were allowed to depart at leisure. (He cautioned that one curious cat could destroy the whole setup!) Newman stated that using such a cage as a holding station for butterflies reared locally gave them a feeling of "belonging," so that when released they usually stayed around the garden. This approach merits trial in varied environments, perhaps with the use of wing-marking (see Chapter 7, on collecting) to help verify established residence.

The foregoing assumes that you are dealing with stock captured locally or reared from locally captured females. Exotic releases—release of individuals of a species or subspecies not occurring locally—are a significantly different matter. If you release a species not capable of reproducing and surviving locally because of absence of a suitable larval foodplant or tolerable climate, theoretically no harm will occur. However, there have been surprises, where a local foodplant did turn out to be acceptable and establishment occurred. Because of this, in many places there are laws prohibiting such exotic

releases, even of a single, reared, unmated individual. Absent such laws, exotic releases are nevertheless irresponsible.

Is it improper to release, and thereby potentially establish a strain or subspecies of a local species originating in another area? This has been done several times in England in an effort to "reverse" an extinction. But it is argued that to do it where the basic species still exists is to introduce a genetic strain that is not optimally adapted to local conditions and may thereby dilute and weaken the adaptation of the local strain. Few biologists condone such introductions. Professional advice should be sought before proceeding with any such project.

12. Goals Reviewed

Among the common questions addressed to the Lepidopterists' Society by people interested in starting a garden for butterflies are: "What should I plant in my garden?" and "How should I plant my garden?" These questions have been addressed. "Where can I buy butterflies for my garden?" That this is off the mark should now be apparent, just as buying birds for a bird feeder would be.

The query "How can I start a butterfly farm to raise endangered species?" has come up many times. The Jersey Wildlife Preservation Trust in Britain's Channel Islands, where they are rearing endangered vertebrates from many parts of the world, or the butterfly farming operations in Papua New Guinea, may be the stimulus for this question. But it has been impossible, thus far, to create a replica of the habitat of an endangered butterfly—biological requirements are inadequately understood. The concept of off-site farming cannot yet be realized. The Papua New Guinea farmers are successful because they do their farming within the natural habitat that they themselves help to protect and sustain.

"Will my butterfly garden help to reverse the decline in the numbers of butterflies in our region?" Here the answer is a qualified "yes"—with an appropriate mix of larval foodplants and nectar plants, you may effect a local increase in a few species—but not in the sense that the few extra butterflies that grow up in your garden will make a significant difference in effecting a regional recovery. Recognize that most suburban areas are landscaped with exotic shrubs, attracting mostly the "generalist" butterfly species, and those often for nectaring

only. By utilizing native plants we can, to some extent, invite displaced butterfly species back to their former habitats.

The difference will more certainly lie in the potential of your garden to open the eyes of your non-naturalist friends to the beauty of butterflies, the natural hazards they face, their specialized ecological requirements, and to the fact that the level of your own success (or lack thereof) in attracting ordinary local species takes measure of the environmental health of your surroundings. This awareness will help you to recruit advocates for action where the action is needed for habitat preservation or restoration.

Miriam Rothschild (Rothschild & Farrell 1983), in England, sums it up nicely: "But you can really abandon any idea of creating a *home* for these angelic creatures—the best you can do is provide them with a good pub. And like all popular wayside inns it must have a plentiful supply of standard drinks always on tap." She wrote this before she had broadened her horizons to enhancing whole fields for butterflies. In 1995 she wrote: "I drill in 60 species of native plants. I find that in my re-created flowering hayfields, the butterflies come in and breed seven years after planting. There is a wonderful show of browns (satyrs) and blues." (pers. comm.)

Even if the space, time, and resources you can commit to gardening for butterflies do not take you beyond the "good pub" level, you will have a satisfying and worthy accomplishment.

For your own enjoyment, keep a record of the moth and butterfly species that visit and what they are doing. As you set an increasingly tasty table, your success should grow apace.

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Chapter 6.

REARING

Rearing early stages of moths and butterflies to adulthood, whether from eggs or from larvae, can be one of the most fascinating parts of involvement with Lepidoptera. In addition, it is the area where the amateur can most readily make significant contributions to the science. Even in the best studied regions, there remain many species whose life histories are partially or totally unknown.

Minimum needs

- *"Carry-home" containers, such as Ziploc bags*
- *"Found" caterpillars*
- *Foodplant on which "finds" were feeding*
- *Simple rearing containers, from your kitchen*
- *Regular attention to the needs of your new-found friends*

The responsibilities and commitments involved in rearing, however, must not be overlooked. Rearing is time consuming, and often requires attention daily or every two or three days. An adequate supply of fresh foodplant must be easily available to you. You may need a "caterpillar-sitter" to cover a vacation, and the requirements for carrying immature livestock satisfactorily through the winter need to be anticipated.

Adequate record keeping can also be time consuming. Without record keeping you can rear handsome larvae and beautiful adults to enjoy and photograph. But if you wish also to contribute to the science of lepidopterology, as you so easily can, you will need to employ sound observation and careful recording.

Do not be put off by these commitments and responsibilities, however. The rewards outweigh them by far.

Except in moist subtropical and tropical climates, where development of Lepidoptera can continue throughout the year, most species spend part of the year in a dormant state called diapause. In temperate climates, this is further qualified as estivation (in summer) or hibernation (in winter). During diapause, development and activity are suspended, and energy stores are conserved until conditions of temperature and foodplant growth allow resumption of activity or development. Information on the timing of diapause in the species you are rearing can be found in books or through experience. The

subject is covered in detail in Section 11.

1. Collecting Immature Stages

Cherchez la femme! As you watch butterflies and day-flying moths in the field, you will often see one flitting tentatively from plant to plant—usually of a single species. Now and then she will stop, curl the tip of her abdomen down to or beneath the leaf surface, pause a moment, and then resume her fluttering progress. If you can make a good visual fix and investigate the leaf she chose, you can find and harvest the egg (leaf and all). If the female is still in sight, you may be able to continue to follow her and increase your harvest.

Serendipitous finds are frequent: eggs noted as a breeze turns a leaf over, caterpillars you find when looking for something else, the larva wandering across the pavement (if such larvae are not ground feeders, they may be searching for a pupation site), or the pupa unearthed as you work in the garden. Some larvae match so perfectly the foliage on which they feed or the bark on which they rest that they are nearly invisible. Here the "discontinuity principle" applies—you notice a shape or contour not quite right for the vegetation, and there is your caterpillar! Other tantalizing finds are the "freebies," either ova or larvae, brought in on vegetation gathered to feed other larvae. This also occurs commonly when larvae are reared outdoors in sleeves (see Section 3 regarding sleeving) placed on vegetation already harboring ova or early instars. Individuals acquired in this way should be segregated for separate rearing.

Ova and larvae may be found by specific search, in season: *Limenitis* ova on willow leaf tips; *Hemileuca* egg-rings on the twigs or stems of the foodplant, most easily found when there is a light snow cover for a background; *Danaus* ova on young milkweed leaves. Because so many species have night-feeding larvae, searching by flashlight is very rewarding, especially in the Southwest.

Eggs can be found by examining twigs about the bases of leaf-buds in winter for the cline hairstreak species with a known foodplant, in areas where females have been seen flying in the previous season (Kuzuya 1959). Look on *Limenitis* foodplants for pendant hibernacula silhouetted against snow. Clusters of black early-instar *Hemileuca* are readily visible at the tips of branches of the foodplant. You may also seek *Schinia florida* larvae mimicking the seed pods of the common

evening primrose, *Papilio polyxenes* caterpillars pruning your carrot leaves, or *P. zelicaon* on fennel. Those species of *Catocala* that, in their later instars, come down the tree to hide during the day can often be found by searching bark crevices on the trunk and the litter and vegetation about its base. It is also worth checking near lights on isolated rural buildings for eggs laid by attracted moths, especially saturniids.

Feeding damage can give good clues: oak leaves stripped to the ribs by communally feeding *Anisota*; tomato leaves ravaged by *Manduca* hornworms. Almost any leaf damaged at the edges, with the edge not yet looking brown, is worth investigating for a nearby resting or feeding larva.

Shelters give useful leads: *P. troilus* rolled in a sassafras leaf; *Vanessa cardui* in a disheveled, webbed tangle on the thistle leaf; the all-embracing communal web of *Euphydryas phaeton* on the turtlehead stalk.

Leaf-mines often indicate the presence of "microlepidoptera" larvae, and at times the form of the mine gives a clue to the identity of the miner, to genus or even species. Whether feces are retained within or discharged from the mine can be a distinguishing feature (Braun 1950). Some miners, as they grow larger, may leave the mine and feed on the surface of the leaf, either exposed or within the protection of a characteristically folded or rolled leaf, or within a portable case.

Fecal droppings are properly termed "fecula." "Frass" denotes fragments of vegetation dropped by a chewing caterpillar. Routine observation for fecula on sidewalks, driveways, and parked autos can be productive. Species that feed inside stems (borers) often leave a conspicuous pile of fecula adherent about an ejection hole in the side of the stem. The part of the plant above the hole may be wilted or dying. Hessel (1954) gave details on foodplants and feeding behavior for many species of *Papaipema*. He pointed out:

- the value of allowing the larvae of some species to mature in situ within the plant on which they are discovered;
- whether to look for the pupa within the plant or in adjacent soil;
- the hazard of a pupa being crushed when a stem you have collected dries and shrinks;
- when to expect emergence, and so on.

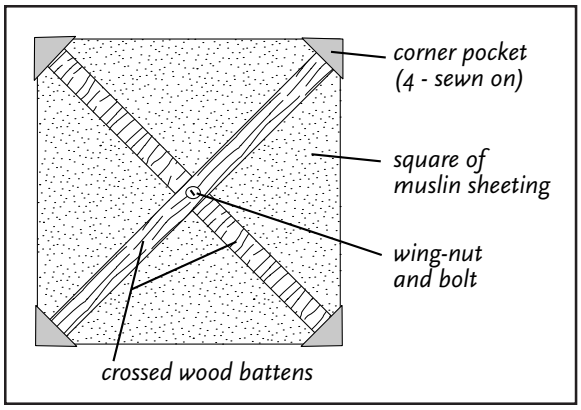


Figure 6–1. Beating frame.

Details from this article, with emendations by E. Quinter, are found in Appendix D.

"Beating" is a more aggressive approach to finding larvae. If the branch on which a larva is feeding is struck sharply with a stout stick, the larva is likely to lose its grip and fall off. Falling larvae are most easily captured by holding an inverted umbrella or a beating sheet (a piece of fabric held flat by a collapsible frame, Figure 6–1) beneath the branch. The umbrella can also (if white) serve for bouncing supplemental flash in some photographs, and to keep you dry in a shower.

Sometimes two strokes work better than one. If the first does not dislodge the caterpillar, it may set it in motion, whereupon the second stroke causes it to lose its grip and fall. A couple of vigorous whacks with a baseball bat on the trunk of a small tree can also be productive (where tree bashing is acceptable!). For collecting *Catocala* larvae, select young foodplant trees with a diameter of about 4 cm (1 1/2"), spread a couple of bed-sheets to cover the ground beneath the tree's branches, strike the trunk sharply two or three times with the ball bat, wait 10–15 seconds, and repeat; harvest larvae that have fallen onto the sheet (Gall 1990). Beating is often most productive at night when larvae are out feeding (McCabe 1991), and many species are obtainable only at night. Brower (1947) suggested a formula for calculating dates for collecting late-instar *Catocala*: onset of flight period for a particular species, minus 20–30 days, equals end of 60-day larval growth period.

"Wrapping" is a useful passive approach to collecting larvae that commute down tree trunks to hide during the day, or to seek pupation sites when fully grown. Crumpled paper, or one or two layers of burlap about 30 cm (1') wide, wrapped around the base of the trunk and tied in place with twine, can provide an attractive shelter. *Catocala* larvae are often obtained this way. But if there is a gypsy larva out-

break in progress, the method is a loser. There may be so many gypsies there will be no more room at the inn. A variation, for obtaining "microlepidoptera" larvae (Rings 1991), utilizes 3 cm (1") strips of corrugated cardboard (corrugations crosswise to the length of the strip) about 50 cm (18") long, attached firmly with push-pins to encircle a tree trunk about 15 cm (6") above the ground. Descending mature larvae find the corrugations suitable for either a pupal cell or a larval hibernaculum. Bands should be placed before 30 May, then monthly or bimonthly into September and October. Removed bands should be brought indoors, rolled into a spiral and placed inside a jar that is then covered with netting. Examine jars early in the morning for moths that have emerged during the night. Bands harvested in autumn should be placed similarly in jars, stored in the cold to allow diapause to break (diapause is discussed in Section 11), then brought indoors in April or May for emergence. In the South, adjust dates to match the flight season.

Ground-feeding larvae can often be collected from beneath a piece of sheet or canvas staked to the ground (McCabe 1991), or beneath boards or scraps of plywood laid down in almost any habitat.

Pupae of some silkworm moths can be located by searching on or near their larval foodplants. *Callosamia promethea* cocoons are spun within a rolled leaf firmly attached by silk to its twig. Brown *C. securifera* cocoons may be found dangling from silk tethers but with the leaf partly separated, on sweet bay (Peigler 1976). *C. angulifera* is untethered and soon falls to the ground. Cocoons of *Antherea polyphemus* are sometimes tethered, particularly if the larva has fed on a small-leaved tree. Similarly attached *Samia cynthia* cocoons remain tied to the ailanthus midrib of the compound leaf while the other leaflets fall, but by early winter cocoon and midrib may fall to the ground (J. Ingraham, pers. comm.). *Hyalophora cecropia* cocoons may be attached to stems and crotches not far from the ground. Since larvae frequently wander before spinning, finding a cocoon on an unusual plant is not reliable evidence that the caterpillar fed on that species.

Artificial and natural shelter sites should also be searched. The burlap bands may harbor pupae as well as larvae. Butterfly chrysalids may often be found beneath fence rails, under the bottom row of shingles (or any other ledge) on the side of a house, or behind shut-

ters. Look also beneath picnic tables and benches in campgrounds, and under the eaves of outhouses. Searching beneath flakes of bark, or inside hollows in the trunk, can be productive.

A fascinating British article from the 19th century (Greene 1857) described the search for pupae in the soil, clumps of moss, loose bark and debris about the bases of trees. Known as pupa-digging, the practice is apparently most productive about the bases of large trees in lightly grazed parkland, along borders and in open places in woods, at the edges of streams and paths, etc. In woods, Greene emphasized looking under growths of moss about the bases of large trees. "Knock off the loose bark and loosen the moss on every tree you pass." The search proves most productive in September and October, shortly after pupation, and before winter predation by rodents has taken its toll. The only tool necessary is an ordinary trowel or a mason's trowel. It is best to start by pulling moss and grass carefully away from the tree by hand, and then use the trowel to lift small divots, working from the outside towards the trunk. Most of the pupae will be on the surface or in the first 2–3 cm (1") of soil, but some as deep as 15 cm (6"). Put the vegetation and divots back and tamp them a bit, in preparation for next year (Dickson 1992).

You can also create attractive pupation sites by placing pieces of bark, boards, or sphagnum closely about the base of a promising tree or shrub.

2. Transporting Collected Livestock

If you go into the field in search of livestock to rear, or if you chance unexpectedly on an irresistible specimen, it helps if you have proper containers for safe carriage. Keeping a supply handy in your car and collecting bag is a good idea, and pocket a few empty vials when you are gardening.

Ova should be carried in capped vials, out of the sun, to reduce wilting of the leaves on which they were laid. Check the foliage to be sure you are not including an egg predator, such as a hungry lacewing or beetle larva.

Larvae must be protected from squashing and from excessive heat and moisture. This means that the foodplant on which larvae are transported should not be visibly wet, especially if larvae are very small. The interior of a car parked in the sun in the Northeast easily

reaches 135°F and in Arizona undoubtedly goes higher. Lepidoptera cannot survive this. Sheltering larval containers beneath a parked car (not on blacktop, and away from the heat radiated from the exhaust system) can often provide sufficient protection. Ice-cooled picnic coolers are also useful. Electrical coolers, available at hardware or camping equipment stores, plug into a 12V automobile outlet, draw about 3A, and reduce the temperature inside the box about 22°C (40°F) below its surroundings. Rigid containers—Band-Aid tins, snap cap plastic vials, or baby food jars—are best for small numbers of caterpillars. Tightly capped containers should have one small hole in the cover, to allow escape of excess CO_2, but some experienced rearers believe this is unnecessary. Inflated Ziploc bags, quart and gallon sizes, are also useful but are susceptible to crushing or to blowing away in the wind, and some noctuids will chew their way out. Oxygen diffuses satisfactorily through polyethylene.

You can safely carry pupae in any crush-proof container (such as a snap-top vial or a Band-Aid tin) with enough soft material to keep an unsecured pupa from being tossed about in the container as you walk. Protection from excessive heat is always important.

Adult butterflies collected for ovipositing must not be anesthetized in any way or stunned by pinching. Large-bodied species can be carried in glassine envelopes placed in a Band-Aid tin to protect them from crushing; wings must be folded up over the back. Small-bodied species travel well in vials. Moths can be anesthetized briefly for sexing; individuals placed in a cyanide jar until they are immobile will usually revive within five minutes. Volatile solvents should not be used for this purpose. Moths can be transported in envelopes or vials, depending on "foldability." Avoiding extra heat is vital, and a portable cooler is useful. Specimens stored in this way should be placed, in their envelopes, in tightly closed glass jars containing a small amount of dampened plain paper towel to prevent desiccation. If the jars are not tightly capped, water from melting ice may be drawn in by pressure changes as they cool or are transported from a higher to a lower altitude (Mattoon et al. 1971).

If the larval foodplant is not readily available where you will be rearing the caterpillars, it is important to collect, in separate bags, enough vegetation to last until your next supply-run. Adding a bit of extra water to these bags will keep the leaves fresh for one to two

weeks under refrigeration. If there is no way to maintain an adequate supply of food for the life-span of the larva, it is better to photograph the larva and leave it in the field.

As you start rearing, think small in terms of numbers of species and of individuals to begin with. And before seeking your first livestock, give some thought to the simplest containers in which to house them.

3. Accommodations for Larvae

Because most caterpillars tend to roam, some sort of restraining container is usually necessary. The form of the container is limited only by the ingenuity of the lepidopterist, and each of us tends to settle for a few favorite types. Containers can be divided into two general sorts: closed, and freely ventilated. Among the closed are plastic boxes, glass jars, flasks made from beverage containers, and wood-frame cages with glass walls. Open types include cages of various styles with screen walls, and sleeves—netting or muslin open-ended bags that are slipped over branches of the foodplant and the ends tied shut with string.

Honeycomb boxes (available from beekeeping suppliers) are made of transparent plastic with tightly fitting lids. Standard size is about 10 x 10 x 4 cm (4 x 4 x 1 1/2"); there is also a size about 7.5 x 7.5 x 3 cm (3 x 3 x 1 1/2"). These stack easily and are useful for rearing single larvae, or larger numbers of hatchlings or early instars.

Baby food jars are useful for early instars, and larger sizes of wide-mouthed glass jars (4 liter [1 gal.] pickle or mayonnaise jars scrounged from restaurants are excellent) serve to house limited numbers of large larvae.

Commercial cup- or bowl-shaped plastic containers in which foods are sold (yogurt, cottage cheese, toppings, ice cream, etc.)—one-piece molded containers, as opposed to those with the bottoms crimped in place—can be cleaned effectively and reused indefinitely. Two-liter (1/2 gal.) and 5 liter (5 qt.) ice cream pots are very handy (Figure 6–2). If the lid is not transparent, it is useful to cut a disk out of the center, leaving an uncut border of 1.5 cm (1/2"). A square of transparent plastic wrap laid over the top of the pot and secured by snapping the lid into place allows a clear view of the activity inside. Using an inverted plastic bag instead of the flat sheet of plastic makes it pos-

sible to enclose longer twigs of foodplant that project above the top of the pot, and thereby increase the rearing capacity of the container. The wrap may be perforated (with a sharpened pencil-point) or not, as the occasion requires. Access is simple when the larvae need to be moved to a clean cage.

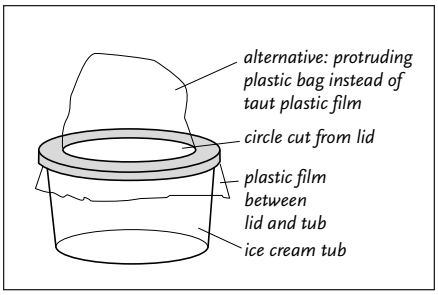


Figure 6–2. Ice cream tub setup.

A very useful container described by Aiello and Cortez (1993) is made from two black-bottomed 2 liter (2 qt.) plastic soft drink bottles.

1. Cut one bottle horizontally (at B, Figure 6–3) at a height of 20.5 cm (8"), to serve as the bottom of the flask, and the other (at C) at 17 cm (6 $^{2}/_{3}$"). The top portion of the second bottle fits down over the bottom portion of the first, making a snug fit, but it is easily removed for dealing with the contents.

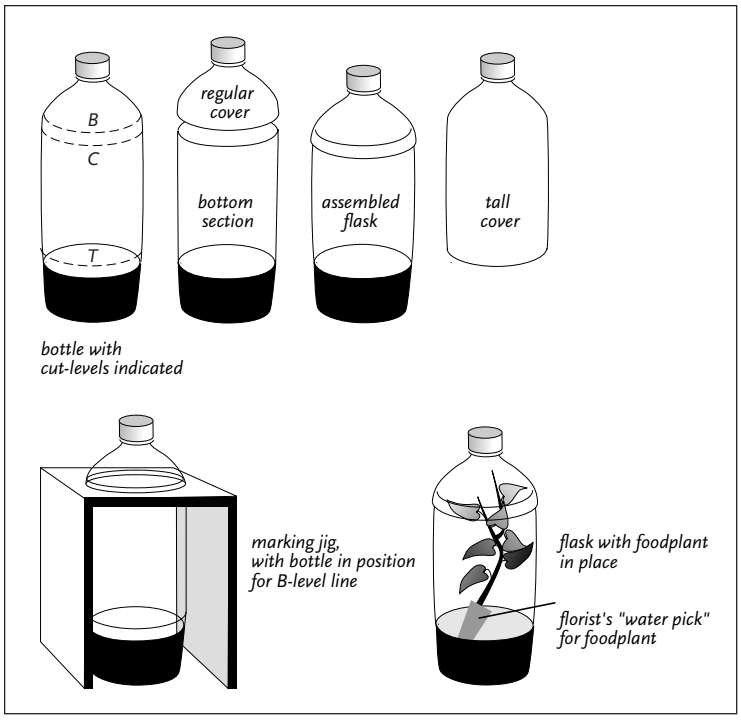


Figure 6–3. Rearing flask made from soft drink bottles.

2. Place a layer of paper towel in the bottom. Put a sprig of foodplant into water in a small vial stuffed at the top with tissue, or in a florist's "water pick," and place this in the container.
3. Then introduce one or more larvae and put on the upper portion of the flask.

4. Close the neck with the original bottle cap to retain humidity, or with a piece of mesh fabric held with a rubber band if humidity is excessive.

This setup is particularly useful when you take serial photographs of a developing larva: the sprig of foodplant can be removed and positioned without disturbing the caterpillar.

Cutting the second bottle (at T, Figure 6–3) about 1.3 cm ($^1/_2$") above the black base, rather than just below the shoulder, produces a larger top section. This increases the rearing space by more than 25% (Winter 1993).

Some rearers build a platform in which a few small holes are drilled (Figure 6–4). Through each hole is thrust the butt end of a small branch into a bottle of water beneath. It is important to plug the space around each branch with moist tissue so that larvae cannot crawl down and drown. Larvae do not instinctively avoid crawling into water, and the surface tension can easily trap them. An inverted jar covering the leafy part of the branch, or a cylindrical screen cage with a lid, keeps larvae from wandering away.

Cylindrical screen cages (Figure 6–5) can be easily constructed by tying a rectangle of screening with string to the desired circumference (using a 1.5–2.5 cm [$^1/_2$–1"] overlap), then sealing the overlapping vertical edges with hot-melt glue. Fiberglass screen is stiff enough for smaller containers, but metal screen is better for larger ones. (Fiberglass does not reliably exclude hungry rodents.) Cake pans, pizza pans, or round metal cake or cookie tins make good tops and bottoms for screened cylinders. Foodplants can be set in bottles of water within the screen cage, but again, the loose space in the neck of the bottle should be plugged with damp tissue to keep larvae from entering the bottle and drowning. This style is the choice when free ventilation is needed.

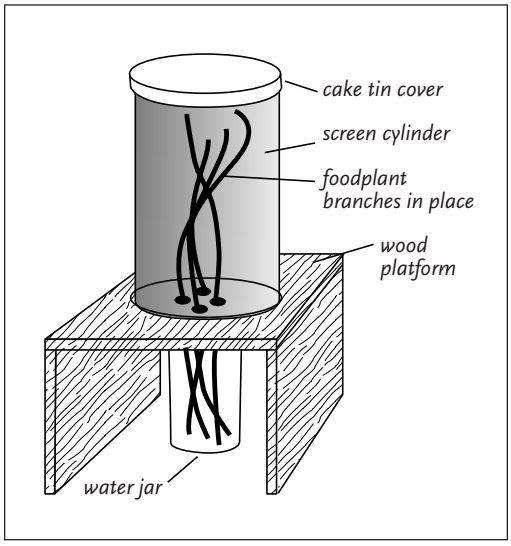


Figure 6–4. Perforated platform setup.

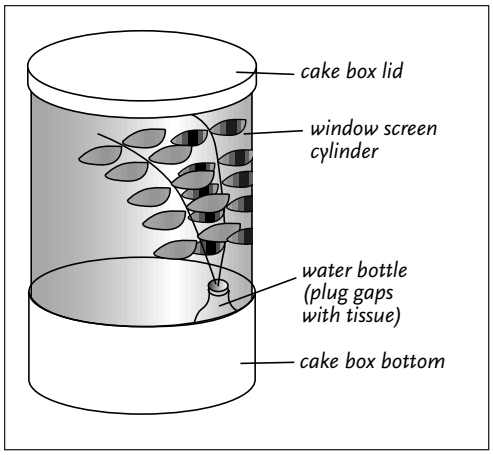


Figure 6–5. Cylindrical rearing cage.

A quickly made container for a potted foodplant uses a cylinder of semirigid sheet plastic surrounding the pot and held by rubber bands (Figure 6–6). A mesh cover completes the cage (Eff 1949). An inverted plastic bag can sometimes be substituted temporarily for the cylinder, but excess humidity soon becomes a problem.

One of the oldest rearing setups is a potted foodplant in a flower pot covered with a lamp chimney, and capped with netting. I once used this method with a single tiny cress plant bearing three *Anthocaris midea* ova (falcate orangetip). The plant grew just fast enough to support all three to pupation and eventual emergence. At cessation of feeding nothing remained but a single half leaf and part of a seed pod!

Sleeves and bags can be made of relatively inexpensive unbleached muslin, or of mosquito netting of a material that does not deteriorate under prolonged exposure to the ultraviolet radiation of sunlight. Many army-navy surplus stores carry nylon mesh that can stand use

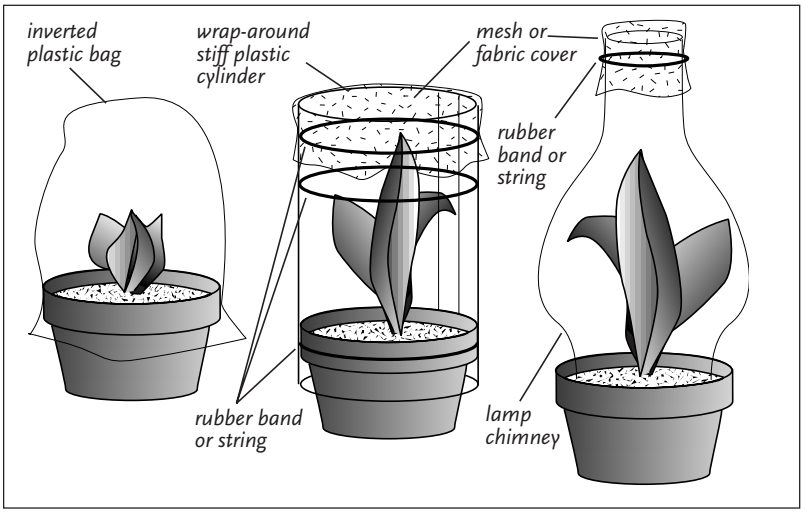


Figure 6–6. Setups for foodplants growing in flower pots.

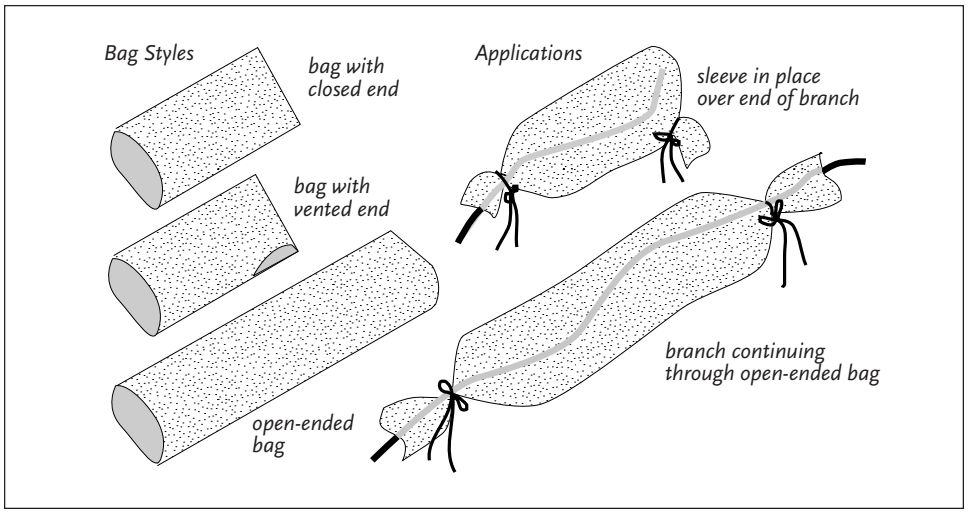


Figure 6–7. Closed bag, vented bag and open-ended sleeve.

in sunlight for several years. Cheese cloth is too flimsy. Both ends of a sleeve are left open; cut edges should be hemmed to prevent raveling. Some lepidopterists stitch one end completely closed, making a bag. Others leave about 15 cm (6") open at one corner as an alternate route for introducing larvae or removing droppings. King-size hotel surplus pillowcases are also serviceable and relatively inexpensive.

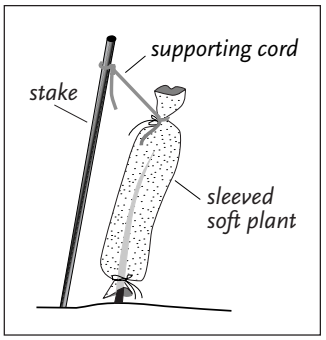


Figure 6–8. Sleeve supported by stake.

Size and shape depends on your own wishes, the nature of the foodplant being covered, and the number and appetite of the larvae being reared. The width of the available material is also a determinant. Narrow sleeves are convenient for long, slender branches or tall herbaceous plants. Broader sleeves or bags are handy for bushy branches. Slip the sleeve over the branch and tie it securely at the bottom with string. Place larvae within the sleeve, and tie the top end shut. If you are sleeving nocturnally feeding noctuids it is good to put in clean, dry leaves or crumpled paper towels for daytime hiding places.

Bags become quite heavy when rained on. You can support the outer end of a bag by tying it with a string to a higher branch. If you

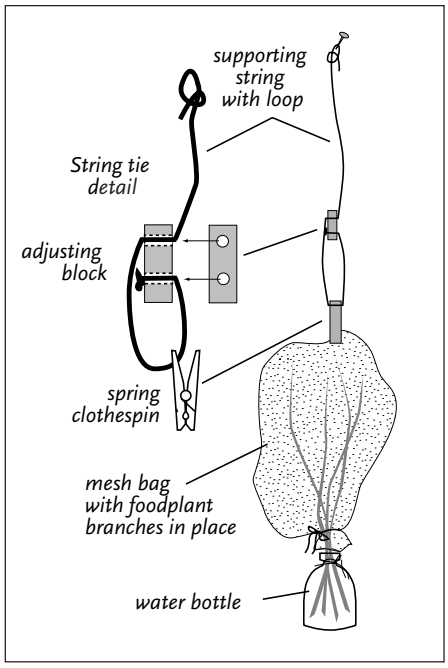


Figure 6–9. Indoor bag setup.

are using a herbaceous plant, tie the top end of the sleeve to a stake driven into the ground alongside to keep the setup erect (Figure 6–8).

You can use bags advantageously indoors or on porches (Figure 6–9).

1. Bunch together several branches of foodplant with the lower 20–25 cm (8–10") stripped of leaves, tie them with string and insert them head-first into a bag held with the open end up.
2. Drop the larvae onto the foliage, tie the bag tightly shut around the bare stems just below the leaves, and place the stems in a deep jar of water.
3. Suspend the top of the bag from a string run over a cup-hook or nail high on the wall or ceiling, and clipped to the bag with a spring clothespin. String length can be made readily adjustable by use of a tent-rope type of stay.

Mesh bags designed to fit your particular situation are best for this use, so that the tenants remain visible. If you wish to keep the bags expanded in a cylindrical shape (Figure 6–10):

1. Make two hoops equal to the circumference of the bag. Use plastic tubing about 5 mm ($^1/_4$") diam.; join the ends by plugging with a short piece of dowel.
2. Attach the hoops to the outside of the bag one-third and two-thirds of the way from the top by sewing them on at intervals with heavy thread.
3. Or use ready-made circular or elliptical embroidery hoops in similar fashion—no sewing necessary.

If you are using a foodplant that wilts quickly even in water (hickory, walnut, ailanthus, etc.) you can maintain it in good condition an extra day or two by covering the mesh bag with an inverted plastic bag. Leave the bottom of the plastic bag open. Adding a few drops of glycerin to the water is recommended to avert wilting (Guilbot 1982).

Rectangular cages with glass or screen sides should be built with a

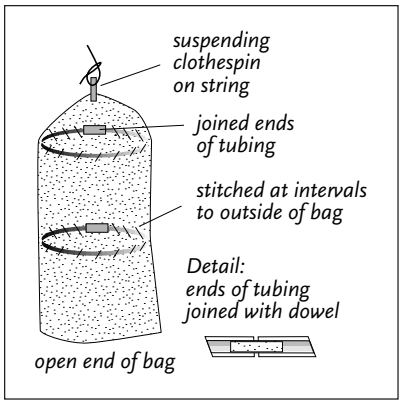


Figure 6–10. Hoops to keep bag expanded.

few particular points in mind: easy access to the contents; ease of cleaning; security against predators, such as small spiders; protection from wind and rain, if the cages are used out of doors.

Open rearing is sometimes used with larvae that tend not to roam between meals, such as some saturniids and sphingids (Codella 1985):

1. Drill multiple holes through the lid of a wide-mouthed jar and fill it with water.
2. Insert individual stalks of the foodplant through the holes and allow the larvae to feed exposed on the tops. Stuff any slack space with tissue so larvae cannot crawl down into the water and drown.

This setup is best used indoors or on a porch, to avoid attacks by birds and parasitoids. Housekeeping is easy, there are no humidity problems, the foodplant remains well hydrated, and the larvae have excellent conditions for growth. Enclosure becomes necessary before prepupal wandering begins, however.

A variation on this plan uses 1 liter (1 qt.) cardboard milk or juice cartons (Reinhard 1981). Cut these down to make two square and two narrow flaps, then slit the square flaps and crease all four as shown in Figure 6–11. Fill the carton with water, fold down first the short flaps, then the slitted flaps; thrust the foodplant stem through the crossed slits. The weight of the water stabilizes the base; water can be replenished by pouring it on top of the flaps.

As an alternative, employ a screw-capped 2 liter (2 qt.) juice carton.

1. Loop a rubber band around the spout of the carton, run it through a generous hole punched the vertical flap of the carton, then secure the rubber band by stretching it back around the spout.

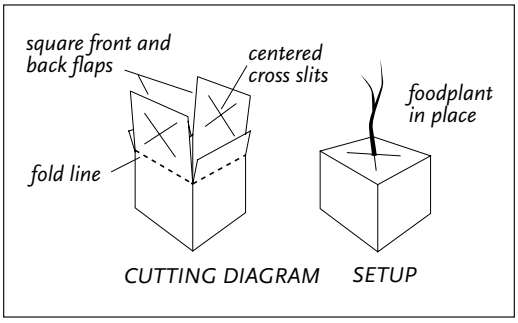


Figure 6–11. Milk carton setup.

2. After placing foodplant branches in the water-filled carton and stuffing any gaps with tissue, lift the upper part of the rubber band from the spout. It will help to hold the branches nearly vertical for greater stability.

Labels are a necessity for containers and enclosures. The appearance of a larva can change dramatically after a molt, and unknowns can become hopelessly mixed without good labeling. The best labels are easily transferable from the old container to the fresh one, and are durable enough to remain legible throughout the life of the larva and pupa. For smooth-surfaced containers, a narrow hand-printed paper strip covered with 2.5 cm (1") transparent tape is excellent. Turn under a short length at each end of the tape, so that it sticks to itself to form tabs, and you have a label that can be lifted and reapplied dozens of times as you transfer larvae to successive fresh containers (Figure 6–12).

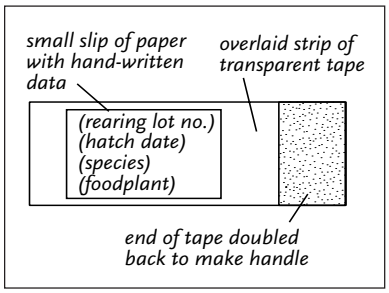


Figure 6–12. Tape rearing label.

For sleeves and bags, the labels can be made from tie-on baggage tags, stiff paper tags with twist wires (office supply stores), or heavy kraft paper attached to the fabric with a safety pin. Writing should be done with a marker resistant to water and sunlight, such as a "Sharpie."

Record on the label the date, place, and stage acquired, the foodplant being utilized, and the name of the insect, if known. If someone else collected the livestock, add that person's name also. An individual serial number for each batch of livestock reliably connects your photographs of this season's uncertainties to the adults emerging next season.

4. Caring for Ova; Hatching and Hatchlings

Ova that are going to hatch promptly the same season usually do so within 5 to 14 days from being laid. If the habits of the species are unknown, observing them a few weeks longer may be prudent. Ova laid on the walls of a container can be left in place, but leave the container open for a day to discharge any excess humidity (mold growth on eggs is lethal). Then keep it closed to prevent dispersal of

the larvae when they hatch. Foliage bearing ova should be thoroughly air-dried before being enclosed. Containers must also be protected from sunlight. Heat kills. If you cover a container with fabric, use a very fine weave. Newly-hatched larvae can walk right through ordinary netting.

Ova that will remain dormant until the next season can be placed in closed containers and held in a shaded and unheated place until the following spring: a shed, an unheated garage, or a covered cellar entrance stairwell (if on the shaded side of the house). For proper development, overwintering eggs need exposure to low temperatures such as they would experience in the wild. These eggs are more tolerant of high humidity than are nondiapausing eggs (McCabe 1991). If the storage area is a few degrees warmer than ambient, your eggs may hatch before food is available.

Diapausing eggs may be brought indoors in spring, when leaf buds of the foodplant begin to burst. They will often hatch in less than a week or two. This subject is covered in detail in Section 10, on hatchlings of various species.

At any season, give hatchlings the opportunity to eat their egg shells, which may be their first meal. The larvae should be supplied promptly with fresh buds or leaves. If for any reason the eggs hatch before leaf buds are opening, even in the cold of winter, food can be accessed by picking off the bud scales to uncover the dormant, unexpanded leaves inside (L.F. Gall, pers. comm.).

Handle newly hatched larvae only with a fine brush.

Sometimes it is helpful to spray eggs with water to initiate hatching—in this case a ventilated container is necessary.

Getting larvae to feed usually calls for nothing more than enclosing them with fresh leaves or buds of their preferred foodplant. Spraying hatchlings and foodplant lightly with a fine mist of water can help to stimulate feeding. The key word is "lightly"—soaking can result in drowning or fatal mold growth. Another recommendation for dealing with larvae that fail to feed initially is to refrigerate foodplant and larvae, in their container, for about 24 hours at 5° C. (41° F.) (Guilbot 1982). When they warm up they allegedly begin feeding.

Some moth species engage in "first instar dispersal," a two- to five-day period of wandering (McFarland 1973). Dropping on silk tethers

leads, in the wild, to widespread dispersal by light breezes— "ballooning." The eggs of these species are smaller than expected for the size of the parent and are laid in masses glued tightly to the substrate. Captive hatchlings travel towards the brightest side of the container, which is also where condensation first develops. Drowning is a risk. This behavior occurs in some ennomine geometrids and in some notodonts and lasiocampids. Steps to avoid the problem are as follows:

1. Furnish the bottom of the container with a layer of paper towel to provide good footing so the larvae can extricate themselves from their own threads.
2. Place the sprigs of offered foodplant so that some leaves are touching the top and some the bottom of the container and are not tightly layered. This gives larvae that have begun to feed some protection from disturbance by those still wandering.
3. Place the container in complete darkness to thwart the attraction to light and at a temperature under 21°C (70°F), if possible. You should inspect these containers several times a day to spot developing problems.

In less extreme situations where wandering to the top or the bright side of a container is a problem, just cover it with an inverted brown paper bag and set it in the least lighted part of the room. Once feeding is well established, the bag becomes unnecessary.

If a proper foodplant for hatchlings is not immediately available, there is the following recommendation: "Place a piece of cut apple in a small box with the newly hatched larvae. Recut a thin slice off the cut surface each day, so that the surface continues to give off moisture. This slows down desiccation of the small larvae, and at the same time, they will often nibble at the cut surface of the apple, thus obtaining some nourishment" (N.B. Tindale in McFarland 1965). Before cutting, the apple should be washed to remove any residual insecticides.

As an alternate delaying tactic, you can place the larvae on a paper towel in a clean plastic box. Spray the towel with water; they will usually drink. Allow the paper to dry somewhat, close the container tightly, and store it in the refrigerator. The larvae survive for 3 to 30 days, and will be ready to feed when you can provide them with foodplant.

While getting saturniids started is usually not difficult, Miller and Cooper (1977) reported a carefully tested method: bag the mated female moth for a single night in a paper lunch bag for egg-laying; store the bag folded for eight days; expand the bag; put three twigs of the foodplant, each bearing four or five leaves, into the bag, top end first; twist the end shut about the stems, and place the butts of the twigs in water. Within 48 hours 80-100% of the emerged larvae should be feeding. If the moth has laid very large numbers of eggs it may be better to cut the bag in pieces, each bearing only a few dozen eggs. Place each piece between the twigs of foodplant in a new, clean bag set up in the manner described above. This should reduce loss from overcrowding.

5. Caring for Larvae

Larvae found feeding in the wild need merely be continued on that same species. If you do not know the plant's name, collect a specimen (as described in Section 15) and try to identify it with the help of a botany text or a botanist. When you know the identity of a moth or butterfly from which you have obtained eggs, field guides or other texts will often tell you the foodplant(s).

Caring for the foodplant depends considerably on the container and the plant being used. A closed container, such as a plastic box or glass jar, should have a layer or two of slightly moistened paper towel placed on the bottom. This helps maintain humidity but also absorbs excess moisture; it makes droppings visible, and easier to clean up; and it is used as a resting pad by some larvae when they are shedding their skins. The leaves supplied should be fresh, not grossly wet, and so placed that they do not lie flat on one another and reduce access for the larvae. If a sprig of foodplant is placed in a small vial with perforated plastic cap, as used by florists in corsages, it can stay fresh longer. In that case, the paper towel should not be moistened.

Food should be replaced frequently, every two to three days for most plants. Tests with woody foodplants, cut and handled as when used for indoor rearing, showed that food values could change significantly in even the first few days (Miller 1987).

If foliage is all consumed, larvae will attempt to eat the less nutritious bark and twigs, or each other. If foliage becomes old, wilted or begins to discolor, the larvae fail to thrive or become diseased. Well

hydrated leaves optimize nitrogen utilization and foster good growth (Codella 1985). Dehydration can lead to poor nutrition, slowed growth and diminished survival. Larvae are better judges of food quality than are lepidopterists, but they seldom communicate early enough to avoid trouble.

Pine is a case in point. It may look fine to us, but it quickly becomes nutritionally deficient (Tuskes et al. 1996). Oak is another example. New growth of most oak species degenerates, visibly or otherwise, in one to three days after cutting. If willow oak *(Quercus phellos)* is available, it should be used for starting oak-feeding larvae because it stays fresh as long as a week. However, it also accumulates tannins earlier than other species, so it is seldom a good choice for prolonged sleeving.

Space is vital. There should be neither too much foliage nor too many larvae in a container. Caterpillars need "elbow room" to move about and to feed easily. They should not have to compete for leaves to eat or twigs to grasp, or be disturbed by other larvae searching for more food. Larval crowding often results in poor growth and increased susceptibility to disease, especially in later instars. Naturally gregarious feeders are an exception. They may fail to thrive if they cannot feed in proximity to siblings.

Housekeeping is important for the health of the larvae. Droppings accumulating in containers rapidly become moldy, and larvae that choose to rest on the bottom of the container are forced to wallow in their own excrement. Disease, debilitating or fatal, readily develops. This is less a problem with sleeved livestock, except in persistent wet spells.

Containers must be thoroughly cleaned before reuse. Wash with detergent and hot water and air-dry. If a container has housed known infected material, also soak for five minutes in a solution of bleach, such as chlorine bleach (sodium hypochlorite 5.25%), diluted 1:10 with water. Air dry, then rinse and air dry again. A residual chlorine odor is not harmful. Implements, such as forceps or brushes used in transfers, should be similarly decontaminated.

While the incidence of disease in sleeved rearing is quite low, it is sound practice to disinfect sleeves and bags after each use. In a washing machine use ordinary laundry detergent and twice the amount of bleach recommended on the container; soak several hours

or overnight, then complete the wash cycle; use an extra rinse to remove residual detergent.

Having a surplus of cages or containers, clean, dry, and ready to use, simplifies housekeeping tasks. With minimal disturbance, you can readily transfer the larvae into a new container stocked with fresh food.

It is best not to handle larvae with your fingers, to avoid injuring them or spreading disease. Some larvae attach their proleg crochets so firmly to the perch that they can be released only by active effort on the part of the caterpillar. A struggle with such an individual can result in tearing off a proleg, with fatal outcome. It is safer to pick up, with forceps, the leaf or twig on which the larva rests, and trim off any unoccupied old vegetation. A resting larva may be just about to shed into the next stage, and disturbing it at this point may hinder its escaping completely from its old skin. Constricting bands of this skin can then interfere with further growth. Premolt larvae are usually recognizable because the new, larger head capsule shows through the skin behind the smaller current one that will drop off during the molt. Dislodged molting larvae may be transferred to a small piece of cheesecloth or muslin, which serves as a substitute silk pad. Old saturniid cocoons have also been used for this purpose.

When you are transferring larvae to a fresh container, it is easy to overlook cryptically colored individuals on the back of a leaf or ones that are hiding or preparing to pupate between layers of toweling. If you hold up the leaf or towel so that it is illuminated from behind by a bright light, the shadows of these larvae will be easily visible. This also reduces the possibility of cutting them in two while trimming excess bits of leaf around another individual.

On the other hand, you will soon discover that certain species of larvae, when touched with a slender, short-tipped camel hair brush, will quickly drop from the plant, with or without a suspending strand of silk. Such larvae can be quickly nudged off into the new container without risk of injury. But any larva not dropping immediately should be respected and transferred as in the previous paragraph. Larvae in many families respond to this approach in the earlier instars. Arctiids remain transferable this way throughout their lives, and also many geometrids until the latest instars. Some individuals in the catocaline genera, as well as many pyralids, may flip into frenzied gyrations

when touched, missing the new container completely. Working on an uncrowded table simplifies recovering these acrobats.

The number of larvae per enclosure is important. Large larvae call for larger quarters with fewer tenants. There should not be so many that they consume the enclosed foodplant within 48 hours. If you are rearing a considerable number of one species, such as to look for variant markings in the adults, it is often convenient, as the larvae get larger, to put most of them out to finish their growth on sleeved branches, retaining a few indoors for stage-by-stage photography. Sleeving is without a doubt the most reliable way to bring tree and shrub feeding larvae to maturity. It is a good idea to estimate the number of larvae to include per sleeve on the basis of having the foodplant last at least a week. It you are going to be vacationing, an even lower ratio may be necessary.

Do not wait for foliage in a bag to be completely consumed before transferring the larvae to a new site. Larvae leave the poorer quality leaves until last, and these are best left uneaten. If you are able to allow a batch of larvae to mature on successive branches of a single tree, this is ideal. Since many trees stressed by defoliation one year will produce unpalatable leaves the next year or two, stay with a tree that is being accepted. This same caveat applies to successive selections of foliage for container rearing.

When you transfer larvae to a new sleeving site, if the current branch can be sacrificed, then clip it off below the sleeve to simplify the procedure. You can then prune off the parts of the vegetation on which the larvae rest, and place them temporarily in a large jar. The contents of the jar can then be readily dumped into the new sleeve. Individuals resting on the inside of the sleeve are a problem. It is best to turn the sleeve inside out and then tickle the rear end of the caterpillar to set it in motion and onto a twig or leaf. Pulling one forcibly from the sleeve fabric may tear off a proleg because of tangled crochets. If you are reusing the same sleeve, just leave the larva on the inside and use care as you slide the sleeve onto a new branch.

If possible, it is a good idea to inspect sleeves and bags every few days in damp weather to clear out accumulated droppings.

If you wish to rear arctiids in sleeves, try willow, black cherry, honeysuckle or sunflower and relatives. A single castor bean leaf can be sleeved for rearing a batch of arctiids (Flaschka 1989a). Healthy,

well-fed larvae will usually survive the winter in clean leaf litter on damp peat, in containers with some ventilation, placed outdoors for the winter. Disease acquired late in the season, with indoor rearing, is often a source of failure.

Borers, such as some sesiids and cossids, can be reared more rapidly by using root vegetables into which the larvae can bore: white or sweet potato, yam, beet, carrot, turnip, or parsnip, used raw. Individual housing is recommended, since cannibalism can occur if larvae are crowded. A hole can be punched into the root, and the larva headed into the hole. Deteriorating roots should be promptly replaced. As an example, rearing time for *Prionoxystus robiniae* can be reduced to one year (McFarland 1968). Such substitute foodplants have been tried for *Papaipema* (E. Quinter, pers. comm.). One species made it all the way from egg to adult, and a few did reasonably well in late instars. Most species showed delayed development, suboptimal growth, and often produced deformed pupae or adults. Quinter recommended such substitution only when the natural foodplant kept poorly or was simply unavailable "back home." Grape Nuts and other dried cereals can be used as an alternate food source for wood borers (Stehr 1987).

Flower moth (heliothentine) larvae, being reared singly in vials containing their foodplant blossoms, can be tided over for a day or two with a slice of green snap bean as a substitute for their proper diet. Larvae of this group are best moved to larger individual vials as they grow, with the last instar vial having the bottom one-third filled with damp soil for pupation. Throughout the rearing, it helps to include a piece of green bean or wedge of carrot to maintain humidity (Hardwick 1996).

At times you may wish to set very young larvae or even eggs out in sleeves to give them optimal rearing conditions. Eggs clustered on twigs can be tied to the foodplant, as can patches of eggs on paper. Actively exploring or dispersing larvae must be sleeved in muslin, not netting, to prevent escape. However, if you have very few ova, it is better to rear the hatchlings indoors through the late first or early second instar before sleeving them out.

When you are installing very young but very active larvae (*Catocala, Zale,* many geometrids), just dump them into the sleeve. They will quickly explore and establish themselves on the new foliage.

But with more sluggish young larvae you need to use wire twists to attach the foodplant on which they currently rest to young twigs with new leaves, or they may fail to find palatable food.

Single-brooded early spring feeding larvae should be sleeved on the largest available leaves when leaf development is just beginning, but thereafter should be put on the youngest foliage available. These species cannot thrive on mature leaves, which contain too much tannin or other defensive chemicals. For many species, *Prunus serotina* (wild black cherry) is an acceptable substitute if the regular foodplant is overmature.

Siting is also important. The sunny side of a tree or shrub is preferable; on the shady side additional shading by the bag may cause the foliage to yellow and deteriorate. On the sunny side the larvae can get shade from the bag and from the foliage.

> ***Note:***
>
> *It is imperative that you never rear anything exotic outdoors in sleeves if there is the slightest chance that it could survive in your climate, regardless of whether or not this would violate any laws.*

Despite your best care, larvae will occasionally escape from sleeves and bags. Larvae sometimes worm their way out between folds in the cloth, or chew their way out, or gypsy moth larvae make holes from the outside. Birds, rodents, and raccoons may tear the fabric. Remember the gypsy moth and all the problems stemming from its accidental release in Massachusetts.

Light serves as a cue in the development and diapause of Lepidoptera, especially gradual changes in day-length, or the daily ratio of light to darkness (photoperiod). Most important are the determination of continuous development vs. diapause, and the development of seasonal variations in markings (seasonal polyphenism) in some species. While most larvae will mature regardless of photoperiod and can therefore be raised indoors even though lights remain on well into the evening, their development, coloration, and emergence time may not be comparable to that in nature. Hence it is most important that data labels on reared specimens include “ex ovo”, “ex larva” to document the fact of rearing. And if you are rearing a species whose life history is previously unrecorded, the use of natural outdoor

photoperiod and outdoor temperature can reduce the possibility of laboratory-induced variations. Species that diapause as larvae can be particularly confused by unnatural photoperiod and temperature, arctiids being one example. A simple timer clock, used for controlling indoor lamps, is useful for maintaining a desired photoperiod in your rearing area.

Some day-feeding sphingid, saturniid, and arctiid larvae (especially certain southwestern species of *Agapema*, *Hemileuca, Pseudohazis, Saturnia, Grammia,* etc.) have, in their later instars, a definite requirement for sunlight for an hour or two daily, preferably in the morning. They should be in well-ventilated containers with some shaded areas always available for them to retreat into.

Many of these also benefit from a light early morning misting; they suck up the droplets as if they were dew. Very hot, dry weather calls for additional spraying.

Observations on larval behavior in the field in the early to mid-morning hours—basking and feeding, vs. hiding in the shade—give clues to their requirements (McFarland 1974, 1988).

You can provide natural conditions of light and temperature on a sheltered, screened porch with limited exposure to sunlight, or in a "rearing tent" (Brewer 1972). This is a collapsible screened tent of the sort used by people to protect themselves from sun and mosquitoes for summer relaxation. Set up on level turf and equipped with a few simple tables, it provides sheltered but natural conditions for outdoor rearing. Be sure that the tent and contents are secured against upset by strong winds.

Mice, with their ability to breach barriers, are noted as a risk in various places in this manual. A cat is not a solution; normal cat behavior can upset your setups, with damage to or loss of livestock.

Fungal, bacterial, and viral diseases, often infectious and either fatal or merely debilitating, occur in nature, and may be accentuated in rearing situations of excess humidity, crowding, and foodplant deterioration. Sometimes these diseases may already be present in larvae at the time of collection, or contaminated foodplants may be the source of introduction. Some viral and sporozoan diseases can be transmitted through the egg to succeeding generations. They spread easily from one larva to another within a container. Inadequate cleaning of implements and containers readily spreads infection from

one group of larvae to another. A mild infection with which a larva may continue to grow and function may nevertheless make it more susceptible to some other, fatal, infection, such as polyhedral virus disease or bacterial sepsis (Guilbot 1982). Signs of infection vary with the disease; a few examples follow:

- Dark spots or white masses on larval skin, irregular growth, or cessation of feeding (without immediate death) can result from microsporidian infections. Survival is accompanied by decreased vigor and fecundity. Microsporidian spores can be picked up by feeding, or can be transmitted through the egg to larvae.
- A larva dangling with an intact skin enclosing a liquefied interior is probably victim of a nuclear polyhedrosis or granulosis virus infection, acquired from eating contaminated food (Diamond 1975). Similar infections may be responsible for major fluctuations of monarch butterfly populations (Urquhart 1966). Larvae seem most susceptible to infection in early instars, and there is no evidence that once infected a larva can survive (Bucher & Turnock 1983).
- A larva that turns pink or red and liquefies rapidly after death may have succumbed to a *Serratia* (bacterial) infection (Guilbot 1982).
- A sick larva with a relatively large head but undersized body, not liquefied, may (if not simply starving) have a cytoplasmic polyhedral virus infection. This is not always lethal.
- White lumpy lines along the sutures of a pupal case are usually the result of a fungus infection. This can be the cause of death, or can occur secondary to pupal death.

If you are rearing successive generations, disinfection of ova can reduce larval mortality (Troetschler et al. 1985). These authors caution that this treatment may increase the susceptibility of the developing ova to drying out, but maintaining the eggs on their host plant leaves, at moderate humidity, seems to avoid this. The specifics, as modified by F. Chew (pers. comm.) involve soaking the leaves bearing the eggs in a 1.5% bleach solution for five minutes, then soaking in three successive changes of clean water for three minutes each. While this concentration of bleach is tolerated by pierids such as *Colias* and *Pieris*, Chew suggests trial of other families at 0.5% or 1.0% until

their tolerance is ascertained. Treatment of eggs in this fashion eliminates only those microorganisms present on the outside of the egg (from the mother) or on the foodplant (from other insects in the environment). If infectious agents are passed intracellularly from the mother, as the polyhedrosis viruses may be, they are not eliminated by this procedure.

Disinfection of larvae during rearing has been employed successfully, using a dilute solution of potassium pyrosulfite, $K_2S_2O_5$ (formerly known as potassium metabisulfite, and available in winery supply stores) as a generator of sulfur dioxide, SO_2, for sterilization. A 1:10 "stock" solution (in water) is made up, then diluted again 1:10 to make a "working" solution. This latter solution may be applied as a very fine spray to wet the entire surfaces of foodplant leaves that are then given to the larvae after complete drying. Alternatively, the spray can be applied very lightly to leaves and larvae alike in an enclosed rearing container. Here, it is important to practice with some expendable larvae, since overzealous application can prove fatal. The goal is to produce just a faint odor of sulfur dioxide (Flaschka 1989b). Use plastic spraying equipment; SO_2 can corrode metal.

Pupae can be disinfected by a 20–60 sec. immersion in 1:10 chlorine bleach

Cannibalism and carnivory are important risks. Some noctuid and arctiid larvae have such a strong propensity for this in the wild that they must be reared in "private rooms", one larva to a container (Schweitzer 1979). Crowding will lead to cannibalism in other species. Cecropias have been noted to dine on their cage-mates when they are short of moisture (Holliday 1988), so daily spraying of foliage in open cages is recommended. Container-reared arctiids and *Celastrina* may eat the pupae of individuals that pupate first; this happens in the brief interval before the pupal shell hardens. Since female larvae generally feed longer than males, the result can be a severely distorted sex ratio in the emerged adults. It is possible that later dispersion of larvae that are naturally gregarious in the early instars may be a protective adaptation to avert cannibalism. Geometrids are almost never cannibalistic, but *Protoboarmia* and *Campaea* become highly so in winter, when they have to feed on buds (D. Schweitzer, pers. comm.).

When larvae of assorted species are collected by beating from a

single tree species, it may be tempting to rear the whole group in one container. This often results in carnivory by one or two larvae, and the population drops rapidly!

Parasitoids are a fact of death when field-collected immatures are reared. An apparently healthy larva will suddenly discharge a few tachinid fly maggots or a bevy of braconid wasp larvae. Or the caterpillar will pupate successfully, but there emerges, in due course, a wasp rather than a moth or butterfly. Not even eggs are spared. A swallowtail or sphinx ovum that develops a mottled appearance may soon give rise to a dozen or more minute trichogrammatid wasps. It is important to retain and label properly the reared parasitoids, indicating, if possible, the species of host caterpillar. The manner of preserving them is similar to that for adult Lepidoptera and is covered in Chapter 8. As parasitoid specimens accumulate, they should be sent to specialists for study. The biology of parasitoids of even common Lepidoptera is often poorly known.

Sleeved rearing does not give absolute protection. Larvae in mesh bags sometimes feed or rest with their bodies in contact with the netting. Parasitoids can readily deposit their eggs through the mesh. A bird may also spy such caterpillars and tear a hole in the sleeve to get at them; other larvae escape through the holes. Mice can chew their way in, seeking pupae. Sleeves can be made from metal screen wire, if these are recurring problems. Further risks are the "stay-behind" predators, particularly spiders, "true bugs" (Hemiptera), and lacewing larvae (Chrysopidae) that are present on the vegetation and get sleeved along with your caterpillars for an extended free lunch. Sharply shaking the branches to clear them of such predators is some help, as is a vigorous rinsing with a hose, if the branch is situated where this can be done.

"Microlepidoptera" can actually be simpler to rear than their larger relatives (De Benedictis 1993). Place the foodplant containing the larval mines or webs in a 20 x 10 x 50 cm (8 x 4 x 18") clear plastic bag of 0.04 mm (1.5 mil) thickness. Keep the bags inflated to form small terraria. The bags should be tough enough to prevent larvae from chewing out, and large enough so puddling doesn't result in drowning; add a few paper towels to soak up moisture. With woody foodplants almost no care may be required, whereas care needed for soft herbaceous foodplants may be quite the opposite. Check folds

and corners of bags for hiding adults before opening. When transferring livestock to a new bag, any individuals spun onto the inner surface of the bag should be cut from the bag rather then picked off. Prepupal or pupal diapause of some "micros" can be very long—17 years in the case of a yucca-feeding moth, *Prodoxus y-inversus* (Powell 1986, 1989, 1992).

To be most successful, the mining and pupating habits of each species must be considered (Braun 1950). Even though the entire feeding cycle occurs within the mine, pupation may occur in the mine, on the leaf surface, in some crevice or underground. If a species is a full-time mine feeder, it is best to flag the leaf, and collect the larva when it is nearly grown. If it overwinters as a partly grown larva, spring collecting gives better rearing results.

Preservation of larval specimens is covered in detail in Chapter 8.

"An ounce of prevention is worth a pound of cure" is the watchword when rearing. The principles of "clean" rearing can be summarized as follows:

- Use plastic or glass containers that have been thoroughly cleaned between uses.
- Pay close attention to housekeeping. Transfer livestock to new containers frequently; replace spent foliage promptly so larvae can feed as freely as in the wild; do not allow them to run out of food.
- Always avoid overcrowding.
- Limit the amount of rearing you do to the time available for the task. It is easy to get eggs from a dozen species of moths, then discover a few weeks later that you need 30 hours of free time per day to care for them all!
- Control humidity. In the early instars humidity is more important than ventilation. In later instars good ventilation in fabric or screen enclosures increases success and reduces the need for frequent housekeeping. Periodic light misting with a hand-squeezed sprayer is often beneficial (not usually necessary for larvae sleeved on growing plants).
- Provide appropriate sunlight for those species requiring it.

6. Preparing for Pupation

Caterpillars give one or several clues as they enter the prepupal

period. All will empty the gut. Instead of formed, relatively dry fecal pellets, most put out a formless semiliquid dark mass, signaling the end of feeding and digestion. Most naked larvae develop a "glassy" appearance prior to pupation. And nearly all begin to wander compulsively, in the wild going down from the foodplant and walking considerable distances in search of a suitable site for pupation. In captivity they will go around and around the bottom of the container, churning up whatever may be there: paper, droppings, soil or peat, or whatever. Because this churning activity can disturb individuals already trying to pupate, transfer larvae to a pupating container as soon as they show signs of prepupal behavior.

Successful pupation calls for conditions that vary from very simple to highly specialized, depending on taxonomic group. Many butterfly larvae pupate suspended from sticks, plant stalks, and leaf ribs. In the absence of such supports they will spin a silk pad on the side or lid of the container and pupate there. (Photography in such a situation is both difficult and unaesthetic.) They need "elbow-room," so the freshly formed, still soft chrysalis will not be impinged upon and distorted by adjacent vegetation. *Hyalophora* and *Callosamia* likewise benefit from twigs and leaves to use as supports.

If you are rearing larvae in sleeves, the cocoon-maker may spin in among the folds of the fabric. In such cases, the bags should be left undisturbed for a week or two. A saturniid may take several days to complete the interior of the cocoon. A *Hyalophora* larva constructs a cocoon with an "escape valve" at the upper end. If you inadvertently invert a partially completed cocoon, the pupa may end up oriented away from the valve, and the eclosed moth may be unable to escape. In addition, a cocoon disturbed before the pupal shell has hardened may yield a deformed adult or none at all.

A few butterfly groups, and a great many moths, pupate in loose litter on the ground, spinning a few supporting bands of silk to hold dead leaves together, or constructing a more formal cocoon. For these, shredded paper or crumpled dead leaves on a base of loose soil or peat is satisfactory.

Many moth larvae burrow into the soil—sphingids, imperial moths, most geometrids, noctuids and notodonts, and many "microlepidoptera." A container partly filled with slightly damp peat or barely moist loose soil meets the needs of these species. Sterilized

commercial garden peat reduces the likelihood of mold development. Sterilization is best done with an autoclave, but an oven at 120°C (250°F) is effective. Do not use microwaves. "Weathered peat"—peat that has been left on the ground out-of-doors—has all the fungus and mildew "cycled out of it" and can be used safely for overwintering pupae (T. McCabe, pers. comm.).

Some larvae are very specialized in their requirements. Agaristine moth larvae and some of the acronictines need soft or punky wood in which to bore a pupation chamber. These larvae can chew their way out of a thin plastic container in search of an acceptable site. A noctuid larva that spends several days wandering about its container from bottom to top should be offered a piece of soft wood (or even a piece of rigid polyethylene foam packing material). It will often begin to bore in immediately. Giant skipper larvae pupate within the tunnels in which they have been feeding, as do many other borers.

If you are leaving on vacation, and have nearly mature, ground-pupating larvae that you are rearing in a sleeve, they can often be managed safely by placing a few handfuls of peat, sphagnum, or crumpled dry leaves inside the bottom of the sleeve. This provides a medium in which the larvae can bury themselves for pupation. It may also protect the pupae against desiccation, but this is not reliable in very hot, dry weather. Mature larvae may sometimes chew their way out of the bottom of a sleeve.

7. Preparing for Eclosion

If you are rearing unknowns, it is obviously essential to keep each species separate right through to eclosion. With known species, holding multiple species of pupae in one emergence cage usually presents no problems, provided you can later sort out the hatched pupal shells, which should be saved and appropriately labelled.

Characteristics desirable in emergence cages are:

- enough ventilation to avoid mold growth;
- ease of sprinkling or spraying to avoid undue drying out;
- security from marauding mice and ants;
- internal rough surfaces or supports on which the emerging insect can get a secure foothold while expanding and drying its wings;
- enough space so that simultaneously emerging individuals will

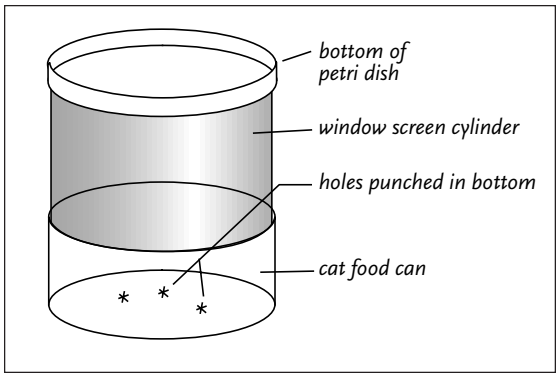


Figure 6–13. Cat food can pupa cage.

not disturb each other during wing expansion;

- visibility such that you can readily see that something has emerged (no blind upper corners or overhangs).

A cylindrical screen cage with top and bottom made from a metal cake or cookie box works well (see Figure 6–5 for details). Cement a layer of rough fabric (such as muslin) to the underside of the metal cover. This will provide a rough surface from which the moth can hang and may also increase success with cage mating. A layer of peat in the bottom can be used to cover subterranean pupae; pupae in leaf litter can be laid on top of the peat, as can saturniid cocoons. Plastic film wrapped around the screening (except for the last 3 mm [1/8"] below the cover) helps maintain humidity without preventing ventilation.

A smaller version of this cage (Figure 6–13) is easily built using a cat food can for the bottom and a plastic petri dish bottom for the lid. This is particularly useful for handling single unknown pupae. Punch a few drain holes in the bottom of the can, from the inside out, with the point of a nail. Fill the can nearly to the top with peat, then lay the pupa on or layer it within the peat. Wrapping the screen with plastic film, as in the previously described cage, helps to reduce the fluctuations of humidity between the necessary periodic sprayings. Fabric inside the lid seems unnecessary in these small cages.

An inexpensive styrofoam picnic cooler with 10 cm (4") of peat in the bottom makes a fine emergence cage and gives plenty of elbow-room, but it lacks visibility and may not be rodent-proof. Glass jars, plastic containers, cardboard ice cream boxes are all useful. Very tight covers should have a number of small holes poked in them. Don't forget to include something for the emerging insect to climb upon.

If you are rearing numbers of pupae of known species (with the egg-layers retained and clearly identified), where mingling of emerged adults will not create a problem, make a removable screened cover (wire screen excludes mice reliably) for a box that you find or build

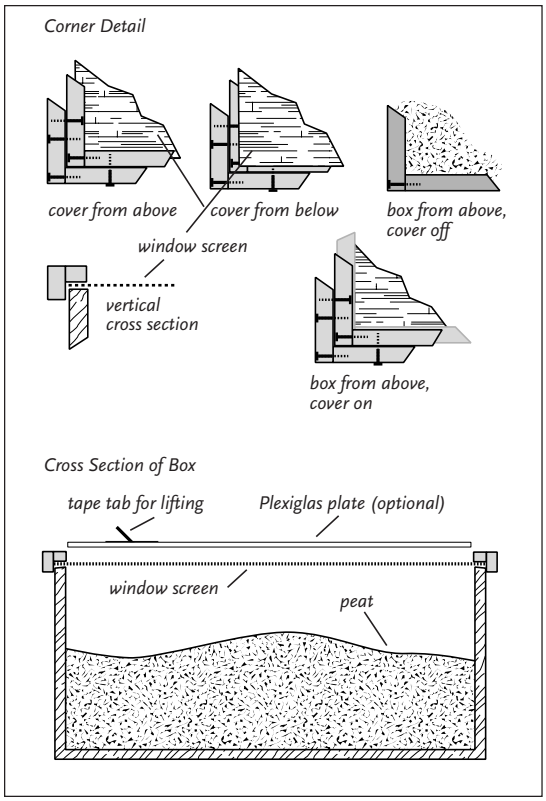


Figure 6–14. Modified Schweitzer pupa box.

(D. F. Schweitzer, pers. comm.). The box can be about 30 cm (1') deep inside, with a floor area commensurate with your needs. Two to 3 cm (nominal 1") untreated stock (not plywood) is satisfactory. Unplaned wood is best. If you use planed stock, roughen any inside vertical surfaces with a wood-rasp or very coarse sandpaper, diagonal to the grain, before assembling. The cover requires a loose-fitting frame about 2–3 cm (1") thick, constructed as indicated in figure 6–14. Tightly staple metal window screen to the under surface of the frame, as shown. With the cover in place, the top inside edges of the box should be visible through the screen throughout the entire perimeter, so that there are no blind spots where emerged moths can be overlooked. Pupae (or estivating larvae—see Section 11, examples 1–4 in Figure 6–25)—are placed within a 10 cm (4") layer of damp peat (or peat over sand).

Keep the cage in a cool cellar in summer, or, for the winter, in a cold garage or shed. Periodic spraying is necessary to keep the peat damp. A piece of thin rigid plastic, slightly smaller than the inside of the box, can be laid on the screen to reduce evaporation but maintain ventilation.

A versatile pupa-pot (C. Henne in McFarland 1988) is particularly useful for bringing "difficult" pupae through diapause.

Begin (Figure 6–15) with a plastic flower pot of the sort used for African violts (with attached saucer and woven fiberglass wick). Fill it with the following mixture:

- Decomposed granite, 6 parts ("Grape Nuts" size, often found

exposed in road cuts in unglaciated granite areas; clean river sand is second best).

- Canadian peat, 3 parts (sold in garden centers).
- Charcoal, 1 part ("Grape Nuts" size, as sold in pet shops for aquarium filters).

On top of this, pushed slightly down into the mix, place a cylinder of clear plastic 10–15 cm (4–6") high, of a diameter to fit snugly just inside the rim of the pot. To the inside of the cylinder, full length, glue three vertical 2.5 cm (1") strips of plastic-coated fiberglass screen (to serve as footholds for the emerging moth). Use waterproof plastic cement (Duco Cement works well) to bind down all the ragged edges

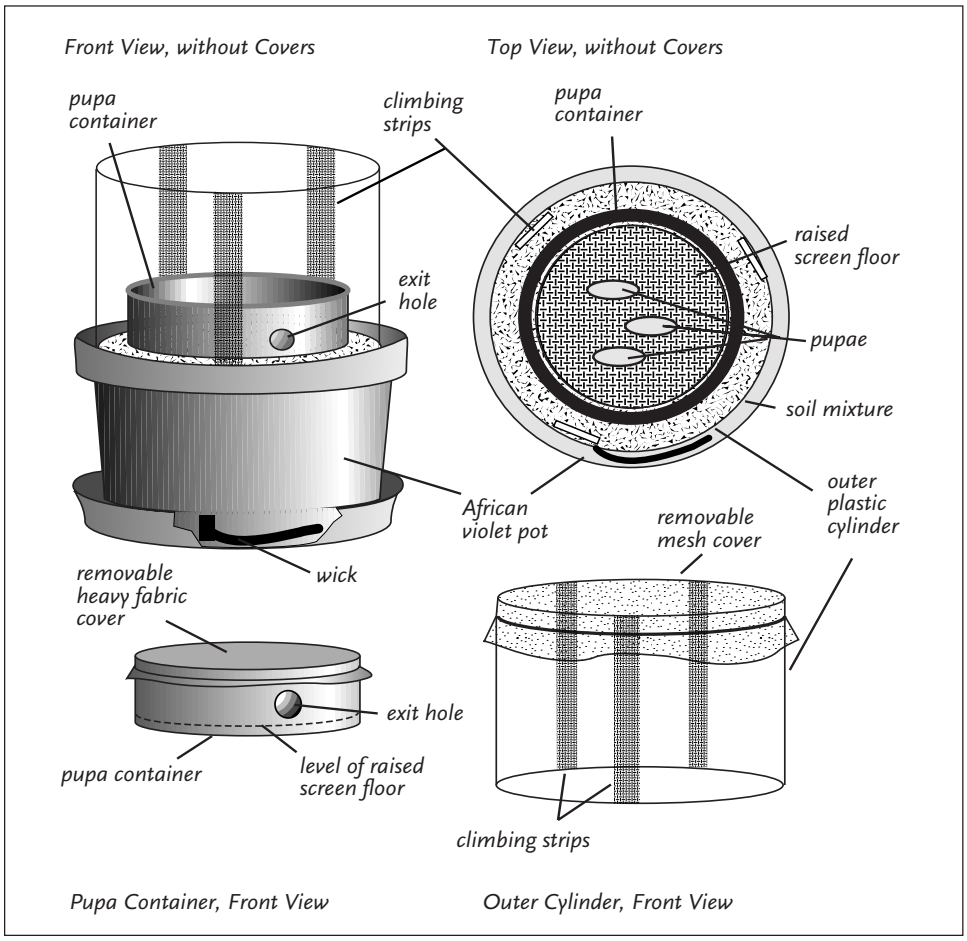


Figure 6–15. Henne pupa-pot.

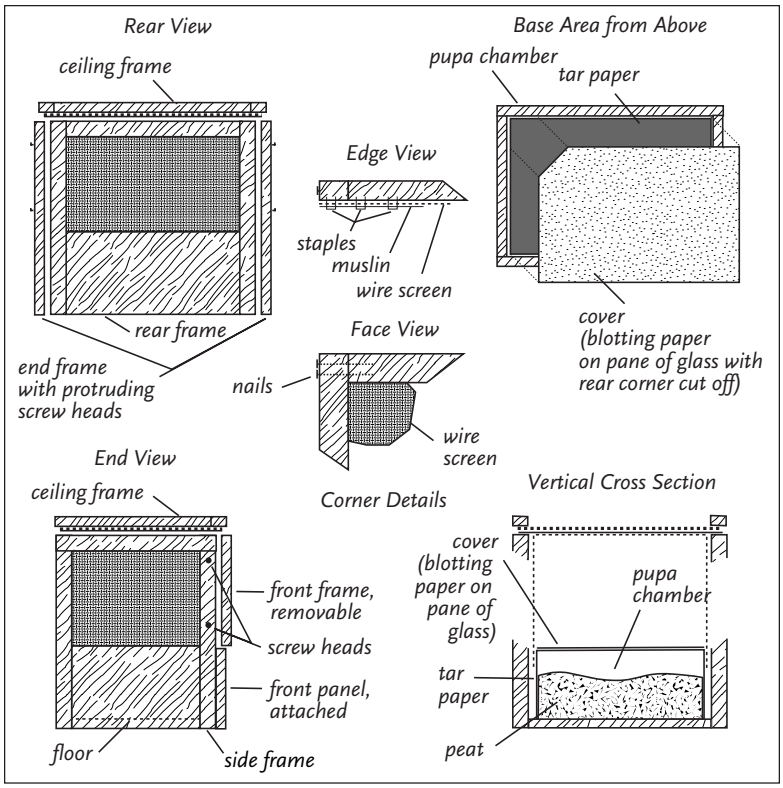


Figure 6–16. Rummel pupa emergence cage.

of the screen. Cap the cylinder with a removable mesh covering.

Inside this setup (Figure 6–15) place a special pupa container, a plastic cylinder with an outer diameter about 2–2.5 cm ($^3/_4$–1") less than the inside of the outer cylinder, and a height of about 4 cm ($1^1/_2$"). For a floor, glue in a disk of fiberglass screen, raised about 0.6 cm ($^1/_4$") above the bottom of the cylinder. Drill a 1 cm ($^7/_{16}$") hole in the side of the cylinder, a bit above floor level. Spray-paint the outside of the pupa container black. Cover the top with thick, tightly woven fabric that will allow slow diffusion of water vapor and air. Lightweight canvas remnants (perhaps obtainable from an upholsterer) should be satisfactory. Be sure to wash them repeatedly until they are free of all chemical residues.

Pupae are laid on the screen floor of the pupa container, its cover is replaced, and the container is set on the soil within the outer cylinder. Make sure that the cylinders are concentric and that the emergence hole is not obstructed. The emerging moth can then climb one of the screen strips and expand its wings while resting on the wall or the ceiling of the cage.

Add water to the saucer just often enough to keep the soil slightly

damp, with brief intervals of complete drying between waterings. Knowledge of the natural habitat—such as marsh or desert—may dictate use of more or less water. The goal is to provide photoperiod, temperature, and moisture consistent with natural conditions at emergence time. For desert species, just before the expected emergence time, cap the outer cylinder with a plastic cover for a few hours each day to fog up the interior; this may break diapause. For temperate species, pupa-pots are best stored where they will cool down in winter, but where they can be easily inspected daily at emergence time. Line them up on a shelf at eye level, with white background, in a place where you can easily inspect them repeatedly—an easy way to keep track of emergences.

A more elaborate cage built by Rummel many years ago is depicted in Figure 6–16. It is constructed of 6–9 mm ($1/4$–$3/8$") soft wood (not plywood), and metal screening. The bottom bin is lined with tar paper thick enough to produce a slight ledge on which rests a removable deck of aluminum or of glass with a sheet of blotting paper on top to exclude light. A right-angle isosceles triangle about 4 cm ($1\,1/2$") on the short sides is cut from one corner of the deck, and through the resulting hole moths emerging from a layer of peat in the bin below climb to the cage above, where they spread their wings. The top of the cage has a layer of muslin, and a layer of screen, capped by a wood frame. To provide access to the cage, one of the longer sides is made as a fully removable door, held in place by rubber bands or string. Such a cage that I built 58 years ago is still in regular use.

Removal of an insect from an emergence cage or box calls for foresight. It must be given time for its wings to expand and several more hours for them to harden. At that point the insect is ready to fly and may do so abruptly if disturbed. Times vary with species. Dim light helps to keep butterflies settled down, so that they can be walked onto a finger. The larger moths, and many of the noctuoid moths, require a warm-up period of wing quivering before taking off. Catocaline moths are spooked easily even at relatively low temperatures. Opening the cage in a small, uncluttered room (such as a bathroom) can be a worthwhile precaution. "Microlepidoptera" can be captured from the emergence cage by allowing them to fly out against the inside of a window pane, where they can easily be covered

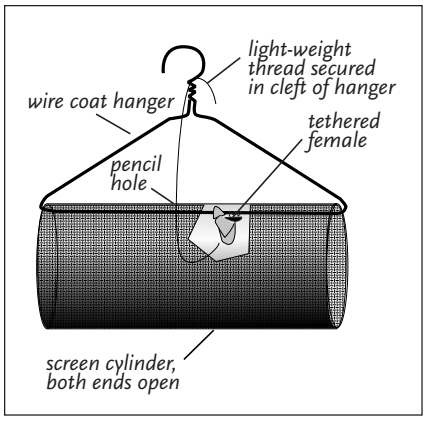


Figure 6–17. Mating cylinder.

by an open collecting vial or killing jar.

Some clearwings, such as the *Hemaris* hummingbird sphinx moths, eclose with a light covering of scales on the portion of the wing membrane that will be transparent. If you wish to retain a specimen in this condition you must pick the moment when the wings have fully expanded and hardened, but before the moth does any warm-up vibrating or flying. A few seconds of wing motion dislodges all the scales from the clear areas. Spreading (Chapter 8) requires great care: manipulate entirely with fine pins; cover carefully with static-free strips, avoiding all sliding motion.

8. Inducing Mating

You may at times be interested in mating reared females, to continue rearing a preferred species year after year, or to rear larvae from an individual acquired as a cocoon. Since the two sexes in moths, in particular, are drawn together by species- or genus-specific pheromones emitted by the female, "tying out" an unmated female often attracts males from the surrounding area. This practice is especially useful with saturniids.

To tether a large female moth, hold it from the front, with your index finger on top of the thorax, and grasp the sides of the thorax with your thumb and middle finger. Keep the wings folded up over the back. Tie a lightweight thread encircling the thorax between fore- and hindwings (or between abdomen and thorax—Tuskes et al. 1996). The tether knot should be at the moth's back so the legs will not get tangled in the thread. You can then tie the moth out among shrubbery, unprotected, or within some type of enclosure to protect it from predators.

One enclosure style (Figure 6–17) is an open-ended screen cylinder, diameter about one-third its length, suspended from a branch in a horizontal position by a wire coat hanger. The free end of the tether is run up through a hole (made with a pencil point) in the center of the roof of the cylinder and wedged into the top of the wire twist at

the top of the hanger, the length being adjusted so that the moth can reach the entire circumference of the cylinder but cannot quite reach either end. A responding male will mate within the cylinder, then depart after sometimes many hours in copula. If the female is not retrieved and freed from the tether soon thereafter, she may begin to lay eggs on the screen (not a problem).

Another shelter is an inverted peach-basket hanging from a tree branch. The tether is attached inside the basket and is adjusted so that the moth cannot walk quite to the edge.

Worth (1980) devised a more elaborate tether, tricky to make but reusable, allegedly easy to use, and most important, tangle-proof (Figure 6–18). Required materials are 5 kg (12 lb) test nylon casting line (might dental floss do equally well?), small fishline swivels, beads such as used to adorn moccasins, spring clothespins, small flat-headed nails, paper clips and—the critical item—thin but stiff brass or plastic tubing with a bore of ca 1.75 mm ($1/16$") (hobby shops).

1. Using a needle of workable size, pass the line successively though a bead, a 4 cm ($1\,1/2$") length of tubing, a second bead, then, leaving a free loop about 8 cm (3") long, back through bead #2, the tubing, and bead #1, leaving loose ends 5–8 cm (2–3") long (Figure 6–18).
2. Place the loop around the thorax of the female moth between the two pairs of wings and behind the middle pair of legs; slide the tube and beads down to touch the back of the thorax, pull the ends of the line gently tight, pass one loose end through one eye of a swivel, and tie the loose ends together to hold the swivel against bead #1. Pass a paper clip through the other swivel eye.
3. Drive a flat-headed nail partly through one jaw of the clothespin so

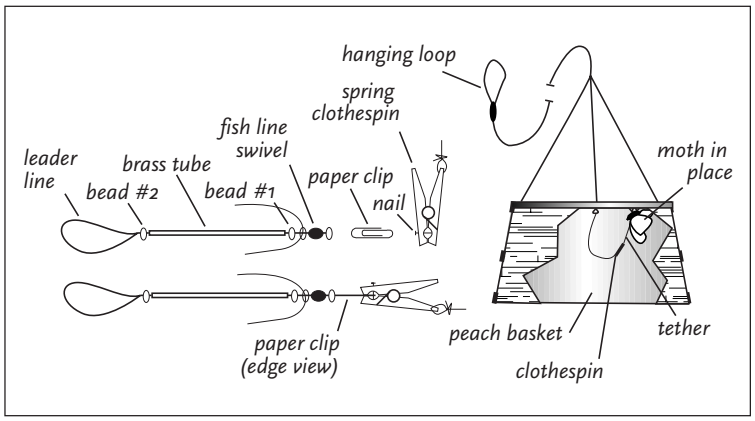


Figure 6–18. Tether design and basket setup (not to scale).

that it just crosses the gap made for the clothesline. Loop the other end of the paper clip over the point of the nail and grip it inside the clothespin jaws.

4. Using a piece of string, attach the clothespin at the center of the wooden bottom inside the above-mentioned inverted peach-basket or other shelter, in such a position that the moth can cling to the inside.

In routine use, you will leave the clothespin and its string attached to the shelter, and remove or replace the moth and her tether at the juncture of the paper clip and clothespin. Release the moth by carefully cutting the tether line. Trying to extricate the moth without cutting the line could injure her, and in the words of the inventor, "After a bit of practice I was able to prepare a harness in only a minute or two and to apply it to a moth in an equally brief time."

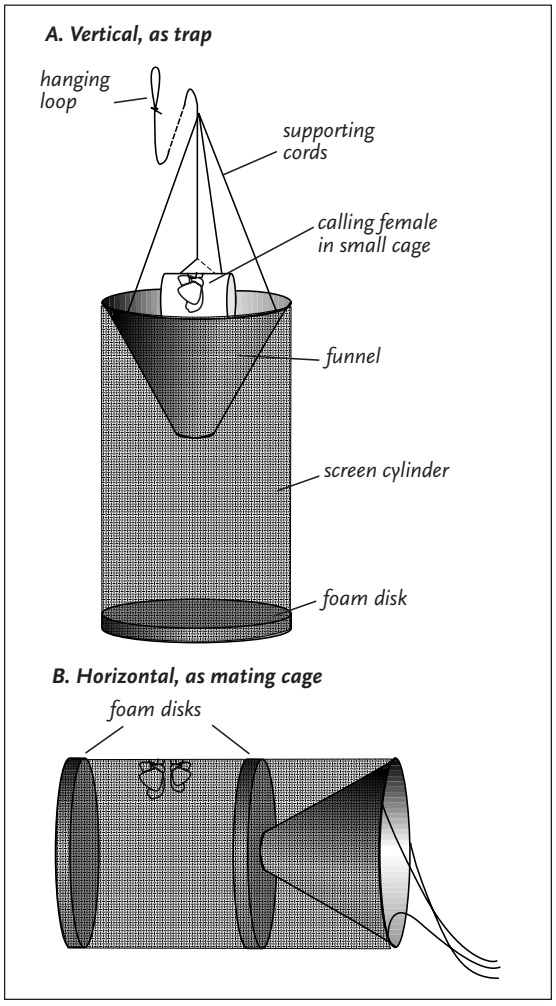


Figure 6–19. Calling trap–mating cage.

A predator-proof enclosed calling shelter can be made from a large coffee can with the metal bottom cut out. The bottom is replaced by 1.3 cm ($^1/_2$") hardware cloth soldered in place. The can is suspended in a horizontal position from a piece of oat-hanger wire attached at the center of gravity of the can. An untethered virgin female moth is placed so as to perch on the inside of the hardware cloth, the can is closed with the original plastic lid, and the cage is suspended about 1.5 m (5') from the ground. Copula-

tion occurs through the screen, and eggs may be laid on it. If necessary, the setup can be left unattended for as long as a week (Miller & Cooper 1976).

M. Collins (pers. comm.) used an inverted variation of the conventional cylindrical bait trap (described in Chapter 7, Section 4) to retain attracted males and protect them from predators (Figure 6–19). Close the bottom with a soft plastic foam disk (held in place by friction), and in the top place a 25 cm diam (10") stemless funnel of smooth plastic, with an outlet of 5 cm (2"). Suspend the trap from three wires attached to the top rim and passed through holes in the funnel margin. Place a calling female in a small screen cage hung just above the funnel. Responding males slide down through the funnel and are retained in the cylinder below. Recover them by removing the bottom disk.

This device, laid on its side, can double as a mating cage (Figure 6–19). Push the plastic disk up near to the funnel outlet, place an unmated pair inside the trap, and close it with a second foam disk.

If still or video photography of responding males is a goal, as with daytime calling gypsies, *Callosamia promethea*, *Anisota*, or *Hemileuca*, then exposure of the calling female within a screen emergence cage is the best approach. Dozens or hundreds of males may respond within a few minutes. If the female is accessible, then the first male to arrive may quickly terminate the action. On the other hand, a calling female may be placed on a tree trunk or bare stick so that the entire sequence can be recorded. Usually she will not fly away.

Cage-mating often occurs spontaneously if multiple individuals of the same species emerge at the same time. The mated female should be watched and removed for egg-laying when copulation has terminated. An unmated caged female may eventually lay sterile eggs. The likelihood of successful cae mating varies with specific groups. Most saturniids and many geometrids perform readily. *Catocala* are outstandingly difficult (Sargent 1976), and many other noctuids are likewise uncooperative.

If you are rearing a particular species year after year and are using siblings for breeding stock, two possible outcomes deserve consideration. The inbreeding may increase the frequency of a color or pattern variation, and this might be an intended goal. On the other

hand, inbreeding also gradually reduces female fecundity and the viability of ova, so the line may peter out. Induced mating with wild males, annually if possible, is an effective way to maintain vigor.

Live storage for short periods may be useful at times. If you have a freshly emerged adult and are awaiting the emergence of the opposite sex, refrigerator (not freezer) storage, in darkness, is effective. Place it in a glassine envelope in a closed plastic container, or loose in a small jar, in either case with a wad of damp tissue to prevent desiccation. Feeding before storage is usually not necessary or even desirable (Shapiro 1982), but it can be helpful with some small noctuids. The insects are usually good for mating as much as 7–10 days later, with extremes of over 7 weeks (or 6 months for Xylenini!). This practice is also useful if you wish to hold a virgin female for field calling at a later date.

Hand-pairing of reared Lepidoptera is feasible, and is used particularly for papilionids, pierids and larger nymphalids. Clarke and Sheppard (1956) described the following procedure: First the butterflies were warmed in a cage for 15 min. at 18–27°C (65–80°F). They were then held one in each hand with the wings folded over the back and the ends of the abdomens touching. Slight lateral pressure on the abdomen of the male caused wide opening of the claspers, which then grasped the terminal segment of the female. Correct positioning was enhanced by the manipulator rotating the ends of the abdomens slightly against one another. After a few minutes "locking" occurred, slight tension did not separate the abdomens, and the head and thorax of the male became motionless. The pair usually remained in copula for 30–45 min., during which time thrusting abdominal movements by the male indicated that mating was proceeding.

A variation found effective for nymphalids (Platt 1969) involved stunning the male and female to be paired in a large sodium cyanide insect killing jar for 1–5 min. at room temperature, and then everting the male claspers by applying slight pressure to the lateral and ventral portions of his abdomen ith thumb and forefinger. Meanwhile the female was held in the other hand, and her abdomen was curved downward so that it was exposed beneath the wings. The butterflies were positioned so that the male could clasp the female's abdomen one segment in front of the ovipositor. During this procedure the male was moved constantly in a small circle to stimulate eversion of

the genitalia and clasping. While still unconscious the pair was placed on its side with the male's abdomen positioned at about 135° in relation to the female's. The male involuntarily opened and closed his valvae, thereby inserting the aedaegus and initiating the contractions of copulation. Once the male had clasped the abdomen of the female and began to recover from the cyanide (usually in 4–7 min. after stunning) the pair was placed on a piece of moist cellulose sponge set on a paper towel, and the male's hindwings were positioned inside the female's with a pointed probe. The pair was then covered with a transparent plastic box, above which were placed incandescent and fluorescent lamps, and the butterflies were left undisturbed while recovery and mating proceeded. Platt (pers. comm.) has since eliminated stunning the female, in contrast to the original report,

When attempts at hand-pairing and cage mating failed with *Phyciodes tharos* because of male indifference and female rejection, Oliver (1979) noted that males escaping onto the inside of a large screened window became sexually aggressive. When brought in contact with a female held with wings raised over the back by flat forceps, copulation often occurred, but if the female was released from the forceps she shook off the male. Clamping the forceps in a spring clothespin and suspending the pair by resting the clothespin on the mouth of a small jar allowed copulation to be maintained. Oliver suggested this approach might be widely applicable to other butterfly species.

9. *Inducing Egg Laying*

Procuring ova from mated gravid females can be anything from routine to supremely challenging. Success can vary strikingly from group to group, and even from species to species. Females of some species begin to lay within minutes or hours after mating, whereas others may hold off for days or weeks. Wild-caught females are almost always mated, especially if their wings show any wear. They may lay eggs for only a day or two, or for several weeks.

Retain and spread the egg-layer and label it with the rearing lot-number, so that it can serve as a voucher for the identity of the larvae. Labels for retained immature and adult rearing specimens are described in Section 15.

Species that take nourishment as adults need to be kept fed and hydrated (Saturniidae are the major, but not the only, examples of species that do not feed as adults). A feeding solution of 8–10% cane sugar or honey in water is commonly advised. However, many noctuids overimbibe on this weak solution and become dangerously bloated; they do better with a 30–50% solution of maple syrup or honey in water (L.F. Gall, T.L. McCabe, D.L. Wagner, pers. comms.). "Trial and error" may be necessary. Also, addition of egg albumin (20% by volume in the feeding solution) seems to increase egg production (L.F. Gall, D.L. Wagner, pers. comms.).

The solution can be offered on a feeder made from a rolled-up piece of tissue or paper towel, a piece of sponge, a cotton ball or a dental "cotton roll," perhaps obtainable from your dentist. If you are using small containers, a small folded piece of paper towel moistened in the solution can be held in place by the jar cover. The feeder should be recharged or replaced daily. The rolled towel or cotton is best recharged with plain water, since the original sugar is left behind as the solution dries. Moldy feeders must be replaced. *Catocala* females can be kept alive, vigorous and productive, for more than a month in this way.

Butterflies and geometrid moths can be hand-fed by holding the female by the wings (folded over the back) and allowing her to drink from a piece of sponge soaked with the feeding slution. *Incisala* and *Celastrina* will usually feed themselves from cotton balls. Often, touching the feet to the sponge is enough to stimulate feeding, but if this fails, it sometimes helps to uncoil the tongue gently with the tip of a pin to bring it in contact with the solution. If you have a number of females to feed, it can help to use a holding device (Figure 6–20). Glue a 2 cm (3/4") disk of cardboard to the inside of each jaw of a spring clothespin, and attach the clothespin to a length of 3 mm (1/8") solid-core solder with its other end fixed in a hole in a block of wood. A butterfly with wings over back and clamped carefully with the clothespin can be positioned precisely over the feeding pad by bending the supporting wire (Cornelius 1989).

Many of the large moths oviposit readily if enclosed in an empty paper bag kept in normal temperature and photoperiod and protected from daytime sunlight and overheating. Some sphingids and many *Catocala* benefit from having leaves or bark of the foodplant included

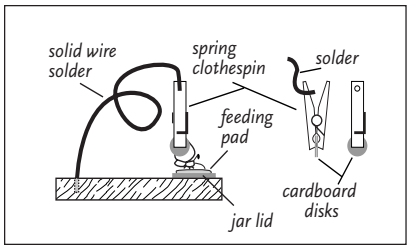


Figure 6–20. Holding device for feeding.

in the bag, but for the latter a crumpled paper towel will often suffice.

Many of the small *Catocala* with yellow hind wings hide eggs in the internal glued seams of the brown paper bag. To find them, cut the bag open, avoiding the seams, and hold the bag up to the light. If you search a seam by bending or peeling it open you will usually rupture the eggs.

To harvest eggs laid on paper towels or bags, cut out snippets of paper bearing the eggs, rather than trying to separate the eggs from the paper.

Smaller moths may also perform in small paper bags, or in glass jars containing a 2.5 cm (1") strip of white paper for oviposition (Godfrey 1972). Some also lay eggs on the piece of folded paper towel used to provide moisture and sugar. Useful approaches to specific groups are detailed below.

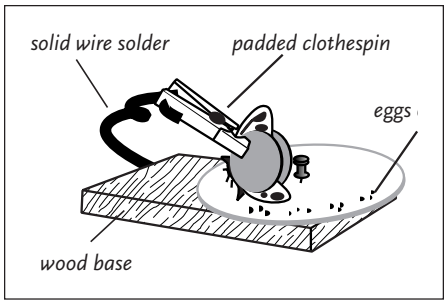


Figure 6–21. Device for egg-laying (Cornelius 1989).

An egg-laying device for saturniids (when you wish to prevent damage to the egg-layer's wings) utilizes a 10 cm (4") diameter disk of rough cardboard (such as from the back of a pad of ote paper) pinned through a central hole to a block of wood (Figure 6–21). The hole must be large enough so the disk can turn freely. A holder (described above for feeding a butterfly) allows placement of the gravid female, clamped with wings folded over her back, so that her feet touch the outer part of the rotatable disk. She then lays eggs in patches as she turns the wheel, and the next morning she can be recovered as a good specimen (Cornelius 1989).

Hemileuca will usually oviposit only on twigs of appropriate caliber; some insist on proper foodplant, and some will use the right size twig of any plant. Some noctuids with very narrow foodplant selection will oviposit only with foliage or buds of that plant present in the enclosure.

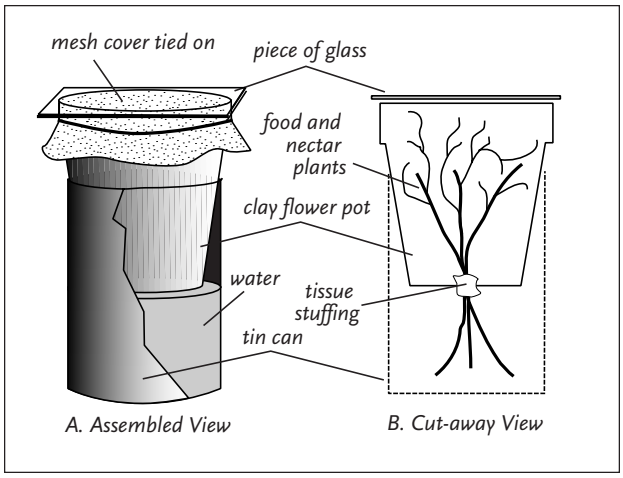


Figure 6–22. Clay pot for butterfly egg-laying.

With butterflies the conditions for inducing laying are often more elaborate. Larval foodplants (with stems in water to maintain freshness) are generally necessary, with enough present so that the insect comes into repeated contact with them. Nectar flowers or a sugar feeder are important. Light and heat, increased in a controlled manner and provided intermittently, are also necessary. Some species perform better if held a day without contact with the foodplant.

A very serviceable container consists of a clay flower pot placed in a tin can or jar filled with water up to the bottom of the pot (Figure 6–22). Sprigs of host and nectar plants are thrust through the drain-hole in the pot and any loose space in the hole is plugged with moist tissue. The pot is capped with netting held on with a rubber band, overlaid with a removable square of glass (to aid in temperature control). Light and heat are supplied by sunlight (glass off) or a lightbulb in a gooseneck lamp (glass all or partly on). Recommendations as to lighting periods vary, from 15–20 min. on and off to 1–2 hr. on and off. Size of container should relate to size of butterfly (F. Richard in Remington 1948).

A variation on the above, attractive now that clay pots are hard to find or expensive, is made from a transparent plastic food container with a mesh cover (Figure 6–23). A hole is cut in the side to receive the neck of a plastic bottle containing sprigs of foodplant (Fee & Boscoe

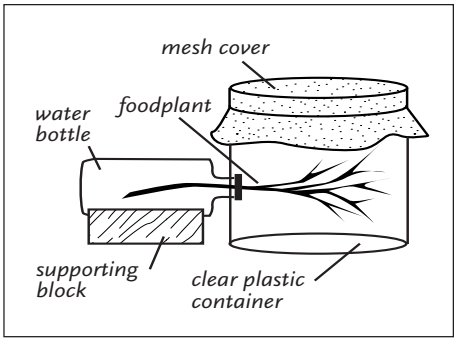


Figure 6–23. Plastic egg-laying pot.

1979). A wood block supports the bottle, and the spaces between stems and bottle neck should be stuffed tightly with tissue. For this design, exposure to low-angle afternoon sunlight was advised.

Some large species do better sleeved on the foodplant in a sunny situation. For monarch butterflies, an ideal egg-laying substrate is a tropical milkweed, *Asclepias curassavica*, enclosed in a mesh bag and placed in the sun. This plant continuously produces new growth on which the females prefer to lay eggs, and it is easy to manage as a perennial pot-plant in the North (Munger 1973). If aphids become a problem on the potted plants (when the plants are not hosting immature Lepidoptera) just set a small open container filled with ethyl acetate on the soil in the pot, then enclose the entire pot and plant in a sealed plastic trash bag overnight in a well-ventilated area.

10. Eggs to Hatchlings

The continuum of induced egg-laying to hatching deserves special attention both for ova of unknown parentage and for selected species. The latter can often serve as patterns for understanding and being successful with others.

Frequently you will obtain ova from a female of unknown identity, or from one whose life history is unknown to you. In such a case you can offer the emerging larvae samples, in a single container, of various foodplants that seem reasonable in relation to your knowledge of its habitat and of related moths. Offered leaves for spring hatchlings should be very young; species hatching later in the season may select mature leaves. With luck, the larvae will find one or more to their liking. One axiom that may be useful is that moths that lay "large" eggs, for the size of the moth, nearly always feed upon some tree, shrub or perennial grass; those that lay "small" eggs usually feed upon herbaceous plants, weeds, or annual grasses (McFarland 1964). But most tree-feeding early-spring hatchlings have small eggs, while hatchlings requiring mature leaves or utilizing conifers have large eggs (D. Schweitzer, pers. comm.). Such rules are clearly subject to geographical, seasonal, and taxonomic qualifications. Unknown hatchlings offered a smorgasbord merit extra attention the first few days, especially with regard to humidity. This means checking the container frequently to reduce or increase humidity as indicated by condensation on container walls, or leaves shriveling, respectively.

It is important to distinguish among:

- a foodplant on which a female lays its eggs in the wild, and
- a foodplant discovered by trial and error by the person rearing the larvae, or
- an acceptable alternate selected by the rearer because the original foodplant was not readily available.

In the first situation the selected foodplant is usually one on which the caterpillar can thrive, but occasionally the female makes a mistake, confused by either visual or chemical cues. The second and third situations merely represent acceptable food sources that may or may not coincide with a natural foodplant. Rearing notes should always indicate how the foodplant was selected.

If you plan to rear a species from another area and are unsure about having a suitable foodplant available locally, try to learn ahead of time the family and genus of the native foodplant or plants, then look in your local botanical field guides to find a species that is closely related. Staff at university arboretums are often willing to help upon request.

Artificial diets are often used in research laboratories to maintain a culture of a particular species in uninterrupted reproduction for many generations. Numerous recipes have been developed. One use of artificial diets, for the amateur, can be to feed hatchlings of a moth whose foodplant is unknown and is not discovered by the multiple sampling technique. If such larvae accept the artificial diet (as they often do), you can learn how the immature stages look. You can then recognize them if you see them in the wild.

One simple, tested recipe (Bergomaz & Boppré 1986) shown on the following page is based on ordinary white, dry navy beans, ground to fine flour with a coffee grinder (as for coffee filtering).

Place 3 mm (about 1/8") wide slices of the diet on a disk of paper towel in a petri dish, and introduce as many as 50 freshly hatched larvae. Provide fresh diet in fresh containers every three days. As the larvae grow, put smaller numbers into successively larger cages; slice the diet no wider than 5 mm ($^1/_5$"). This recipe proved satisfactory for many arctiids and for the various noctuids and lymantriids on which it was tried.

You can also employ this artificial diet to finish a late rearing indoors after outside foliage has deteriorated in the autumn. Adding

Navy Bean Diet

Navy Bean flour	*75.0*	*g*
Brewer's yeast	*17.5*	*g*
Ascorbic Acid	*3.5*	*g*
Cholesterol	*0.5*	*g*
Sorbic acid	*0.5*	*g*
Methyl-p-hydroxybenzoate	*0.5*	*g*
Streptomycin	*0.4*	*g*
p-formaldehyde	*0.15*	*g*
Agar	*15.0*	*g*
Water	*381.0*	*ml*

Mix the first eight dry ingredients and keep them separate from the agar. Bring the water to a boil, take it off the heat and stir the agar in until it dissolves; then stir in the dry ingredients and mix them thoroughly with an electric mixer. Pour the mixture into plastic dishes to a depth of 4 cm (about 1 1/2"), let them stand at room temperature 12 hours, then store them in a refrigerator; the material will stay in good condition for about five weeks. To make up smaller amounts than the above 500 g, measure in the ratio of 1 part agar, 6 parts dry mixture, and 20 parts water.

Chemical ingredients and agar are obtainable from some of the supply houses in Appendix L.

finely ground foodplant leaves when you make up the diet will often encourage feeding. You can grind the leaves earlier in the season, in anticipation of this need, and store them frozen.

The following are a few species for which effective strategies have been worked out for egg-laying and for getting the hatchlings started.

Celastrina ladon can be induced to lay in a setup consisting of a 200 ml (6 oz.) conical clear plastic drinking glass covered with a piece of nylon stocking held in place with a rubber band (Figure 6–24). Melt a 6 mm (1/4") hole through the bottom of the cup with a hot screwdriver, and melt a similar hole in the top of a 500 ml (1 pint) plastic deli container filled with water. Put a sprig of the foodplant bearing flower buds into the cup and push it to a position where the female, walking on the mesh cover, will repeatedly encounter the buds. Feed the female daily (but only two or three times weekly for most spring lycaenids). Use a petri dish with a disk of paper towel moistened with sugar solution; place the female on the paper and cover with a plastic cup. When she has fed, transfer her to the oviposition setup and place it in a sunny window. Females so treated lay up to 30 eggs a day for a week or more. Each day transfer the females to a new oviposition setup, and inspect the buds in the old one for eggs (usually placed at bud bases). If some are

present, put the setup aside for five or six days, then look for the tiny larvae with a hand lens (Oliver 1982a).

It is most convenient to rear these larvae in plastic petri dishes containing a paper disk moistened with one drop of water, and a few clusters of buds. Place a single bud bearing a newly hatched larva on each cluster. Protect stacked dishes from dehydration by enclosing them in Ziploc bags. Limit to four or five small or two or three larger larvae per dish and transfer to fresh foodplant every three days at most. Thirty to 40 days in the larval stage is usual. When the pupae are formed (usually on the foodplant or on the petri dish), move them to a screened cage for emergence. In this species, exposure of the larvae to abnormally long day-length from artificial lights can readily lead to adult coloring and markings different from those that would have occurred in the wild for the same brood.

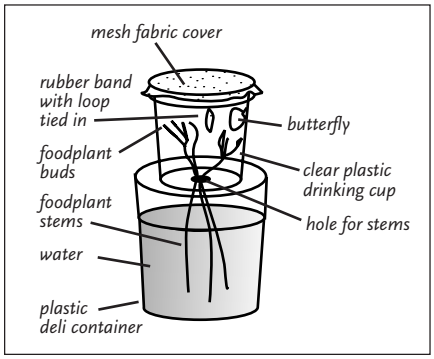


Figure 6–24. Celastrina egg-laying setup.

Species whose larvae feed on forest-floor detritus, such as *Calycopis,* will oviposit readily if confined over a layer of natural litter, with fresh flowers provided for nectaring. While *C. isobeon* has been found to thrive on a wide variety of fresh foodplants (*Wisteria* and *Oxalis* excepted), it does particularly well on vegetable and even insect detritus, preferring this to fresh leaves (Johnson 1985).

Mitoura and *Incisalia* will usually lay if enclosed in a 4 liter (1 gal.) jar with a vased foodplant and a muslin cover. Sleeving females on the foodplant is also effective. They tolerate group rearing.

Speyeria often oviposit readily in a mesh-covered bucket containing a violet plant and some small twigs or coarse debris, and exposed periodically to sunlight or a bright light bulb. Eggs are laid on the debris more often than on the plant. Depending on the age of the female when captured, egg-laying may begin at once or only after as long as a month or more—late-season captures usually lay promptly. Sometimes these females will even perform in brown paper bags exposed to light and warmth. Because egg-laying can continue over an extended period, it is important to feed the females once or twice

daily (Mattoon et al. 1971).

For *Cercyonis*, lay a liter (quart) jar on its side, scatter cut lengths of grass lengthwise in the jar, introduce the female, and close the jar with fabric mesh. The female should be fed daily. If she is very fresh, or newly mated, it may be a week before she starts to lay, thereafter producing 10–30 eggs per day. Short pieces of grass bearing eggs are snipped off and placed in petri dishes; hatching occurs in 6–12 days at 24–30°C (75–85°F) (see Section 12 for carrying these larvae through the winter in normal diapause). Larvae can be reared right in the petri dishes, or in other containers, with damp paper toweling in the bottom and stocked with short pieces of grass. Kentucky blue grass *(Poa pratensis)* is well accepted and keeps long enough so that the containers only need to be changed every three days (Oliver 1983). Rearing on potted enclosed plants of grass or sedge also works, although the very young larvae are hard to keep track of. Using artificial light to maintain a long photoperiod keeps the grass growing and also averts diapause.

Certain noctuid genera respond best to particular conditions (Godfrey 1972, McCabe 1991). *Phoberia* will lay under dead, peeling bark. *Leucania, Pseudaletia* and *Lithomoia* respond to hollow grass stems or the crevices about grass leaf sheaths. *Apamea* utilize flower heads of grasses—here, a caution: grasses selected for captive oviposition may already contain wild-laid *Apamea* ova; this could result in a mixed culture.

Some day-fliers need more space, and a close light source turned on and off at 15-minute intervals to raise the temperature in the enclosure 6°C (10°F) will trigger egg-laying. The eggs of these species, however, are adversely affected by the frequent temperature changes and should be removed as promptly as possible.

Elliptoid-eyed *Schinia* oviposit best in 4 liter (1 gal.) containers enclosing a few sprigs of buds and flowers of the foodplant placed in water in properly plugged vials. Other flowers, as nectar sources, should be placed in the jar; weak sugar solution (5%) sprayed on the foliage can provide a further source of energy. The sprigs of foodplant are removed (and replaced) daily and examined under a hand lens or dissecting microscope for ova. On hatching, larvae are reared individually because of a cannibalistic tendency. Young larvae feed only within the buds or flower bases. If the flower buds have heavy sepals,

it is best to open these with forceps so that the larvae can feed in the softer internal portions (Hardwick 1958).

Females should be provided with a jar proportional to the size of the moth for egg-laying. Place a few sprigs of foodplant (of assorted species, if the foodplant is unknown) in water in a small vial, and invert the jar over them on a base made of a stiff piece of cardboard on which a piece of white paper has been laid. Then tip the jar a bit so that the moth can be introduced under the edge. Keep the containers out of the sun and at temperatures close to natural. Inspect them daily for eggs (McGuffin 1981). *Stamnodes* and *Hydriomena* often need loose bark flakes on small dead stems. Some geometrines lay best on smooth surfaces, such as the sides or bottoms of a baby food jar. Several feedings may be needed over a 10-day period to get good egg production and avoid premature death of the female (McFarland 1988). You can often get good results by enclosing the female in a small jar with a crumpled wad of paper towel, or towel rolled tightly to simulate bark crevices.

11. Dealing with Diapause

In anticipation of those seasons of the year which are unsatisfactory for sustained growth or development of a particular species, Lepidoptera undergo diapause, a quiescent period that continues until conditions become favorable for further growth. Such conditions are a combination of changing day-length (photoperiod), temperature, moisture and availability of suitable food. Except in moist subtropical and tropical climates, where development can often continue throughout the year, most species spend one or more periods of the year in diapause. In summer this is termed estivation, in winter, hibernation. Where moisture is adequate throughout the year, temperature controls both plant growth and lepidopteral activity. Where temperature is always equable, seasonal wet-dry cycles control. But in any situation, given temperatures conducive to lepidopteral activity, larvae must be ready to feed when palatable and digestible foodplant is available. The timing of diapause in any species' life cycle has evolved to accomplish this.

A few examples follow. *Hemileuca* and gypsy moth ova undergo full development of the embryo within the egg during the summer, diapause at that stage, and are ready to hatch with the advent of warm weather and bud-burst in spring. Many geometrid larvae hibernate in

the middle instars, then may even use a different foodplant the following season. Fully grown dusky wing *(Erynnis)* larvae of the last brood overwinter in the leaf shelter they have been inhabiting, then pupate within it the following spring. Most sphingids pass the winter in pupal chambers in the soil or under leaf litter. Satyrids at high latitudes or high altitudes go through two winters as hibernating larvae before experiencing enough warm weather to complete their development. Some *Hemileuca* and other saturniids, instead of eclosing in the first autumn, will remain in pupal diapause and emerge right in season one to several years later, as will *Alypia* and some sphingids. These should be maintained in as near to a natural cycle (for their place of origin) of light, humidity, and temperature as can be attained. Some nymphaline butterflies hibernate as adults (for example, mourning cloak and red admiral), and the monarch makes a two-way migration to warmer latitudes to overwinter. Varied strategies of spring-hatching larvae are detailed below. Each of these patterns must be considered and the appropriate one accommodated in your rearing.

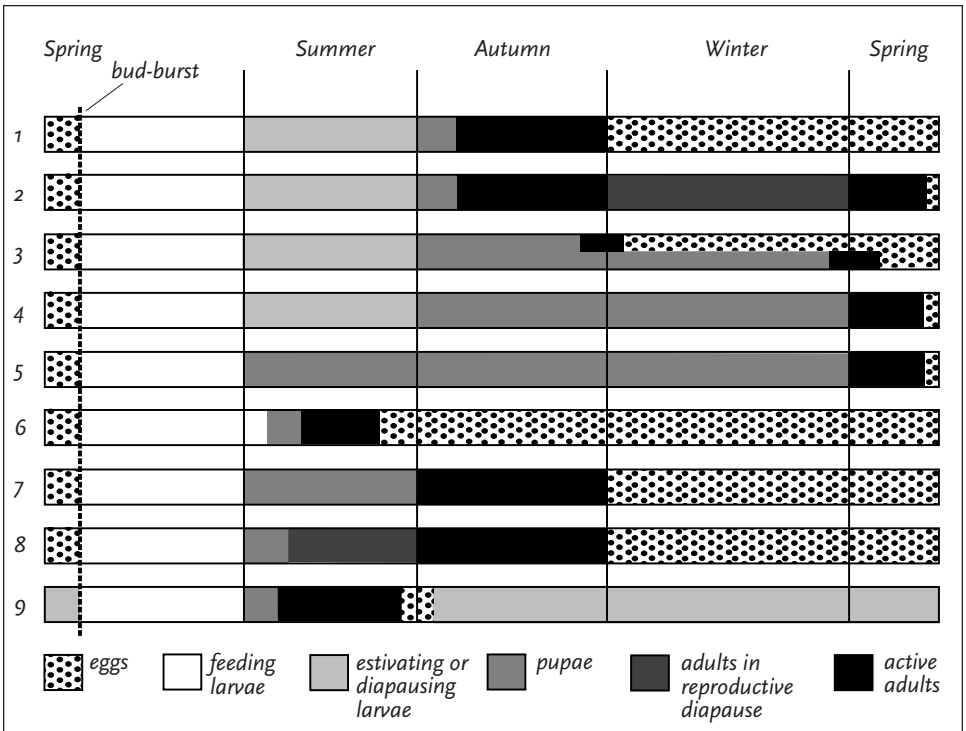
In areas where winters are mild, many species, instead of diapausing as mid-instars, will feed on buds intermittently throughout the winter, whenever the temperature exceeds 4–7°C (40–45°F). *Campaea, Protoboarmia, Nemoria lixaria, Abagrotis, Anomogyna, Rhynchagrotis,* and *Colias eurytheme* follow this pattern. You can maintain them in uncrowded sleeves or in secure enclosures on the ground.

If temperate or cold-climate species are not given many weeks of exposure to cold, they may not be triggered to break diapause, and development will not resume. If you are rearing northern species in a southern clime, providing some time in the refrigerator will allow for continued development for the hibernators—for example, 8–10 weeks at 6–15°C (40–60°F) for *H. cecropia* (Tuskes et al. 1996). Northern strains can tolerate lower temperatures than this, but those of southern origin should be kept above freezing.

12. Spring Hatching and Hatchlings

Early spring hatching from ova that have overwintered, or were laid in spring by hibernated adults, is a strategy occurring in many species of temperate and colder-zone Lepidoptera. They are generally

single-brooded and thrive only on fresh young foliage. Almost all are tree and shrub feeders, and they must complete their feeding before the foodplants produce enough chemicals to make the leaves indigestible, usually in a period of four to six weeks. Once the larvae have attained full growth, they then (according to species) undergo one or two periods of prolonged quiescence (diapause), which may occur in one or two of the developmental stages: larva, pupa, adult, or egg.



Examples

1. Some sallows— such as *Sunira bicolorago, Chaetaglaea sericea.*
2. Pinions—*Lithophane*; some sallows such as *Eupsilia.*
3. Fall cankerworm moth—*Alsophila pometaria.*
4. Spring cankerworm moth—*Paleacrita vernata.*
5. Quakers—*Orthosia*; half-wing looper moth—*Phigelia.*
6. Underwing moths—*Catocala.*
7. Linden looper moth—*Erannis tiliaria.*
8. Cooper underwing—*Amphipyra pyramidoides.*
9. Fritillaries—*Speyeria.*

Figure 6–25. Strategies for exploiting earliest spring foliage.

After attaining full larval growth in late spring, different species split up the remainder of their life cycles in various ways, as depicted in Figure 6–25:

- Adults becoming active or emerging in the autumn lay eggs then (Example 1), or go into hibernation soon and lay eggs in early spring (Example 2).
- Each species uses just a single strategy, although with some, part of the brood may emerge and lay eggs in late autumn, the remainder emerging in late winter (Example 3).
- Early or late summer pupae may remain in diapause until spring (Examples 4, 5).
- Early summer pupae may develop through adult to egg during the summer (Example 6).
- Larvae pupate at end of feeding, then go into diapause; adults emerge in autumn, mate and lay eggs (Example 7).
- Summer-emerging adults fly but enter reproductive diapause and remain sexually inactive until autumn, or in some species until the following spring; they then mate and lay eggs (Example 8).
- Larva hatches at end of summer without diapause, then hibernates as unfed larva until bud-burst the following spring (Example 9).

Eggs in many of the above instances undergo complete embryonic development within the egg soon after being laid, then remain in diapause until bud-burst.

"Bud-burst" ordinate dating in Figure 6–25 varies with latitude and climate, and with the onset of the growing season in your area in a particular year. The dates and duration of the four seasons thus vary from place to place and from year to year. This phenological calendar, punctuated by the blooming time of index plants, relates the timing and duration of events to one another as they actually occur in nature. It is far more useful in understanding Lepidoptera than is the temporal calendar, with its rigid and regular intervals. (Convention requires, however, that we use the temporal calendar for all notes, records, and labels.) Where winters are mild, behavior may vary, with moths laying eggs or larvae feeding intermittently in clement weather.

A different sort of phenological calendar occurs in lower latitudes where the major variation in the climate cycle is wet–dry rather than

hot–cold. Egg-hatch is timed to coincide with the flush of foodplant new growth, and the interval between larval maturity and the next egg-hatch is split into quiescent and active stages in various ways, according to species.

Learning about or working out these variations is part of the allure of rearing. Knowing the many possibilities can increase your success.

Schweitzer's emergence box described in Section 7 is an ideal container for larvae that estivate in soil, as are many of the smaller, covered containers. Important considerations are:

- Keep the containers cool and in subdued light.
- Monitor moisture to maintain a middle ground between desiccation and mold development. The natural conditions in the native habitat of the species being reared are a guide to the level of moisture required.
- Include a rough surface for clinging while wings are being expanded.

Larvae that run short of food in late instars, and some reared on cut foliage that do not appear malnourished, may cut short or omit estivation and emerge in mid- or early summer.

13. Managing Hibernation

Successful rearing often hinges on proper understanding and handling during the winter. The handling of ova has already been considered. Hibernation of adults is not usually part of a rearing project. Larvae, and sometimes pupae, are the challenge, as discussed below.

While one recommendation is to overwinter larvae out of doors in as near natural conditions as possible, they also can be carried through in tightly sealed containers in a household refrigerator (Oliver 1982b). Using 11 x 11 x 15 cm ($4^1/_2$ x $4^1/_2$ x 6") plastic deli boxes, put 4 cm ($1^1/_2$") of moist soil in the bottom, just moist enough to stick together when squeezed in the hand. A broken 100 mm (4") plastic petri dish is placed on this, then petri dishes containing larvae are stacked on top, as many as will fit. The lid of the deli box is applied tightly; storage is in the refrigerator at 3–4°C (38–40°F). Viability should be 60-80% for as long as six to eight months.

For *Speyeria* and those *Cercyonis* that diapause immediately after hatching, a recommended approach is to examine the eggs daily,

allow newly hatched larvae to finish eating their eggshells, then transfer the tiny caterpillars by means of a fine camel hair brush to a petri dish lined with a circle of paper towel moistened with one drop of water (Oliver 1982b). Twenty or so larvae are put into a dish, which is then put into the plastic container and capped tightly. After allowing a week's time at room temperature for larvae to choose resting sites, the container is moved to the refrigerator.

In the case of satyrids that diapause as partly grown larvae, gradual cessation of feeding over a period of two weeks indicates the onset of diapause. The larvae often settle on the side or roof of the container, and if petri dishes are being used, they may be stored without being disturbed. Any individuals resting on vegetation must be gently persuaded to move onto the inside surface of the container. All vegetation must be removed to discourage mold growth.

Because of the adverse dehydrating effects of mechanical refrigerators, the use of a large portable ice chest has been recommended (Brown 1965). If there is a drain hole, a thermometer in a drilled cork is placed in that, or through a hole drilled near the bottom of the sidewall. Larvae are placed singly into shell vials (previously sterilized with boiling water) closed with a plug of absorbent cotton, with a label inside the vial; the vials are placed in racks in the ice chest. Cooling and humidity are initiated by 10 ice cubes placed free in the chest, and then 5 kg (10 lbs.) of ice in a sealed plastic bag is added. From then on, ice bags are added from time to time to keep the temperature below 2–3°C (36–38°F), plus a few loose ice cubes if there is no free water left. Nymphalid larvae that feed gregariously in early instars are carried over best in groups rather than singly.

Another method for handling diapausing *Speyeria* larvae utilizes wooden blocks (Mattoon et al. 1971). A 1.5 cm ($^1/_2$") hole is drilled, with the grain, through about 4 cm ($1^1/_2$") cubes of basswood or other soft lumber. To kill mold spores, blocks are sterilized in an autoclave (a vegetable steamer or pressure cooker should be effective). Disks of very fine nylon chiffon, stapled over the holes, are used for closure. Newly hatched larvae, up to 100 per block, are transferred into the hole and the end netting is fixed into place. The blocks, on their sides, are then soaked until partially wet in distilled water not deep enough to enter the holes. Blocks are then stored in the vegetable pan of a refrigerator at temperatures above freezing. They are inspected

weekly for possible resoaking and for mold. Living larvae in a moldy block should be transferred to a new block. When the reference article was written, frost-free refrigerators, which are very dehydrating, were not in common use. More frequent checking is necessary with a modern machine.

Reared nymphalids that diapause as half-grown larvae, such as *Euphydryas*, *Chlosyne, Phyciodes*, or *Clossiana*, usually settle down in clusters in a curled leaf (Oliver 1982b). These can be nudged off the leaf with a brush, into a petri dish prepared as for freshly hatched larvae, and given a week at room temperature to settle down before being refrigerated. When these larvae are ready to resume feeding after diapause, it is sometimes necessary to expose them to sunlight to inspire them to do so. One-half hour exposure, with due precautions to avoid overheating the rearing container, has proven helpful (Evans 1950). Larvae of such species can often be found sunning themselves in the wild in early spring, and in captivity they need sun exposure for optimum health. *Asterocampa* larvae can be carried through their diapause, from midsummer through the winter, in dried curled leaves in closed, top-perforated containers with paper toweling in the bottom. In the warm months spray them monthly, air dry, and re-close the containers; withhold water in the cold months.

Noctuids that overwinter as larvae usually do so below ground. However, many cutworms feed intermittently in winter on herbs or dead leaves. House them in flower pots with muslin covers and plenty of food and shelter (rolled dead leaves). Place the pots out of doors, exposed to rain all winter. The larvae usually tolerate ice and cold well.

Large larvae, such as arctiids, are best handled in a coffee can containing some damp curled dead leaves (Oliver 1982b). Scatter the larvae through the leaves, then seal the can tightly with its own cover and refrigerate.

Geometrids that overwinter as partly grown larvae should be observed for cessation of feeding. Then transfer them to clean dry jars containing a few leaves of foodplant but no excess moisture. Close the lids securely. Place multiple jars in a box, separate them with crumpled paper to avoid breakage, enclose the box in a plastic garbage bag, and bury the bag in a hole by the north side of a building or in a shaded area in woods. When the snow has melted and the

foodplant has begun to leaf out, dig up the jars (McGuffin 1981). Ova and pupae can be handled in the same way.

When *Euchlaena* and similar larvae diapause in the autumn, place them in containers with a deep layer of leaf litter; water them occasionally on warm days during the winter. They will need food promptly when they break diapause in the spring. In mild climates the containers can be placed in a styrofoam cooler on a shaded porch.

If you wish to try carrying partly grown "microlepidoptera" larvae through the winter, Braun (1950) suggested a box made of square-meshed galvanized screening (hardware cloth), lined on the inside with window screening to exclude earthworms. This is placed on the ground, elevated from the surface by a few pebbles, in a place never reached by the sun. The livestock are housed in jelly glasses capped with heavy muslin, tied on; these are then placed, inverted, in the hibernating box. Moisture control is automatic.

To terminate diapause in larvae stored in petri dishes, remove the container from the refrigerator, add a drop of water to the paper liner in the dish, a few sprigs of foodplant, and stack the dishes in sealed 28 x 28 cm (11 x 11") Ziploc bags at room temperature and light (Oliver 1982b). Dishes should be examined every two or three days to remove feeding larvae to new containers, and to remove any dead larvae or moldy vegetation; mortality is often highest just at this point.

14. Manipulating Diapause

Diapause can be avoided in many overwintering larvae by rearing them indoors with lights on for 18 hours a day (Oliver 1982b).

Speyeria larvae, newly hatched, have been induced to omit diapause by using an effective but tedious method (Grey et al. 1963). When the larvae have settled down, transfer each by camel hair brush to small bits of paper towel soaked with distilled water. When the larvae crawl away, transfer them immediately to a violet leaf, where they promptly go to sleep again. This disturbing has to be done daily for many days to be successful. In one group of larvae, first feeding occurred as soon as 11 and as late as 31 days after hatching. Thereafter, development is continuous without special attention. Diapause can be broken after variable storage periods (Mattoon et al. 1971) by using a petri dish on a white surface with a soaked but not flooded

disk of paper towel in the bottom, and a young tender violet leaf. A 60-W incandescent bulb is positioned 20 cm (8") above the leaf. Up to 30 larvae are placed in the container on the wet paper. As each begins to move about, it is transferred to the leaf, and the cover is put on the dish. Feeding may occur in as little as 30 minutes, but usually in one to three days. Use of a ring of expanded foam plastic to surround the sides of the petri dish will insulate and reduce condensation on the sidewall.

Larvae of *Actias luna* reared in a photoperiod of 16 hours or more of light per day will usually skip diapause (Wright 1967). Cocoons of nondiapausing *A. luna* tend to be nearly white, as opposed to brown in overwintering cocoons, making separation possible. This difference can occur naturally in the border zone between single and double-brooded populations.

An example of a desert moth that may seem to "stall" late in the pupal stage was described by McFarland (1966). *Euproserpinus phaeton mojave* that had developed to the point of showing wing markings through the pupal case failed to eclose until the plastic box in which a pupa was lying on damp sand was placed in the sun. The box steamed up, the moth emerged immediately, and then, astonishingly, expanded its wings by raising them straight up over its back. Wing expansion was replaced by drooping when sunlight was interrupted, but resumed with restoration of sunlight. Pupae not exposed to the sun died without eclosing. This observation underscores how much is to be learned about the biology of desert moths.

Pupae of almost any species being reared for emergence at a particular time (such as material for a school or camp nature program) can be held in a refrigerator for delayed emergence. In a frost-free refrigerator, frequent spraying is advisable to avoid desiccation, or they may be placed in a sealed plastic bag. The amount of lead time out of the refrigerator before emergence varies with species and can only be learned by experience, but *Actias luna* may emerge after only a few days, *Samia cynthia* after four to six weeks. In one study (Seeley 1966), 85% of cocoons of *Antherea polyphemus*, *Callosamia promethea, Hyalophora cecropia*, and *H. gloveri* removed from refrigeration and held at temperatures 18–27°C (65–80°F) emerged in 20–30 days.

In some saturniines *(A. luna, Saturnia mendocino, A.*

polyphemus) diapause is altered in the pupa by photoperiod manipulation. Maintaining long day-length triggers adult development; imposing short day-length induces diapause.

15. Keeping Records

The purposes of rearing include obtaining intact adult specimens, preserved larvae (photographs of larvae are of minimal value in classifying micro- and many macromoths) and pupae, learning details of life histories and foodplant specificity, etc. So that you can relate specimens, photographs, and notes permanently and unambiguously to one another, you should define each rearing lot with your name and a unique number indicating the year the batch was initiated and a sequential number for each successive new lot within a calendar year. An example is "J. Powell No. 89F44"—year 1989, month F (for June: A=Jan., B=Feb., etc.), and the 44th lot of the month. If a lot is subdivided, as for rearing under different conditions or on a different foodplant, or to distinguish parasitoids derived from the lot, the number becomes "89F44.1", "89F44.2", etc. Placing the year first and using a letter for the month simplifies chronological sorting in a data base. This number must be documented on every specimen (ova, preserved larvae, pressed larval mines, pupae, parental or offspring adults), photo, and log and data entry related to that lot.

Figure 6–26 shows examples of labels for reared specimens. They are used to clarify the distinction between a foodplant on which ova were laid or larvae were found feeding in the wild, and food accepted by the larvae as they were being reared to maturity. Be very careful with specimens collected as cocoons. The larva may have walked from the plant on which it fed to a different species before spinning its cocoon, and your label should indicate "cocoon found on... " and not "reared from...."

Record-keeping for a rearing project ideally involves use of a flow chart into which is entered, on a daily basis, the progress of the larvae being reared. Basic information should include the order, family, genus and species of the organism (as far as known—but ultimately updated); developmental stage initially collected; date and locality where collected, and by whom; foodplant order, family, genus, and species; if parasitoids emerge, the order, family, genus and species; and dated comments, including the frame numbers of any photo-

graphs. This flow chart can be kept on paper, or on a personal computer with ultimate printout when work with a rearing lot is completed. Examples of a form for each medium are shown in Appendix A.

Many times an incorrectly identified foodplant has appeared in print, was quoted by subsequent authors, and eventually was accepted as fact even though unverified (Shields et al. 1969). If a foodplant record seems doubtful, it is worth retesting. Keep in mind that one species may lay eggs and feed on different plants in different locations. Negative results should be published as a brief note. If a utilized foodplant cannot be readily identified by the rearer, a proper specimen of the plant should be preserved for determination by a qualified botanist. Reference to the location of the specimen (in what herbarium it was deposited), and its reference number there should be kept with the retained Lepidoptera specimens.

A "proper specimen," in the case of small herbs, is the entire plant, including roots. For larger plants, save leaves attached to the stem (basal leaves plus stem leaves, if they are different, as in mus-

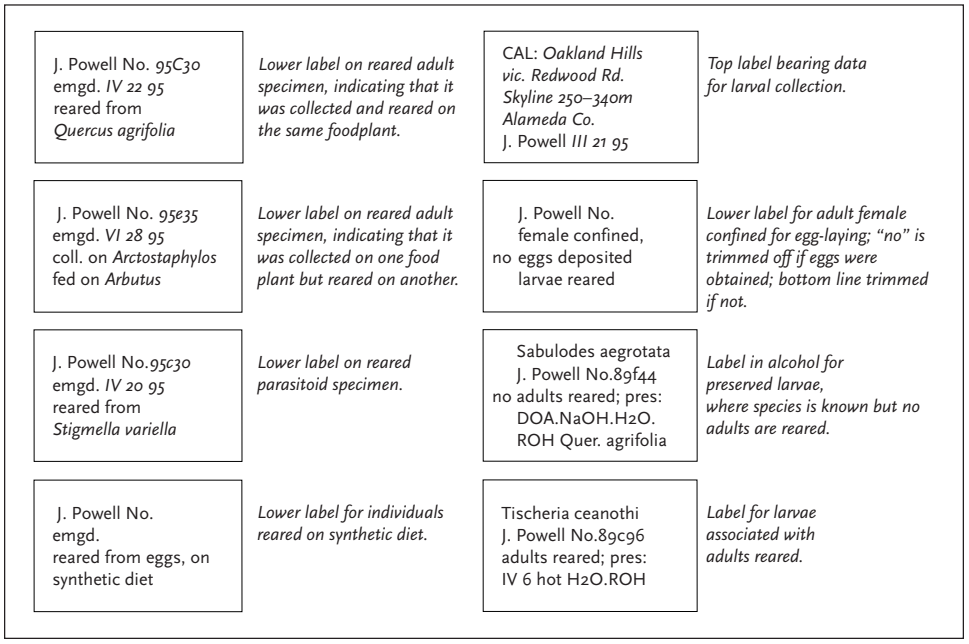


Figure 6–26. Labels for rearing-associated specimens.
(Plain type is preprinted. Material in italics is penned in by hand.)

tards), flowers, the fruiting body, all pressed and dried in a plant press (or a big phone book), together with details as to bark and habitus, if a tree. Also record full data as to locality, altitude in a mountainous situation, date, habitat, and soil type (Remington 1947).

If you do not have access to a university or museum with a botanical department, advice may be obtained from the Plant Specialists Index (Holmgren & Holmgren 1992), available in many technical libraries. The specialist should be contacted regarding willingness to do the determination before any material is sent.

16. Reporting Life Histories

There is no question that over several centuries much information on lepidopteran life cycles has been obtained by amateurs, only to be lost through failure to publish the information and thereby share it with other lepidopterists. As an amateur you may feel that your work is not significant, that everyone probably knows the information already, or that you could not begin to write an acceptable scientific article anyway. One way to resolve the first two concerns is to look in Tietz (1972) to see whether the life history had been published up to that time, and whether a foodplant used in your area is listed. Also look for information on early stages and foodplants in reference books and field guides. Other useful sources are Crumb (1956, western), Ferguson (1975, eastern), Forbes (1923–1960, eastern: Appendix J), Friedrich (1986, European: Appendix J) McFarland (1975, western), and Stone (1991, Appendix J). If your search is fruitless, contact the editor of the *News of The Lepidopterists' Society* saying: "I have worked out details of the life history of species 'x.' Is a report on this of interest?" The response would indicate whether the information is common knowledge, or whether knowledge of that species is incomplete or unknown and that a report should be submitted, specifying whether for the *News* or for the *Journal of The Lepidopterists' Society* .

The third concern—how to write such an article—is addressed in a piece reproduced in this manual (Aiello 1993) and found in Appendix A. Its author points out that records and notes are the backbone of scientific observation and that memory fades faster than the setting sun. Reliance on anything but notes, photographs and retained specimens is fraught with error. It would be worthwhile to reread

Aiello's article every spring at the start of the rearing season, just to brush up on record-keeping discipline!

Appendix B is an article written for this manual by Drummond on writing for the *Journal of The Lepidopterists' Society*.

The volume of information presented above only skims the surface, and many important techniques, particularly with regard to dealing with larval diapause, are waiting to be devised or discovered. The combined observations of many amateurs, as we seek to improve our rearing success, will be a major source of progress in this field, if they are recorded and disseminated.

Reviewed and augmented by
Michael M. Collins, Tim L. McCabe
Noel McFarland, Dale F. Schweitzer

For further practical information on rearing Lepidoptera, consult:

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Chapter 7.

COLLECTING ADULT LEPIDOPTERA

Effective collecting of adult Lepidoptera involves knowledge of place, timing, and equipment. Chapter 1, on observation, elaborated on the first two. Habitat requirements dictate the place where a particular species may reasonably be sought. The larval foodplant must generally be present, yet all habitats suitable for the foodplant may not be suitable for the insect. Timing, as to the clock as well as to the season, is likewise important. Onset of the growing (and flying) season can vary greatly from year to year with temperature, especially in mountainous regions, and with the presence or absence of rainfall in arid regions. The stage of development that you seek may be available only at limited times in each year. Field guides and other volumes for the identification of Lepidoptera usually give copious information on these points (see book lists in Appendix J). Lastly, you must have appropriate equipment, and know how to use it, whether collecting a species in hand or on film.

Minimum needs

- *A "starter" net*
- *Small envelopes and pill vials for bringing specimens home alive*
- *A killing jar charged with ethyl acetate (nail polish remover)*
- *Postage stamp forceps for handling specimens*
- *Home made paper triangle envelopes for storing specimens*
- *Pest-proof plastic temporary storage boxes, from your kitchen*

Minimum needs for Chapters 7 and 8 can be obtained as a "beginner kit" from BioQuip (see Dealers and Suppliers in Appendix L).

There was a time when collecting was something that everyone interested in Lepidoptera did almost automatically, often simply to have a collection to be enjoyed and admired. In some hands collecting knew no bounds, even when the numbers of specimens taken exceeded the collector's ability to care for them properly. In today's social climate, the pendulum is swinging towards a "no collecting" philosophy, ignoring the many reasons for collecting that have continuing or increasing validity—helping children become familiar with insects; determining the present fauna of an area, as compared with the past, and defining its ecological and conservation needs; continuing to fill the many blanks in relation to our knowledge of life histories and taxonomy. Collecting in a restrained manner, and for definable and well thought-

out purposes, is clearly desirable. With the privilege of collecting, however, comes the responsibility for proper long-term care and eventual transfer of the collected specimens to an appropriate repository where they will be accessible for study by the scientific community (see Chapter 8, Preparing Specimens; Chapter 12, Guidelines for Collecting; and Chapter 13, Disposition of Collections). If you are not inclined to follow through on these responsibilities, your collecting should probably not go beyond the photographic level.

The need for collecting permits is being extended to more and more situations. National Parks and National Wildlife Refuges have been off limits for a long time, as well as some State Parks, while in National Forests and State Forests no restraints were imposed. It is now best to assume that there may be restraints on collecting on all public lands (and all Native American Reservations) and to make prior inquiry. In some situations this may mean merely presenting yourself to the local manager at the time of your visit, for approval—and it should be obtained in writing. At the other extreme, it may be necessary to make formal application several weeks or months in advance, giving in detail the purpose of your activity. Approval may be contingent on your agreement to submit a report of your findings within a specified number of months, and to deposit all specimens in a university or similar collection. To provide here a list of contact persons for permits is out of the question. It is necessary to inquire locally as to the identity of the warden, ranger, or other authority in charge of the area you wish to investigate.

Prudent lepidopterists will obtain written permission for whenever and wherever they collect, private or public land alike. This has been increasingly emphasized by the United States Fish and Wildlife Service (USFWS). Any trespass on private land, even for "mere" butterfly watching or photography, calls for prior permission. The issue may not be primarily what you are doing, but whether you have a right to be where you are doing it. All permits should be filed permanently as part of your collection. The stress and expense of prosecution for ignoring these "details" can take quite a toll.

One of the outstanding characteristics of lepidopterists is our proclivity for making our own equipment, frequently out of odds and ends available about the house. This chapter and the next, as well as the preceding one on rearing, give considerable attention to home-

made devices. The resulting saving of money and the exercise of ingenuity add greatly to the attraction of the avocation.

1. The Butterfly Net

When one thinks of butterfly collecting, the first thing to come to mind is the butterfly net. Two styles used in the 18th century have been largely consigned to museums—the "batfolder," a two-handled device with a loose fold of netting between, and the "scissors net," a pivoted device bearing two small net bags which could be clapped together with one hand. The third style, the "racquet net" is the one we use today, in modified form (Allan 1937).

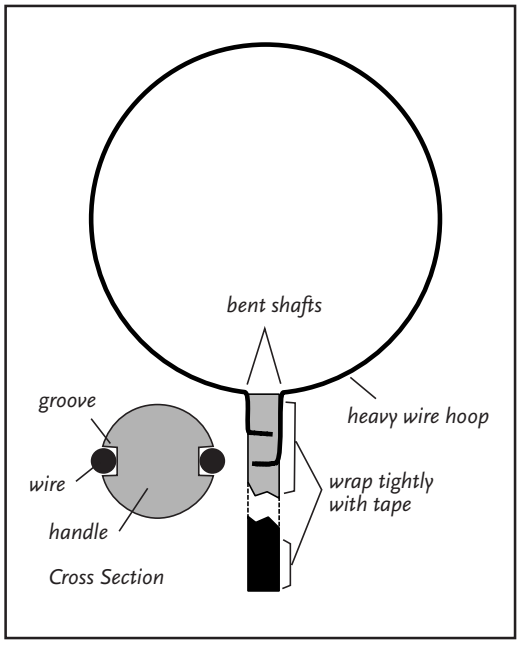

Figure 7–1. Net frame construction.

Currently used nets consist of a hoop 30, 38, or 46 cm (12, 15, or 18") in diameter. The hoop is fixed, in a removable fashion, onto the end of a 2 cm ($3/4$") diameter handle of hardwood or metal about 1 m (3') long. While one of the heavier grades of coat-hanger wire may be satisfactory the first time you make a net, more durable frames are made from heavier wire. No. 9 US wire gauge clothesline wire (hardware stores) is strong and stiff enough and is very satisfactory.

One method of fixing the hoop to the handle is illustrated in Figure 7–1. After forming the wire into a circle, bend 5 cm (2") of one end of the wire, and 6 cm (2 $1/2$") of the other end outwards 90° from the hoop. Then bend the last 1 cm ($1/2$") of each end 90° so that the two bent ends point toward each other. Drill two parallel holes slightly larger than the diameter of the hoop wire through the handle 4 and 5 cm ($1 1/2$ and 2") from the top. Make a shallow groove in each side of the handle from holes to top. The bent tips of the hoop wire are then seated into holes on opposite sides of the handle,

with the shafts lying in the grooves. A tight wrapping with friction tape attaches the hoop securely to the handle. Talcum powder, or dust from the ground, will remove the stickiness from the tape. A piece of wire 132 cm (52") long makes a hoop about 38 cm (15") in diameter, which is a commonly used size. Some collectors prefer larger or smaller nets, for particular purposes. Securing the hoop to the handle with tape simplifies removing the handle for travelling or for putting on a new bag (fresh tape must be used each time the handle is reattached). The handle can be cut in two, and the ends joined with an aluminum ferrule, if greater collapsibility is desired. Wrapping the bottom end of the handle with a few rounds of friction tape will give an improved hand grip.

The bag (Figure 7–2) is made of netting that is strong but not harsh to the touch. Nylon is good and should be of a quality that is resistant to ultraviolet light and of a weave that will retain a stable mesh size, such as marquisette mosquito netting. Cheesecloth is flimsy, snags easily, and is virtually useless. The bag is made to fit the circumference of the hoop at the top; length should be 2.5–3x the diameter of the hoop, and the bottom tapered to a broadly rounded parabolic shape. Avoid a pointed, conical bottom: this damages larger specimens, which become jammed into the apex. The edges of the seam are folded under and sewn a second time to eliminate unraveling edges in which an insect's legs can become entangled. A 12 cm ($4^1/_2$") strip of heavy muslin or light canvas, folded lengthwise and stitched to the top of the bag, makes a tube through which the hoop wire is passed. Dying the bag green or brown reduces the "startle effect," as compared to white.

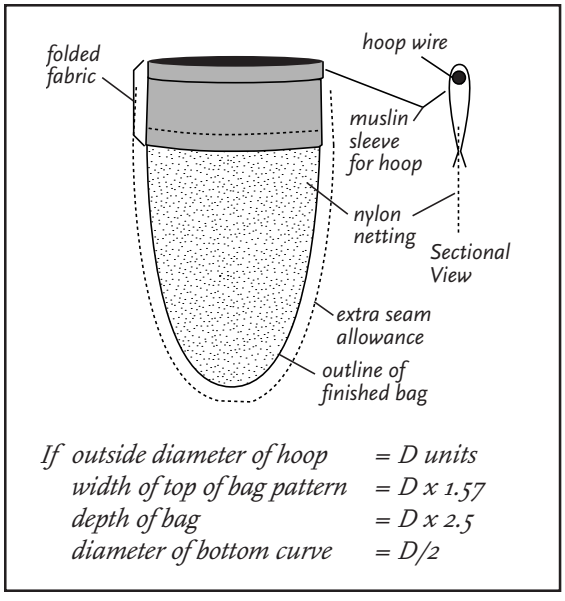

Figure 7–2. Net bag pattern.

You can quickly make a "starter" net using a discarded

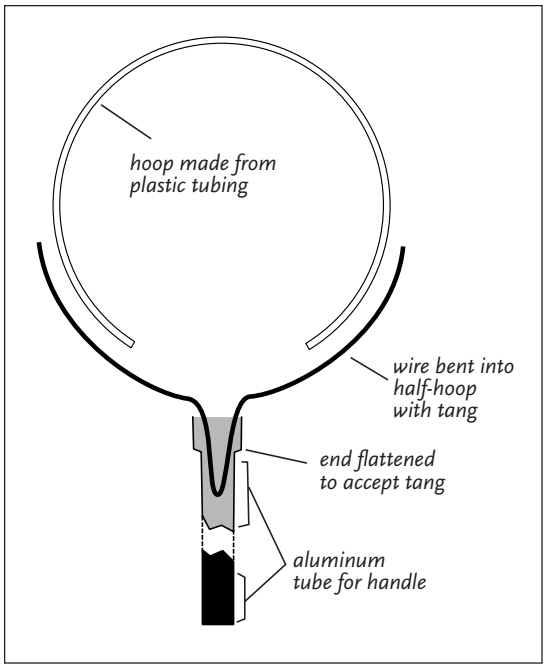

Figure 7–3. Holliday's collapsible net.

fiberglass or bamboo fishing rod for the handle and a heavy duty coat-hanger wire for the net hoop—just reshape the hanger into a circle, narrow the hook part and tape it onto the handle.

Holliday (1989) described a homemade collapsible net he found easy to transport in a backpack or suitcase while travelling (Figure 7–3). He flattened one end of a 1 m (3') length of 16 mm (5/8") diameter aluminum tubing into a narrow oval. A 90 cm (3') piece of 3 mm (1/8") diameter springy steel wire (from a discarded lampshade frame) was bent into the shape of a slingshot, with each arm making a quarter of a circle; the tang was thrust into the flattened end of the handle and adjusted, with the help of Vise-grip pliers, so that the fit was snug but removable. The circle of the net hoop was completed with a length of semirigid plastic tubing of a size to fit easily onto the wire prongs. The net bag (a variant of that in Figure 7–2), attached to the plastic tubing, was made with a 28 cm (11") cleft at the seam side, so that tubing and net could be slipped off the prongs. The cleft could be held closed with a bit of velcro, or with a few burrs or sticktights picked up in the field. The net was easily taken apart for packing, and the handle could be shortened by cutting it in two and joining the halves with a wooden dowel. The security of the tang could be increased by pushing a small twig into the end of the handle.

If you can acquire a telescoping tubular aluminum tripod leg, the small end can be adapted to provide a handle for either of the two nets described above. In the first design, the hoop is attached with a fresh wrapping of friction tape each time it is set up. A handle of this sort, used by the author for 25 years, extends from 0.5 to 1.25 m (20 to 50")

and travels easily in a suitcase.

In tropical situations, where butterflies of many species seldom come down within reach of a conventional net, extra-long net handles are an asset. Extension handle sections that snap together securely are available commercially in 30 cm and 60 cm (1' and 2') lengths. These are designed for use with a net hoop that can be twisted and folded into a compact package one-third its working diameter, making it easy to carry while traveling. Such equipment is equally useful for temperate-zone lycaenids and others that perch high on branch-tips. Replacement net bags are also marketed. For domestic use, try the aluminum pole, extensible from 2 to 4 m (6 $^1/_2$ to 13'), that is designed for painting stairwell ceilings. A conventional net can easily be clamped or taped to the end, and the pole can be transported on a cartop carrier.

The use of the net will be described after other equipment has been considered.

2. Related Equipment

A collecting-bag, or some equivalent, is almost a necessity for carrying the various bits of equipment that are needed in the field. A bag with a shoulder strap, external pockets, and internal compartments is very useful. An old camera gadget bag serves this purpose. It can be set down quickly, if the occasion demands. You can custom-equip an army service belt with assorted pouches to meet your needs. Velcro closures can be added if not already present. Photographers' jackets or vests, with multiple pockets, provide another convenient way to carry numerous small items. Regardless of the design, the carrier should be planned and used in such a way as to minimize jostling of the contents while in the field, and so that both your hands are free for handling net or camera.

What goes into the bag depends upon the particular bent of the lepidopterist. The collector eventually learns to strike a balance between what is indispensable, what would be handy to have along, and what constitutes "clutter" and impairs one's mobility—always an individual decision, tempered by experience. Having the bag always packed and ready to go saves valuable time when an opportunity to go into the field presents itself.

Contents of the Bag

Envelopes, for carrying individual collected Lepidoptera, come in two sorts: glassine "stamp envelopes" (contents semivisible), and paper "coin envelopes" (contents invisible). The transparent mylar dragonfly envelopes, which retain moisture too well and foster mold growth, are not appropriate for Lepidoptera. Glassine retards moisture diffusion considerably, requiring extra attention to drying in humid climates, and use of chlorocresol as a mold inhibitor (see Chapter 8) if the insects are not to be spread the same day. Paper allows more rapid and complete drying. Both can be labeled with ink or ball-point pen. End-opening and side-opening styles are made, and flaps should not be gummed. Convenient sizes are 4.5 x 7.3 cm (1 3/4 x 2 7/8"), 5.9 x 9.2 cm (2 5/16 x 3 5/8"), 9 cm (3 1/2") square, and 8 x 13 cm (3 1/8 x 5 1/16"), and other sizes are made. Oversize specimens can be accommodated in homemade paper triangles (see "Storing Specimens," Chapter 8). Glassine envelopes are most commonly used.

Glassine envelopes are available from Worcester Glassine Envelope Company (Appendix L) and both glassine and paper from biological supply houses and some hobby shops.

Snap-cap plastic vials or tubes are very useful for carrying home larvae (one species per vial) and ova, and for transporting small butterflies and skippers alive (one individual per vial). A butterfly in a vial in a dark pocket settles down and travels safely, without damage from jostling. It remains in good condition for ovipositing, or, if a specimen is the goal, the vial can be placed in the freezer for a few hours before spreading. This is a particularly useful technique for skippers, since muscle stiffening is avoided. Allow time for complete thawing. (Caution: I have occasionally dropped a vial containing a frozen lycaenid and had the butterfly shatter like glass.) Two useful vial sizes are 22 x 52 mm (5 dram) and 30 x 72 mm (13 dram). Clear colorless vials give a better view of the contents than do amber transparent ones, but the latter are perfectly serviceable. Buy the snap-cap type, rather than the child-proof closures. These are available through druggists (possibly with a little help from your physician) in cartons of 200 at rather low cost, and from biological suppliers. Clear plastic film cannisters, often obtainable free from photo labs, are equally useful.

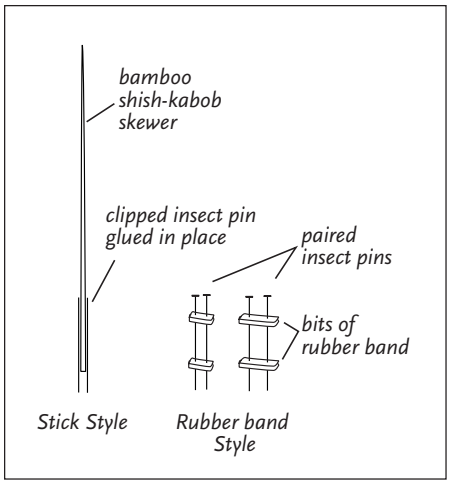

Figure 7–4. Pickle forks.

When collecting "micros" you will need a supply of very small vials for transporting your captures alive. These are described fully in the article by Landry and Landry (1994) reprinted in Appendix F.

Some collected insects killed in the field die with the wings folded downward, rather than up over the back. To avoid scale loss in transit or storage, and to simplify spreading later, you will need to reverse the wings before putting the capture into an envelope. This is quite simple if done within the first 15–30 minutes, before the thoracic muscles stiffen. Insect pinning forceps, with rounded gooseneck tips, or flat-bladed stamp forceps (be sure to remove any burrs from the edges of these with emery paper), are used to allow fingerless handling of the specimen (Figure 8–2). Some collectors develop the knack of stabilizing the insect with a single pin through the thorax, but rotation on the pin is a problem. Instead, the lepidopterist's pickle fork, a homemade device (Figures 7–4 and 8–3), can stabilize it reliably while the wing position is being reversed. Use the forceps to grasp the under-half of the thorax beneath the wings, and thrust the two tines of the fork into the midline of the top of the thorax. Then move the forceps blades carefully upward to flip the wings up over the back and keep them there, maintaining a hold on the insect by the sides of the thorax and base of the wings, not by the wings alone. Remove the fork and use the forceps to insert the specimen into an envelope.

There are two types of forks, each easily made (Figure 7–4). The first uses two insect pins, No. 1 or thinner, and two pieces of flat rubber band, less than 2 mm ($1/16$") wide and 5 mm ($3/16$") long. The pins are thrust through the two pieces of rubber, with the points 1.5–3 mm ($1/16$–$1/8$") apart, depending on whether you are making a fork for small or more robust specimens. The upper and the lower pieces of rubber are pushed 2 and 0.5 cm ($3/4$ and $3/16$") up from the points of the pins, respectively, to hold the pins parallel and perpen-

dicular to the rubber supports (Winter 1980).

To make the other type, clip the heads from two triple-zero insect pins and glue them to opposite sides of a bamboo shish kebab skewer, with the points extending about 0.5 cm ($3/16$") beyond the end of the stick. This can be cut to a length of 4 cm ($1\ 1/2$") or longer, as convenient. Either type can be carried easily in the field in a snap-cap vial with a little padding in the bottom to protect the points; each can be cleaned after each session of use by thrusting the pins through a few layers of tightly woven cloth.

Carry your forceps on a necklace cord, to prevent loss, and tuck them in your shirt pocket when not in use. Drill a small hole through the top end of the forceps, and string through it a loop of braided nylon cord (this tangles less than twisted cord). Tie the cord to a length such that when it is around your neck you can work easily with the forceps at lap level. If you spray-paint the forceps red or orange, they will be easier to locate if dropped.

Boxes, particularly the metal tins used for various drugstore and confectionery products, are useful for carrying stocks of empty envelopes, enveloped specimens, and for larvae and pupae too large for vials. Sealable plastic bags are helpful for carrying colonies of larvae or extra foodplant.

Plastic sandwich boxes with layers of paper towel or toilet tissue make excellent storage containers for specimens, especially on extended trips. Place a wad of damp tissue in the box to keep the specimens relaxed until you have time to spread them. Add a few crystals of chlorocresol or paradichlorobenzene (PDB) to prevent mold growth, tape the edges, and store in a cooler, refrigerator or freezer.

A needle and thread and Band-aids can be used for repairs to net or collector, respectively. Insect repellant can make a day in the field more pleasant and also reduce the personal risk from disease-carrying arthropods.

An aspirator is occasionally used for capturing very small microlepidoptera. The description and use of this specialized implement will not be considered here.

Killing Jars

These are part of the basic equipment of the collector. The jars are

generally glass with tightly fitting screw-on metal lids and should be as wide-mouthed and deep as possible. Plastic lids and plastic jars are fine if you determine that the plastic is not degraded by the killing agent you use. The sealing gasket inside a metal lid must also be unaffected.

Before choosing to use any of these agents, you are urged to read about chemical hazards in Chapter 10, Section 8.

Ethyl acetate is a commonly used killing agent that kills quickly and has little tendency to stiffen specimens, especially during the first hour. It is preferred by increasing numbers of collectors. To make a jar for ethyl acetate, pour a 2 cm ($3/4$") layer of thick plaster of Paris onto the bottom of the jar and allow it to harden. When it is totally dry (no sweating on the inside after the cover is put on) charge it by adding enough ethyl acetate to saturate the plaster. Pour off any excess. When the jar becomes weak, as evidenced by slow knockout time, add more ethyl acetate. This agent is available from chemical and entomological suppliers (Appendix L). Clear nail polish remover with 80% ethyl acetate is sold in drugstores in most countries. Acetone-based polish remover is not suitable.

If a charged jar begins to sweat inside (common on hot days), open it long enough to dry it out. Wings touching the moist surface will immediately lose their scales.

Ammonia makes an effective killing jar, prepared with a pad of cellulose wadding or blotting paper in the bottom (do not use absorbent cotton—it snags legs badly). A few drops to 1 ml ($1/4$ tsp.) of liquid ammonia will charge the jar. A strong solution of ammonia is necessary—30% ammonium hydroxide in water. "Household ammonia" is a very weak solution and quite ineffectual. Advantages of this agent are that the killed specimens remain relaxed, and that it is harmless, though irritating to human eyes and noses. Specimens are quieted down less quickly than with ethyl acetate, and larger ones may require an hour for sure killing. Disadvantages are that greens easily turn brown, and some reds and browns may be dulled, unless the specimens are transferred to an empty jar within a few minutes after dying. High volatility results in pressure buildup in the jar in hot weather (Dickson 1992), and in humid situations the inside of the jar may sweat and need to be wiped dry. Ammonia is seeing increasing use, and its lack of stiffening is particularly advantageous for

"micros" (see Section 8–2). Ammonia solutions must never be stored in the sun or a warm place—excessive pressure will develop within the container, creating a potential hazard (in the form of escaping ammonia gas) on opening (Sokoloff 1980).

Ammonia from ammonium carbonate (see Keystone Universal Corp., Appendix L) has long been used in England and is being adopted widely in the U.S. Put a 2 cm (3/4") layer of the crystals in the bottom of a thoroughly dry plastic peanut butter jar and hold it in place with a slightly oversize disk of dry synthetic sponge pressed down firmly (Gilligan & Gilligan 1990). Ambient humidity releases enough ammonia fumes to charge the jar. An alternate method is to cover the crystals with 2 cm (3/4") of dry sawdust held in place with a couple of cardboard disks. To make self retaining disks, measure the interior diameter of the jar (using homemade cardboard calipers, Figure 7–5), draw two circles of that diameter on a piece of cardboard from the back of a pad of paper, and cut out the circles with pinking shears. Be sure that the inner teeth of the shears notch the drawn circle just a little. Make one hole with a paper punch near the edge of each disk (to simplify later removal of the cardboard when you need to recharge the jar). Insert the disks into the jar one at a time and tamp the edges down with the top end of a pencil; be sure the holes are not lined up together.

Chloroform and carbon tetrachloride are no longer used as killing agents because of their cumulative toxicity to humans (by inhalation of escaping fumes) and because of excessive stiffening of the specimens.

Cyanide, as the potassium, sodium, or calcium salt, was the preferred killing agent in the past. Because of its extreme and rapid lethal toxicity to humans, it is no longer marketed to the general public. Its use cannot be recommended.

If you nevertheless choose to use this agent, the jar should be prepared in the least hazardous manner. Select a glass or rigid plastic jar with a screw-on metal cover and an intact gasket. Working in a well-ventilated area and away from children, spread a 0.5 cm (1/4") layer of cyanide crystals on the bottom, then a 1.5 cm (1/2") layer of dry sawdust, vermiculite or dry plaster of Paris, then a topping of 1 cm (1/2") or more of thick plaster of Paris (thicker layers for bigger jars). The jar is then set outdoors on a breezy sunny day, uncapped, in an

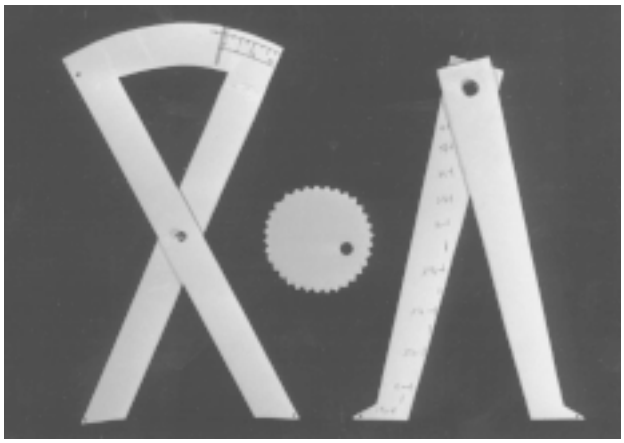

Figure 7-5. Cardboard calipers and disk.

area absolutely inaccessible to children, until the plaster is totally dry. Depending on the ambient humidity, this may take more than one day's exposure. In an incompletely dried jar, the cyanide crystals will soon liquefy, the jar will be dangerously strong, and its life will be greatly reduced. On the other hand, a "weak" jar that still has visible crystals in the bottom can be perked up by the addition of two or three drops of water. The bottom of a cyanide jar should be crossed with radial strips of fiberglass reinforced plastic packaging tape, extending up the sides a bit beyond the level of the plaster. These tapes should then be covered with circumferential bands of tape around the base of the jar. The upper portion of the jar should be encircled with overlapping wide bands of transparent packaging tape. If the jar is accidentally dropped, the cyanide crystals will be safely contained.

A "spent" cyanide jar should be disposed of safely. Lippert (1991) advised wearing protective goggles and filling the jar half full with chlorine bleach (sodium hypochlorite), outdoors. He then broke up the contents with a wooden stick or dowel, stirring occasionally over a period of a few hours. This converted any residual cyanide to the relatively harmless cyanate. This procedure should be carried out in a location inaccessible to children.

Injection (using a hypodermic syringe and needle) of a few drops of ethyl acetate or 70% isopropyl alcohol into the thorax of an insect will cause instant death. Injection of ammonia is equally effective (Cobb 1949). Emmet (in Friedrich 1986) cautioned that injection of ammonia can compromise the green colors of Lepidoptera, and advised injection of ethyl acetate, instead, in such species.

The list of items that would be useful to carry along on an extended collecting trip can be quite lengthy, and it will vary according to the terrain you will be visiting and the emphasis of the trip. Some

will be carried in your collecting bag, others will stay behind in your car or camp. It is very useful to keep in your word processor a complete list of your collecting and photography field equipment, including needed spares. Before a trip, run off a copy, cross off as many nonpertinent items as you can—you won't need waders in the desert—and ready the rest. Two hours down the road you won't start recalling all those items you should have brought along!

3. Using the Net

The net may be handled in a number of ways. An insect may be intercepted in flight as it approaches you, but many butterflies, seeing the approaching net, dodge effectively at the last moment. An upward swing, from beneath the calculated flight path, or a swing overtaking the insect from behind, is more often effective. Any of these maneuvers is best undertaken with both feet on the ground. The classical headlong dash across the field nets very few butterflies, and more often leads the sprinter into injurious or ignominious encounters with chuckholes, hornets' nests or obscure watercourses. A patrolling butterfly can often be captured if you stand and wait for it to make its next circuit. Keep in mind that too rapid or vicious a swing can result in severe wing damage to the butterfly or moth. Wielding a net in each hand is hardly to be recommended—it bespeaks more of greed than of competence.

Once the butterfly or moth is in the net, the handle must be quickly rotated 180° to fold the net bag shut and prevent escape. If at this point the butterfly ends up quite near the rim of the net, a quick sweep through the air with the net open will usually move it to the bottom of the net (or give it just enough time to escape!). The handle is again rotated 180°. Holding the end of the closed net bag up can also encourage the butterfly to move to the narrow end of the bag. Once the captive is in the end of the bag, the netting can be held shut above it, making close examination possible. An unwanted or bedraggled specimen can easily be released unharmed.

Insects are more readily captured when they are settled—nectaring, perching, basking, or simply at rest. If your prey is on a flower as tall as or taller than the length of your net bag, grasp the bottom of the bag and hold it out horizontally with one hand, releasing it just after you start a brisk horizontal backhand swing with the

net, with good follow-through, turning the net over once the butterfly is within. Skill lies in judging the height of the swing to avoid hitting the stem (and spooking the butterfly), decapitating the flower (and being ejected from the garden), or swinging too high and capturing naught but air.

A butterfly perched on top of a flower closer to the ground than the length of the net bag can be captured by clapping the net down on it from above, at the same time keeping the tail of the net elevated with your free hand. The disturbed butterfly will usually fly up into the end of the bag, at which point the bag can be quickly raised, swished, and turned to close it. This same approach can be used to capture a basking or puddling specimen. Some satyrids, and other species in alpine and arctic situations where the air is too cool for sustained flight, may drop to the ground and walk out beneath the rim when an attempt is made to capture them this way. Occasionally such a miscreant can be picked up by its folded wings with forceps, or between your second and third fingers, as it tries to sneak away. And some even climb down among rock rubble, so that it is necessary to engage in a bit of quarrying to recover them.

Butterflies perched on tree-trunks can sometimes be picked off by a sweep of the net, barely touching the trunk with the net rim, but too often projecting branches make this impossible. A better approach is to place the net quickly over the insect with the net bag held out horizontally. The instant the insect flies, the net is swept and closed in the manner described for catching an insect on the wing. A moth resting on a tree trunk in the daytime, however, is often asleep and can be captured by covering it with the mouth of an open jar held against the trunk—a very useful approach for underwings.

Try to protect your net bag from trouble. A wet net will remove many scales from any lepidopteran caught in it, so caution is necessary around wet foliage, stream banks and puddles. Clapping the net down over the insect, while the end of the bag is held aloft, is usually effective. The same approach is recommended for capturing a butterfly feeding on dung or carrion. Some of the best nectaring flowers seem to be located among brambles or along barbed wire fences, so it is well to survey your opportunities for snagging (or ripping) the net before you swing. A torn net can be repaired temporarily and quickly with a patch of duct tape (a couple of 15–25 cm

[6–10"] pieces can be wrapped around your net handle) applied to the hole inside and out (Baggett 1981). A needle and thread and a few small safety pins, carried in the collecting bag, are useful for more satisfactory but less rapid repairs. The best solution is to carry a spare net bag that can be installed in place of the damaged one. Lepidoptera seem to have a "sixth sense" when it comes to finding the hole in the net!

4. Recovery from the Net

If your plan for a captured insect is to retain it alive, as for egg laying, cage mating, or other reason, it can be removed from the net in several ways. Since two hands are required for making the transfer, it is handy to have an envelope ready in advance, holding the flap between your teeth and the net handle under your arm.

If your capture is a butterfly or broad-winged moth that is quiet and not struggling:

1. Carefully maneuver it into a position with the wings folded over the back (and no fold of net in between).
2. Grasp the sides of the thorax gently between thumb and forefinger, from the outside of the net.
3. Reach inside the net with the other hand and grasp the insect by the sides of the thorax, to lift it out of the net.
4. Slip the insect into an envelope and close and crease the flap.

If you have taken a lycaenid, small skipper or small moth, active or quiet, it is safer to remove it using a snap-cap vial. Isolate it in the small end of the net as you would for enveloping, but do not try to arrange its wings or grasp it.

1. Hold the vial cap between your teeth and use the end joints of your 2nd and 3rd fingers as a temporary cap.
2. Maneuver the vial inside the bag and beneath the insect.
3. Uncover the vial so that the insect can fly down into it,
4. Then cover it with your fingers when you see the bug at the bottom.
5. Take the vial out of the bag and slip the cap beneath your fingers and onto the vial. Pay attention to avoid a "rim job"—crushing a wing tip against the lip of the vial!
6. Place the vial in a collecting bag or a dark pocket.

A butterfly will stay quiet and safe through a day-long hike. A

female moth may lay eggs en route.

With a little practice, either of these maneuvers becomes automatic and reliable. The vial approach is ideal for skippers, because it can be performed quickly and reduce scale loss.

A vial should house only one insect at a time. Make exceptions only for copulating pairs.

If you are certain that you wish to retain the insect as a dead specimen for a collection, pinching the thorax at the second step of the envelope sequence, above, is an effective way to immobilize it. Then complete the sequence.

The pinch must be applied precisely to the thorax, with enough pressure to stun but not enough to damage the body. Pinch too far forward and the head may be damaged or detached, or too far back and the abdomen may burst. Properly pinching small lycaenids or active small skippers calls for dexterity beyond the reach of many of us, although Ferris and Brown (1980) recommend pinching the thorax of skippers from above and below, rather than sideways, a procedure requiring extreme care to avoid scale loss. Pinching most moths without causing grievous wing damage is out of the question.

Some collectors carry the enveloped specimens in a crush-proof box in a pocket or collecting bag, but as a pinched insect is not necessarily dead, placing it (in its envelope) in a killing jar, for half an hour at least, seems a better practice.

An underwing moth (and most of its relatives) caught in a net requires quick and careful transfer to avoid unsightly loss of scales, especially those on the top of the thorax.

To use a killing jar, trap the insect in the small end of the bag as before:

1. Unscrew the cover of a rather narrow jar, but hold it in place with thumb and forefinger, while using the other hand, outside the net, to keep the moth trapped in the end of the bag.
2. Maneuver the jar inside the bag and beneath the insect, uncover the jar so that the insect can fly down into it, and replace the cover when you see the bug at the bottom.
3. To keep a specimen in the best possible condition, transfer it, as soon as it is motionless, to a glassine envelope; then handle it as described under "pinching," above.

A tiny moth caught during a day trip is best field-pinned as soon as

it becomes still and put into a small pinning box carried for that purpose (Section 8–6).

If you use a vial-cap with small holes drilled through it, the vial can be placed in a killing jar to knock out the specimen for enveloping.

At times you may inadvertently capture several insects at once. Since transfers to jar or vial must be handled one at a time, this increases the opportunity for scale loss or for escape of the specimen you wanted most. Multiple capture should therefore be avoided, but if your prime target is in the midst of a cluster of nectaring or puddling butterflies, you may have no choice.

Some collectors prefer to remove medium to small sized specimens to a small killing jar. Worn or damaged specimens can be released in a minute or two, after becoming subdued. They revive in a few minutes.

Clearing the net of bees, hornets, and unwanted Lepidoptera can be done easily by holding it open with the bag hanging down. If the insects do not quickly fly up and away, they can be encouraged to do so by flipping the bag inside-out by means of a quick to-and-fro swing of the handle. Any accumulated debris or detritus can be removed in the same fashion. Burrs and thorny twigs and leaves should be untangled one hook at a time. Spikelets of some of the larger grasses may become detached and penetrate the mesh of the net. Since these commonly bear barbs pointing distally, they are best removed by pushing them carefully through the mesh, butt end first. If they are not removed they can cause extensive damage to a captured butterfly and to the net material.

A lepidopteran resting with the wings folded over the back can often be handpicked by grasping the folded wings gently between forefinger and thumb, or between the sides of two adjacent fingers. The insect can then be held more securely by the sides of the thorax, using the thumb and forefinger of the opposite hand. If the butterfly is quite small, you can use straight postage stamp forceps (not the angled type) to pick it up. It is important not to rub the upper surfaces of the wings back and forth against each other while performing these manipulations. I have watched a person adept at locating lycaenids that were resting beneath foliage on cool, drizzly days, who used this approach very effectively.

5. Collecting at Lights

How to locate moths and butterflies for daylight and crepuscular collecting has been covered extensively in Chapter 1 on observation. Night collecting is another story, and much moth collecting centers on use of lights. Bear in mind the importance of having advance permission for collecting on private land. The unexpected presence of flickering lights in field or woodlot leads to investigation; some landowners investigate with firearms, and police are not necessarily patient with rational explanations.

"Existing lights" (lights you did not put up yourself for collecting purposes) are a rich source of material. Porch lights, storefronts, all-night gas stations and convenience stores, walls of buildings next to illuminated parking lots, highway rest stops all attract moths that settle down and rest until daylight. Many people have built extensive and scientifically valuable collections by regularly visiting such sites at night, or before dawn before the birds begin to clean up the area. Birds are avid collectors and recognize the benefits of the lights too! Moths found in such situations are generally very subdued and can be knocked into an open killing jar with a little flick of the cover. Trying to use a net to nudge or sweep a specimen off a wall most often leads to damage or loss. A net should be tried only if the specimen can be reached in no other way. Females to be held for egg-laying can be transferred from the killing jar to an empty container as soon as they stop moving. They will recover. Females usually make up a very small percentage of moths attracted to light (the reverse is true for Mimallonidae). However, Tuttle's observations on saturniids (Tuskes et al. 1996) indicate that portable lights set up close to larval foodplants can take as many (ovipositing) females as males.

Lighted public rest-rooms are excellent moth-traps, and they retain their captives right through the following day. But respect that little icon on the door. If you don't match it, send in a friend of the opposite sex.

Quality of Light

The type of light source is significant. Ordinary incandescent light bulbs attract moderately well, and are not to be disparaged (the special yellow bug-bulbs do not attract). Fluorescent "blacklights" (BL), of the sort that produce considerable visible light, are excellent because of

the ultraviolet (UV) that they emit. They should not be screened with any plastic that excludes ultraviolet, a mistake made by some of the earlier manufacturers of blacklighting equipment. The shields designed to protect the tubes transmitted visible light longer than 400 nanometers (nm), but prevented passage of wavelengths in the 5 nm attractant range (Flashka & Myers 1974). The purple "blacklight blue" (BLB) blacklights (used for fluorescent mineral displays) attract well but are less easy to collect at. Best of all are the mercury vapor (MV) lamps, because of their excellent UV output and greater intensity. The lamps must be carefully selected, however; some are specifically shielded to reduce UV output, and their usefulness is thereby compromised. The desired wavelengths for attracting Lepidoptera and the spectral emission characteristics of various light sources are summarized in Figure 7–6 (Brou 1992b, Sylvania 1994a, b).

Lepidoptera are attracted most by the shorter, ultraviolet, wavelengths, 300–375 nm, less by the visible (to humans) violets and blues (375–490 nm) and least by the yellows to reds (570–750 nm).

BL fluorescent tubes emit mainly at 300–400 nm and peak at 360 nm and also have considerable visible light. Note: "F15" BL tubes, in contrast to the other F-number designations, have a less intense output (about 40%) over a slightly broader range, and peak at 375 nm. BL tubes in the portable units from entomological suppliers are almost always "F15," whereas tubes purchased from electrical suppliers, to use in homemade devices employing household straight or circular fixtures, are in the other F-classes and peak at 360 nm. I know of no comparison tests of field efficiency and, as a practical matter, both sorts work very well.

BLB fluorescent tubes are made with special filter glass transmitting the 360 nm emissions and only a small amount of deep violet visible light. They are more expensive and much less satisfactory than BL tubes, but they are sometimes an advantage in field situations where visible light might be objectionable, as near campgrounds.

Clear MV lamps emit high output at about 360 nm, as well as visible light down to but not including orange and red. Output shorter than 350 nm (most of the sunburn range) is occluded by the characteristics of the outer borosilicate glass bulb. If the outer bulb is broken, dangerous amounts of UVC, (below 300 nm) are emitted.

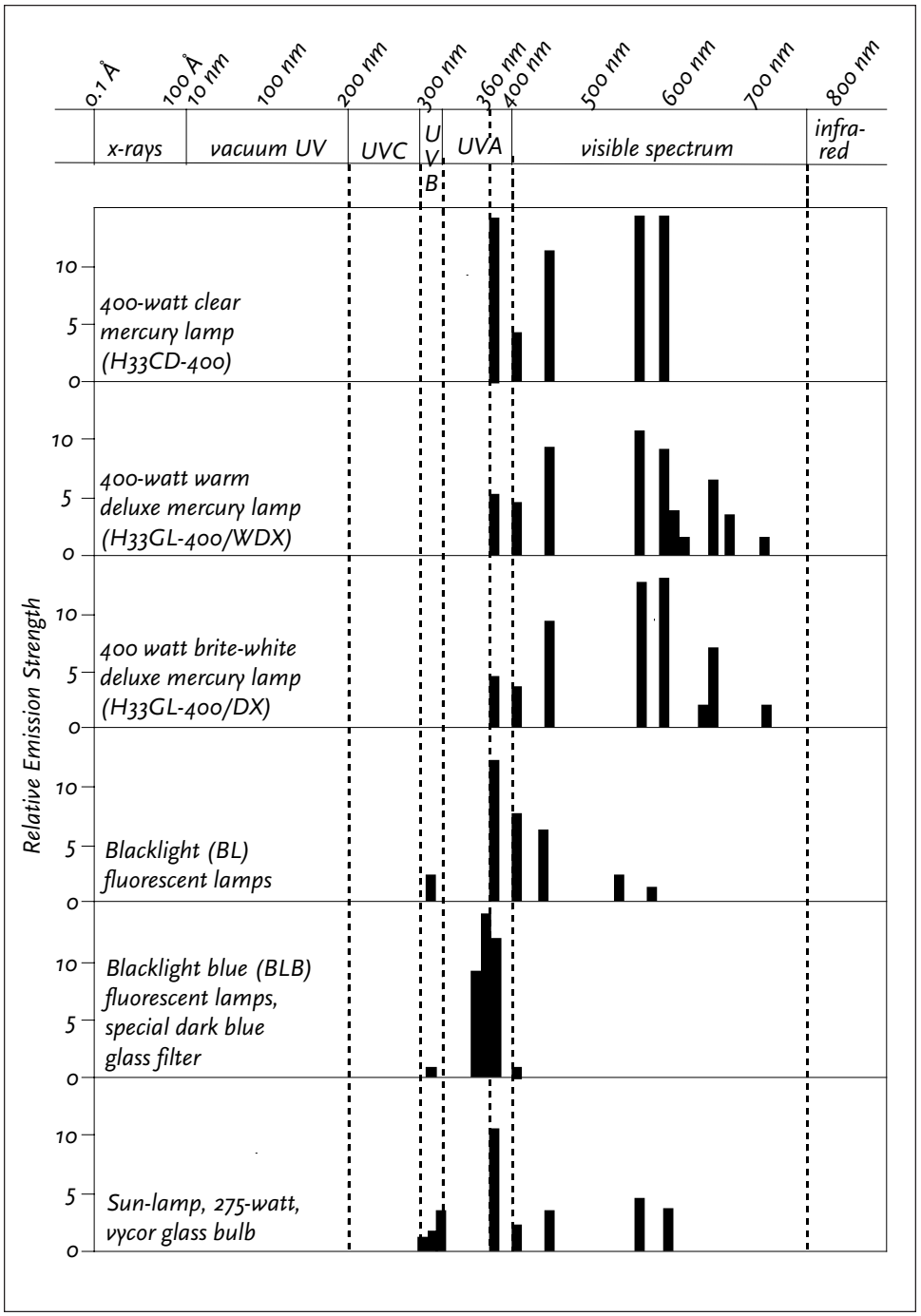

Figure 7–6. Emission spectra of various light sources.

Each type of MV lamp requires a specifically designed ballast. "Self-ballasted" lamps have considerably lower efficiency and shorter lives than those with an external ballast.

Coated "white" and "warm white" MV lamps (coded "DX") have the intensity of the 360 nm output reduced 60–70% by an internal phosphor in the outer bulb in favor of increased emissions at the red end of the scale. These are less effective, and are uneconomical in power consumption. Lamps classified "clear" are to be preferred. While the light output of fluorescents diminishes at lower ambient temperatures, this is not of major significance at the temperatures at which moths are on the wing. MV lamps are not so affected.

"Sunlamps," designed for tanning, produce radiation in the 290–350 nm (sunburn) range, and much stronger output at 365 nm. Special glass allows passage of UVC (below 350 nm). These lights are effective for collecting, but are hazardous for the collector.

Proper eye protection should invariably be worn when working around UV light sources, for protection not only from radiation but also from flying shards if a bulb explodes spontaneously (for more details see Hazards, Chapter 10).

The slightly peach-colored sodium vapor lamps that are being used increasingly for commercial all-night light because of their lesser expense are very poor attractors, and some of the above-mentioned "existing" situations are becoming less productive. This is probably a good thing, as there had been real concern that the MV lights were seducing too many males (moths, that is) away from their reproductive responsibilities.

Before invention of the electric light, oil lamps and candles attracted many moths and were the mainstay of night collecting. The potential use of kerosene, propane, or Coleman gasoline lanterns should not be overlooked, if electricity is unavailable.

Power Sources

Availability of electric power is a consideration in the choice of a light source. With the advent of screw-in fluorescent units that can handle a BL circular fluorescent tube, it becomes convenient to use a BL as an ordinary porch light, or for any setup powered by 120VAC house current. But BL is also highly portable. Units can be purchased powered by 12VDC sources, such as an automobile cigarette

lighter socket, or any 12V battery. These include battery packs designed for these units, or small car batteries, marine batteries, etc., or a portable 50W plug-in inverter can run a 120VAC unit from a 12VDC automobile battery via the dashboard outlet.

The MV lamps require a 120VAC power source, either house current or a portable generator. Honda or Homelite portables with outputs of 400–650W as 12VDC or 120VAC have proven very serviceable. If the current is interrupted, as from a brief generator stall, there can be a 4–6 minute delay until the MV lamp cools enough to be restarted, but the superior attractancy of the MV easily outweighs such inconveniences.

All things considered, 12VDC BL is the most commonly used for field collecting.

Setups

These consist of sheets and traps. The former require at least part-time attendance, while the latter are left unattended overnight, or in some configurations, over several nights.

The sheet is simply a white bed-sheet of any size that proves convenient, and it can be rigged vertically or horizontally.

A horizontal sheet is the easiest to set up or to move to a new location. Lay the sheet on the hood of your vehicle, and let it hang to the ground in front. A few anchoring heavy-duty cupboard door magnets will hold the sheet in place if it is breezy. Plug your 12V BL unit into the dashboard outlet and lay it near the front of the hood, or suspend it over the hood using the support described below for a vertical vehicle sheet. A Coleman lantern set on the ground in front of the vehicle encourages moths to settle on the dependent portion of the sheet. Because this arrangement takes so little time to set up or to dismantle, it makes it easy to move to a more promising spot in the event of poor collecting, or to sample different altitudes in the mountains. The number of moths flying through your headlights as you drive can give a clue to a good collecting area.

In open or grassy places where there are no vertical supports available, the sheet can be spread flat on the ground and the light supported above the center. The MV lamp is well suited to this approach. Its base, as provided by entomological suppliers for the U.S., is threaded to take a tripod screw ($^1/_4$" 20-thread National

Coarse) so it can be mounted on a camera tripod or bolted onto a stable base placed on the ground (such as a square piece of plywood or an inverted metal pie plate). Adapters are available for interconversion for tripods designed for metric-threaded cameras. For this type of collecting, *eye protection is absolutely essential.*

A sheet can be held vertically against a wall with tacks or pushpins, or held against the side of a vehicle with magnets. A BL fluorescent tube is suspended 25 to 30 cm (10–12") out from the upper center of the sheet, from a stick or other support. If the tube is too close to the sheet, the sheet will be illuminated mainly in the center and attracted moths will settle down less well. If you collect regularly at a vehicle, it is handy to cut a square stick of wood about as long as can be stored on the shelf beneath the rear window. Attach magnets to one end and to the center, and make a notch at the other end from which to hang the BL tube. This stick, mounted on the middle of the roof, and projecting beyond the sheet, supports the light in the desired position. The same support works if you prefer to spread the sheet on the hood of your vehicle.

If you have a portable power source and are working where there are trees, then the sheet can be supported from a sash cord or clothesline strung between two trees (several feet farther apart than the width of your sheet) where there is enough open space for you to work at both front and back of the sheet. The line should be rigged so that it is taut and horizontal, with no sagging.

Figure 7–7 shows the arrangement of the suspending cord. Portion "A" is the beginning of the line, portion "B" is the suspending portion, and portion "C" is the excess line. Some excess is desirable, so that you can make use of larger or more widely spaced trees when necessary.

To hang the sheet, fold a small margin over the line and hold it in place with spring clothespins. The bottom end of the sheet should be spread flat on the ground. A few paper egg boxes can be placed on the bottom to give shelter under which buzzy moths will quiet down.

If the air is at all breezy, the bottom fold of the sheet should be weighted with a few stones to keep the sheet from flapping. A windy night is a poor choice for collecting at a vertical sheet.

The blacklight tube can be hung in front of the sheet from an overhanging branch, if available, or a pair of lights can be suspended

on opposite sides of the sheet. Make a hanger from coat-hanger wire (Figure 7–7). Join the ends with a loose "bowstring" of medium-weight string, length 1 1/2 times the span of the hanger. Place the hanger on the line at the center of the sheet, and stabilize it with one spring clothespin immediately to its left, and another immediately to the right of the fully extended "bowstring." Then hang a blacklight on each end of the hanger.

Because the best collecting is often on a night with rain threatening, it is good to rig your line so that it can be taken down quickly and easily even if it is wet.

"Slip-rigging" is an easy way to set up a readily removable line (Figure 7–8; "A," "B" and "C" mean the same as they do in Figure 7–7.):

1. Pass two turns of the line around the first tree and hold on to an arm's length of loose end "A" to use for step 3. The extra turn helps to keep the line in position around the tree until you set the knot.
2. Form a slip knot in line "B" near the tree. Be sure the running side of the slip knot points toward the second tree.
3. Make a short loop in line "A," pass it up through the slip knot,

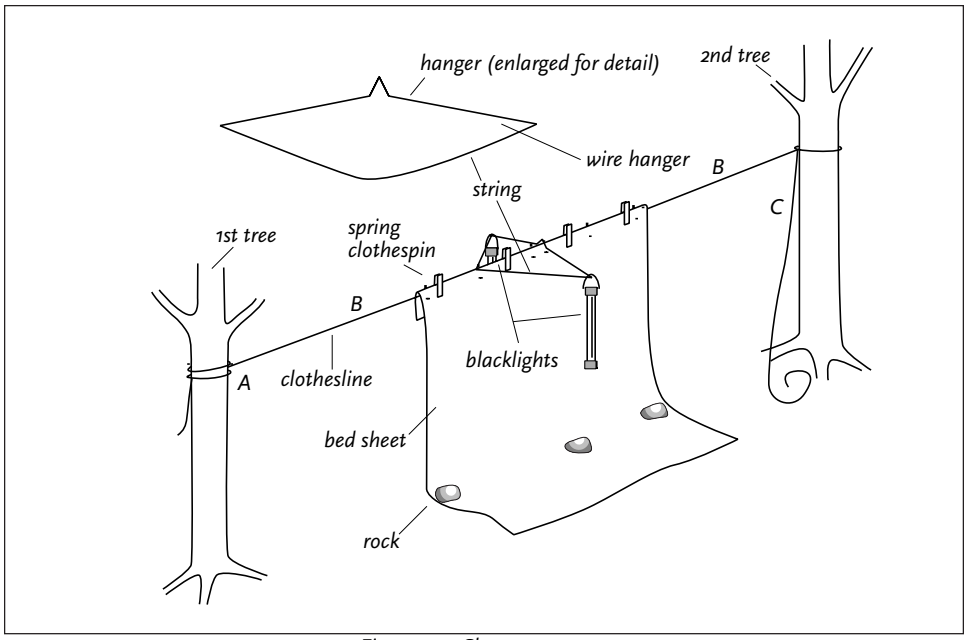

Figure 7–7. Sheet setup.

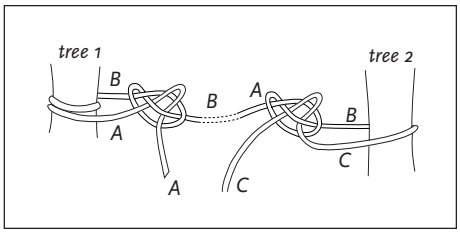

Figure 7–8. Slip-rigging details.

and pull the knot as tight as possible, to "strangle" the loop.

4. Near the second tree, form another slip knot, again with the running side pointing towards the second tree.
5. Pass the line once around the tree, form a longer loop from surplus line "C" to pass through the slip knot, and pull the knot tight. Tighten the line until it is taut by pulling on the arm of the loop that comes from the tree.
6. To take down, pull hard on the free end of the line at each tree to pull the loops out of the slip knots.

If you are not into knots, "block-rigging" may work better for you (Figure 7–9; the designations "A," "B," and "C" are comparable to the same terms in Figure 7–7). Start off by making and permanently assembling the tension block and hooks, as follows:

1. Cut a piece of broomstick about 10 cm (4") long. Near one end drill a transverse hole just a little larger than the line you are using. Near the other end drill a pilot hole, parallel to the first hole, and screw a 1.25 cm ($^1/_2$") screw-hook into it.
2. Run the short end of the line ("A") through the hole, being sure that it comes out on the side that has the screw-hook.
3. Close one side of a 1.25 cm ($^1/_2$") S-hook, and open the other side wide enough so that it can be hung onto your line. Tie the short end of the line permanently to the closed side. (This is a onetime procedure.)

To set up the line,

1. Pass the S-hook twice around the first tree and hook it over the long side of the line "B." Work out any slack where the line encircles the tree.
2. Move the block along the long side of the line "B" almost to the second tree and pass the line "C" once around the tree.
3. In this loose part of the line, form a slip knot, in a position where it will just reach the horizontal part of the line near the tree. Be sure the running side of the slip knot is the line you have brought around the tree, and not the surplus line "C" beyond the knot.
4. Hang the slip knot on the screw-hook and tighten the knot

snugly.

5. Now slide the tension block along the horizontal line toward the first tree until the line is taut, as in tightening a tent-stay.
6. To take down, loosen by sliding the block toward the second tree; slide the slip knot off the screw-hook; then lift the S-hook off "B"at the first tree.

A net is little needed in collecting at a sheet, except for capturing and disposing of large "nuisance" insects that repeatedly crash into the sheet and disturb smaller visitors, or for moths that refuse to settle on a horizontal sheet. Vigorous moths taken in a net quickly lose the scales from the back of the thorax. Most moths are most easily captured by nudging them off the sheet with the edge of the lid into a partly opened killing jar. With practice this can become a one-hand operation, holding the lid ajar between your thumb and index fingers. Be cautious to avoid a "rim job"—getting the specimen caught between the cover and the rim of the jar. Have several small to medium jars available to use in rotation, transferring the catch periodically to a large "dump jar" so freshly killed moths will not be damaged by active new captures.

If you use only one light, also check the back of the sheet. Many moths roost there, and also beneath the fold at the top of the sheet.

Never put beetles in the same jar with moths. They lumber about, die slowly, and damage moths severely.

Covell (1972) recommended putting a layer of felt over the plaster in the "dump" killing jar to reduce jostling of the dead moths in transit after collecting. This is helpful, but because many tarsal claws become tangled in the felt, the moths have to

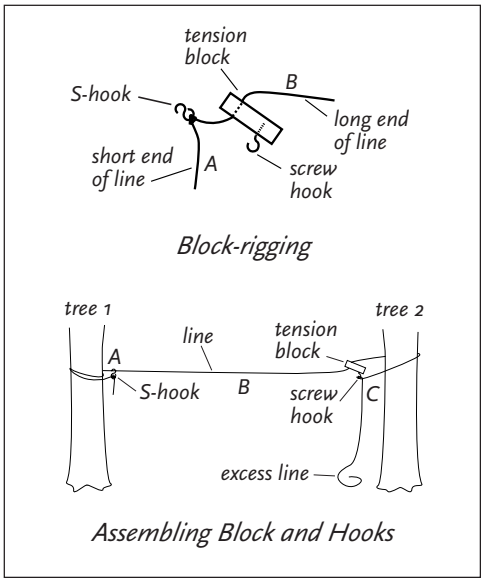

Figure 7–9. Block-rigging details.

be removed from the jar with care.

If you are collecting at a sheet on the ground, coaxing a moth into the killing jar is a lot trickier, since it cannot fall in. This method of collecting can also be hard on your knees and back, but it works. Sponge rubber knee pads help. Another approach is to suspend the light source from a tripod set on the ground sheet; pile empty egg cartons on the sheet, and before bedtime select desired moths from their resting places beneath the cartons, knock the rest off, shut off the light and retire. This is a very simple approach for collecting at home (Dickson 1992).

McFarland (1966) described a highly efficient and semipermanent sheet setup. The sheet was roughly 5 x 2 m (16 x 7') and was made not of pristine white bed-sheeting, but of "off white" canvas or heavy muslin. It was supported vertically in a parabolic configuration by 13 poles set in the ground. Figure 7–10 shows 30 cm (1') grid squares, but you can use a larger or smaller grid. Lay the curve out carefully and locate the light-source at the focal point of the parabola. McFarland used two 45 cm (18") 15 W blacklight fluorescent tubes (F15T8/BL), one facing the sheet, one facing outward. They lighted the entire sheet uniformly, and moths quickly settled down on the front, the back, and on four 1.25 x 0.5 m (4' x 18") "aprons" snapped side by side to the bottom inside edge of the sheet to serve as ground cloths. They also settled on the rope stays used to stabilize the posts against wind, and beneath curved pieces of bark laid on the ground cloths. McFarland (pers. comm.) concluded that the best parabolic curve was that shown in Figure 7–10. The setup was fussy and cumbersome to erect, but it was excellent for season-long collecting at a single site. Wooden poles can be used.

Light-traps

These are made in many configurations and degrees of portability, from self-contained bucket-sized devices to permanent walk-in rooms. All are similar in principle: the moth attracted to the light passes through a slot in a trough or falls down a funnel into a container or space from which egress is possible but unlikely. Entrapped moths settle on the walls of the container or room, or on empty egg cartons stacked in the container; they remain quiescent but alive and well. The collector, before the trap is heated by the sun in the morning,

selects the individuals he wants for specimens and for egg-laying, and releases the rest. There is no question that birds enjoy the bounty of a trap as it is being emptied, but it is difficult to document any overall deleterious effect on the local moth populations.

If the trap is to be used as a killing trap, it is commonly designed with a reservoir for ethyl acetate. All insects that enter are killed. Such traps are used in various research projects and population surveys, or in situations where the trap can be visited only every few days. In the latter instance the light is turned on and off by an electric eye. Killing traps waste a lot of insects and are best reserved for special, planned projects.

A simple and effective killing trap consists of a light source with vertical baffles placed over a wide-stemmed homemade funnel, the stem of which is inserted through and fixed to the center of the lid of a 2 liter (2 qt) jar. The jar is prepared by lining it with a 6 mm ($1/4$") layer of plaster of Paris, rotating the jar slowly until the plaster has hardened. Because the heat and expansion of the drying plaster can break the jar, wrap it with tape to avoid cuts, then stand it in 15 cm (6") of water for a day or so until the plaster is hard and dry. For use, 20–25 ml (4–5 tsp) of ethyl acetate is poured down the side of the jar, which is then rotated for even distribution. A jar so charged will be good for a whole night in warm weather, and several nights if it is cool (Denmark 1956).

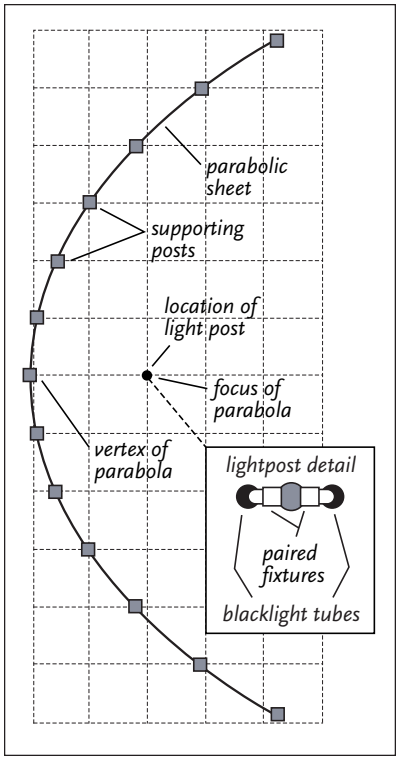

Figure 7–10. Parabolic sheet setup.

Hardwick (1968) reviewed the principles of light-trap design. Lightweight, weakly flying moths are best collected in a box-trap having one wall provided with two glass panes sloping inward to form a narrow horizontal aperture, with the light source within. Rather than MV, a BL is better—the small moths seem less attracted to a light of high surface brilliance. For a "funnel trap", the light source is suspended over a funnel that leads to a

lower chamber. Vertical baffles increase the catch by causing a colliding moth to lose air speed and drop down the funnel. Hoods designed to protect the bulb and keep water out of the trap reduce the catch: many specimens of larger moths descend towards the light from an acute angle, and "if the trap is roofed, such an approach path is eliminated." Hardwick stated that the 125-W Osram MV bulb can survive heavy rain, but others have had opposite experiences with some types of MV bulbs, and they are quite expensive. Japanese MV lamps have exploded with the first raindrop to strike them. Brou (1992b) cited more than one million hours of MV lamp use with no thermal damage in the heaviest rain. He sealed the space between lamp base and socket with vinyl electrical tape.

In lower latitudes, where temperatures remain elevated through the night, large beetles often create havoc within a trap. In live traps they plough about like small bulldozers throughout the night, and in killing traps they succumb far more slowly than Lepidoptera. Beetle separators have been designed to meet this problem, and they are built into some commercially available models. For areas where beetles, especially scarabs, are a major problem, Common (1959) described an MV funnel-type killing trap that not only reduced the take of scarabs by 85% and beetles overall by 75%, but also quickly separated and subdued those beetles that were captured. Hardwick (1968) advised compartmentalizing the bottom of the collecting chamber to restrict beetle movement, or covering the bottom with a pad of cheesecloth, in which the beetles burrow or become entangled. Beetle separators are complicated to describe and construct, and it is best to consult the articles above, and Denmark (1964).

While excellent traps can be purchased from various suppliers listed in Appendix L, many lepidopterists prefer to build their own.

Box-trap design features (Martin 1977) are shown in Figure 7–11 and include:

- A rectangular box with the front open.
- Two panes of glass, placed to form a horizontal slot 2–2.5 cm ($3/4$–1") high, and held in place by gravity against wood support strips glued and stapled to the sidewalls and inside the top and bottom edges of the box.
- A BL unit mounted across the inside of the rear panel.

To examine your catch you will need to design the box so that

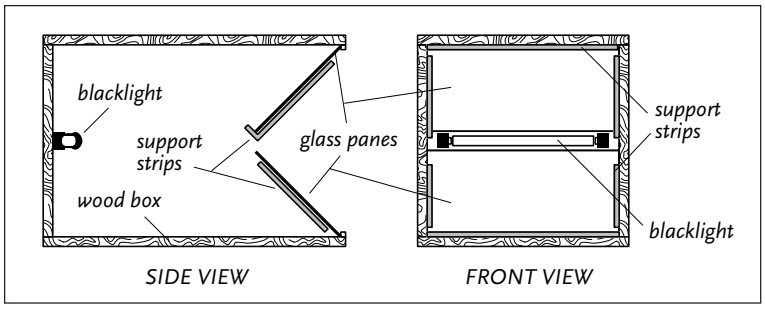

Figure 7–11. Box-trap.

either the rear two-thirds of the top is hinged for opening, or the back panel is held in place with hooks and is removable. The latter is better. Place a couple of egg boxes in the bottom of the trap for shelter.

Because a truncated cone provides the most important element of many homemade traps, cone construction (Figure 7–12) is considered in detail here. Clarify first:

1. the purpose of your cone—tub-trap cover, light-trap funnel, bait-trap funnel;
2. the profile or slope of the cone, usually 30° for a tub-trap cover, 75° for a light-trap funnel, and 60° (or 45°) for a bait-trap funnel;
3. the diameter of the base ("entry ring") of the cone, determined by the size of the ring or tub-rim to which it will be applied;
4. the diameter of the truncated end ("outlet ring") of the cone (if you do not include a zipper in the side of the trap, make this outlet large enough for you to pass your hand and a small killing jar through it).

You have selected a profile slope for your cone, and you have decided on the diameters of the entry and outlet rings. To draw a cutting pattern on heavy paper or light poster cardboard (heed the old adage "measure twice—cut once!"), you need to know the two radii (R) that will give you entry and outlet rings of the desired diameter. You also need to know the number of degrees of circular arc (S) that will produce the desired profile slope.

For a cone with a profile of slope P, entry (or outlet) diameter D times the "slope factor" for the profile equals that radius for the cutting pattern. Sector S equals the required degrees of arc for that pattern (Table 7–1).

If you wish to determine actual cone height, D times "height

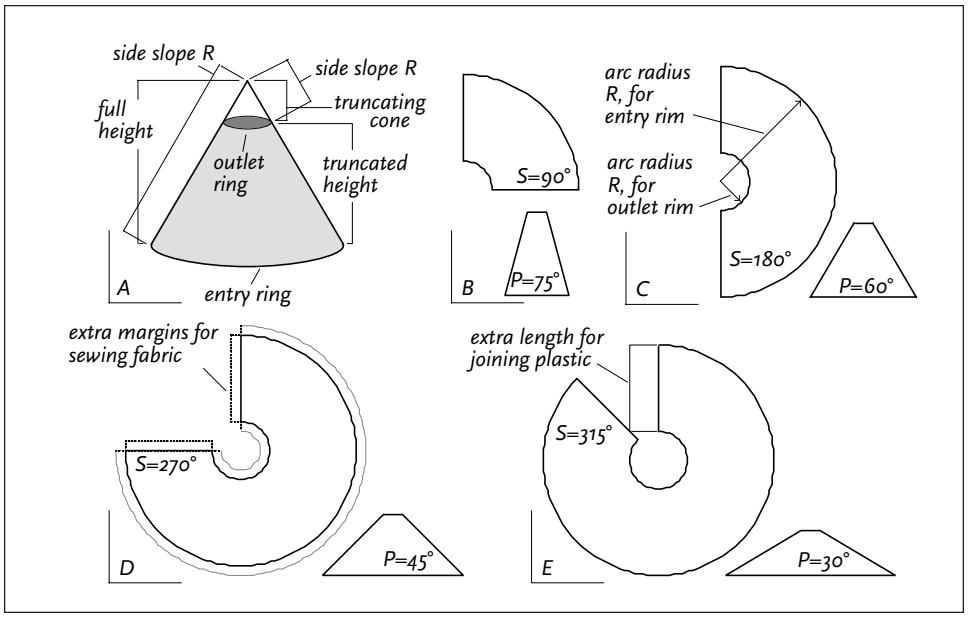

Figure 7–12. Making cones.

factor" for P° equals cone height.

These factors have been derived empirically. Height factor equals height divided by base diameter. Slope factor equals base diameter divided by slope length.

Note: When you are making a template for fabric, add 1–1.5 cm ($1/2$") extra material along all edges for sewing. When making a template for plastic, add 2–5 cm ($3/4$–2") extra along one straight edge for overlap joining at the other straight edge.

The following is a very serviceable and durable adaptation of the British tub-based "Robinson trap," easy to construct and operate (Figure 7–13).

To build a tub-trap, start with a galvanized steel washtub about 28 cm (11") high with 56 cm (22") rim diameter, or the next smaller size. Through the central 12 cm ($4\ 1/2$") disk of the bottom of the tub drill a number of 4 mm ($3/16$") holes as rain drains. Cut a 3 cm ($1\ 1/4$") high cylinder from the top of a plastic food container and center and cement this to the bottom of the tub to make a well about the drain holes (use silicone tub-caulk).

For the dome, from flexible UV-resistant plastic make a truncated cone with the lower diameter overhanging the tub rim by about 2 cm

Profile P	Slope factor	Sector S	Height factor
30°	1.7	335°	0.3
45°	1.4	270°	0.5
60°	0.88	180°	0.88
75°	0.5	90°	1.5

Table 7–1. Cone factors.

($3/4$") all around, and the upper diameter 20–30 cm (8–12"). The shape of this dome can be maintained by overlapping the ends of the material and securing it with pop-rivets. Make a similar, smaller inverted cone with its larger diameter just larger than the hole in the dome, and its lower diameter about 5 cm (2"). Invert this in the hole in the dome as a funnel, and hold it in place with plexiglass cement. Caulk any gaps with tub-caulk. From a small plastic bottle cut an open cylinder to tape to the bottom of the funnel as a stem. This stem can reach to about 5 cm (2") from the floor of the tub.

Near the edge of the dome drill three small holes at 120° intervals for attaching the security cords shown in Figure 7–13. Drill three additional holes, farther in from the edge and diametrically opposite the others, to accommodate three short bolts (detail in Figure 7–13). These keep the dome from sliding sideways off the tub. Pop-rivets with spacers can be used instead.

From rigid plexiglass cut three identical pieces for baffles, angled to match the slope of the funnel, and cut out centrally to accommodate the lamp and its base. These baffles are cemented to each other at the bottom (angled at 120° to each other) and at the top to a circular plexiglass top plate bearing a central 6.4 mm ($1/4$") diameter hole for attaching the MV lamp unit. A bolt through this hole and screwed into the threaded hole in the inverted lamp base will hold it in place. Stabilizing nylon spring-loaded security cords keep the baffles and dome in place in gusty winds. Construction of the baffles can be modified to accommodate a vertical BL or a circline BL tube powered by a portable 12V battery or 120VAC.

Stack the bottom halves of paper egg cartons (2 x 6 egg style), bottoms up, around the inner wall of the tub to the height of the rim. If you stack them in an orderly pattern, you will be able to pick up one carton at a time without disturbing the moths roosting on or inside

adjacent cartons.

Place the dome unit on the tub and hook the three security cords beneath the bottom edge of the tub. Place the light unit on the funnel and hook its cords onto the rim of the dome unit.

Retrieve individual moths by knocking or nudging them into a killing jar (for specimens to keep) or individual small paper lunch bags (for egg-layers). Skill is acquired gradually!

A collapsible light-trap (Figure 7–14) has been described by JM Johnson (pers. comm.)—an inverted adaptation of Baggett's bait-trap (Figure 7–18).

Two 46 cm (18") disks are cut from 10–13 mm ($3/8$–$1/2$") exterior plywood or particle board, and a central 35 cm (14") disk is removed from the center of one of them to produce a ring 5 cm (2") wide. These two pieces, protected with several coats of oil-based enamel, make the entry ring (top) and the bottom of the trap.

A 150 x 71 cm (60 x 28") piece of fiberglass screen will make the cylindrical sides of the trap, with a 90 x 71 cm (35 x 28") piece for the interior cone (with surplus for a cone for a second trap), that is cut according to the layout in Figure 7–12. A 40 cm (16") piece of plastic tubing, joined into a circle by a dowel plug, makes a ring 12.5 cm (5") inside diameter for the outlet of the cone. Sew a 56 cm (22") plastic zipper, to open from the bottom upwards, and parallel to the short edge of the material, onto the side of the sidewall screening. Open the zipper, cut down the center line with a razor blade, then with scissors trim the rough screen edges on the inside so they will not jam the zipper.

To assemble, form the cone by sewing the free radius a–b to the overlap line c–d (Figure 7–14) using heavy polyester-covered cotton thread, or join it with hot-melt glue. Position the cone in the inside rim of the entry ring, staple it in place (make small radial slits in the edge as necessary to simplify fitting), and trim off any excess. Smooth any rough edges with silicone tub-caulk. Invert the top and cone, set the plastic ring on the small end of the cone, sew or glue it in place (more radial slits), and again smooth with caulking. Run three strands of monofilament line from the plastic ring to screw eyes on the floor of the trap to stabilize the cone. Now staple the outside

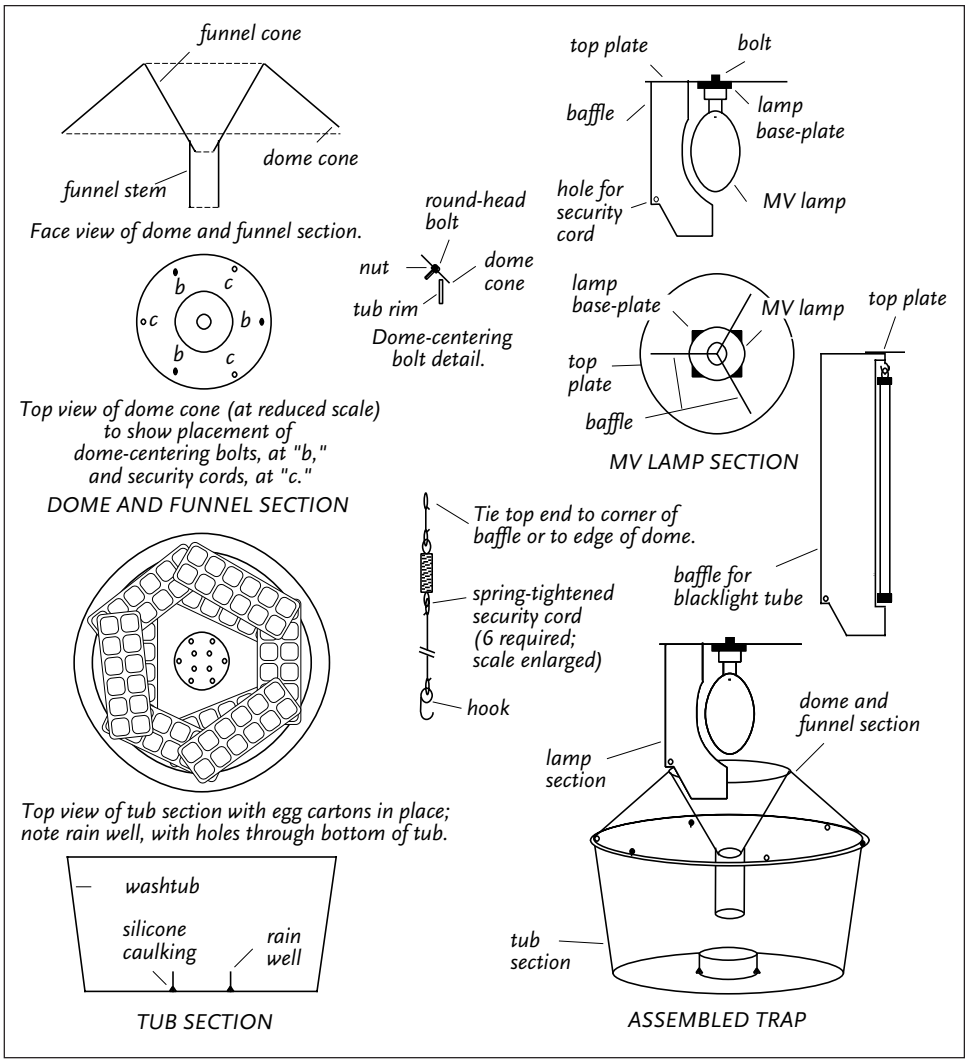

Figure 7–13. Tub trap.

cylinder screen to the circumferences of the top and the bottom of the trap; there will be a 7–8 cm (3") overlap. Set four eye-screws into the top circle of the trap at 90° intervals, and use these to hang the trap while you sew or glue the side seam overlap.

Make four 3 mm (1/8") plexiglass baffle vanes 15 x 45 cm (6 x 18") according to the outline in Figure 7–14. Eight strips of aluminum or galvanized steel 2 x 38 cm (3/4 x 15") from a heating duct workshop (scrap) make brackets for the baffle vanes. Bend them against a

wooden jig as shown in the drawing. The diagonal measures 64 mm ($2^1/_2$"). Attach the brackets to the vanes with 1.25 cm long ($^1/_2$") No. 6 bolts. These are bolted at 90° to one another by means of the brackets. The vanes can be disassembled for packing.

Suspend a 15W BL at the center of the baffles and connect it to a 12V rechargeable battery (spray-paint one corner of the battery to identify the positive pole easily). Using nylon cords through the four top eye-screws, and elongated wire S-hooks, suspend the trap from a branch, a horizontal line, or other support. Visit the trap at earliest light, before it warms up, and remove desired specimens by passing hand and jar through the lower part of the zippered opening, or by picking them off with forceps. Unwanted individuals can be released.

If you collect where showers are frequent, instead of the wood floor you can use a wire hoop to which a floor of screening, and the bottom edge of the cylinder wall, are sewn and glued. Do not alter the upper ring design, however. If you make a trap with the cone attached directly to the upper edge of the cylinder, there will be a narrow angle where many moths will become jammed and damaged.

Productive Collecting Sites

With existing lights, trial and error will show which lights are most productive. If you set up a light to use year-round at home, it should be visible from lawns, edge areas, and woods, if present. Pruning edge vegetation strategically to allow the light to penetrate woodlands is useful. Try to pick a spot where other light sources in the vicinity will not compete. If you are using an MV lamp, it is a courtesy to site it, or baffle it, so that it does not illuminate your neighbor's bedrooms. Identical setups, on opposite sides of a house, even in apparently uniform terrain, will often draw strikingly different lists of species (W.J. Kiel, J.M. Johnson, pers. comm.). Placement height above the ground is rather unimportant for noctuids, but is critical for some groups of non-lepidopterous insects (Hardwick 1968).

When you collect in territory new to you, as when travelling, look for edge situations, to exploit both field and forest; bogs and marshes; woods roads; saddles or passes if wind is absent and temperature does not drop off too rapidly; or canyons and watercourses. But keep in mind that a quiet canyon can become a wind tunnel as radiational cooling develops, and a flowing watercourse can provide a distracting

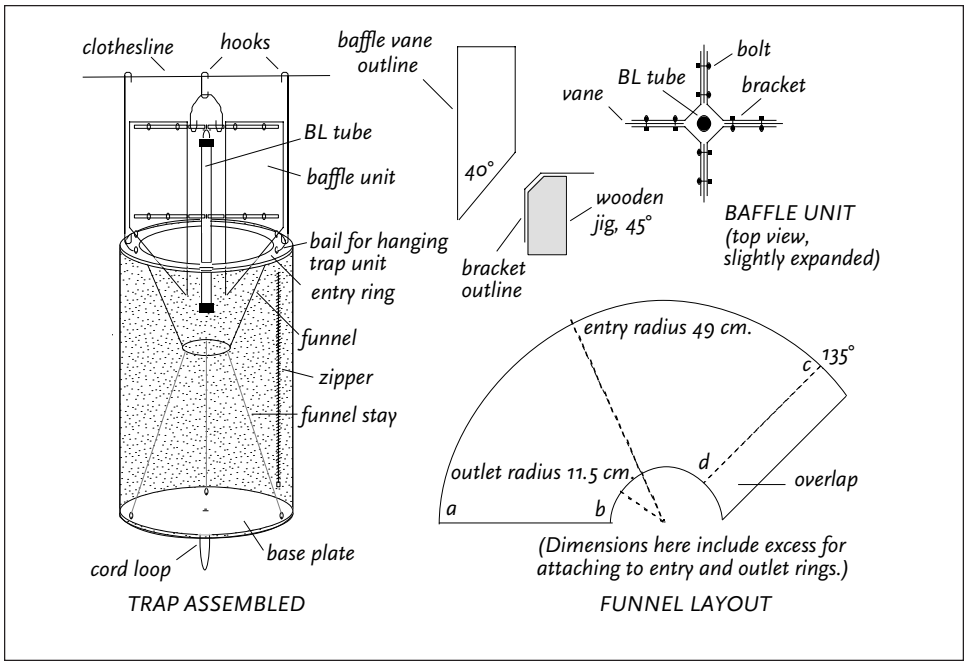

Figure 7–14. Collapsible light trap.

plethora of caddisflies. Moonlight is generally detrimental. A drizzly or rainy night can be outstanding. If you are to spend several days in an area where collecting is exceptionally rich, it can be prudent to collect for just a few hours each night, but at successively later times. The assortment of species coming in can change greatly from hour to hour. Some underwings, for example, remain inactive until a few hours after midnight, and some large tropical skippers arrive in the hour before dawn. The conventional habit of collecting from dark until fatigue overwhelms around midnight leaves the last half of the night unsurveyed!

How do moths react to artificial light sources, and how does moonlight affect this?

As for the eye itself, the individual facets (ommatidia) of the lepidopteran compound eye can be looked on as separate tubular structures. At the base of each tube is a light-sensitive cell, and inside the upper part of the tube is a cuff which can be expanded to exclude light. When the moth eye is dark-adapted, the ommatidia are all open, and if you shine a flashlight on a moth feeding in the dark you see the eye as a red reflection.

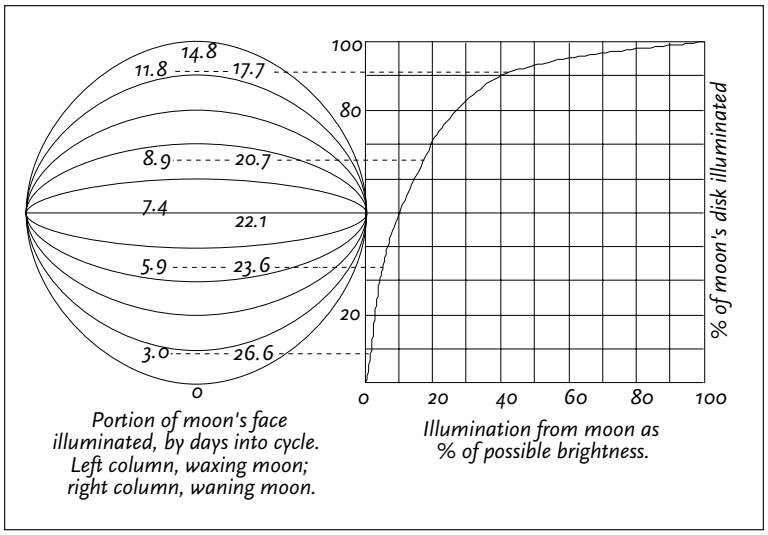

Figure 7–15. Brightness of the moon.

The moth apparently sees an artificial light source with those facets "... which face the light source directly, and perceives a dark area around them due to suppression of the light receptors of the neighboring ommatidia by a process known as optical inhibition, which has been demonstrated physiologically. Instead of being attracted to the light, the suggestion is that the moth is trying to fly into the dark area it perceives around the light." "... the moon reduces the contrast between the trap [light source] and its background and makes it less effective." (Fry & Waring 1996, Hsiao 1972)

With regard to the relative intensity of light in the vicinity of the trap, Rings (1996) reviewed work of J. Bowden that measured the "region of influence" of the trap light, that is, the circle beyond which trap light intensity no longer augmented ambient light from other sources, especially the moon. When the moon was full this "region of influence" was much smaller and therefore impinged upon the flight paths of far fewer moths.

Figure 7–15 indicates the relative amount of light reflected from the moon at various points in the lunar cycle (Bowden 1973, Brou 1994), based on latitude 23.5° from the equator, with variations to be expected for different angles of incidence at lower or higher latitudes. Notice that when half the disk is illuminated, 7–4 days into the cycle, the reflected light is only about 10% of that expected from the full moon, and that only in the last three days before the full moon does the reflected light exceed 40% and go rapidly to 100%. As the moon wanes, its reflectance diminishes a bit more rapidly (on the order of

1–2%, probably not significant). The curve illustrated is the average of the waxing and waning curves. Moth collectors plan their most important excursions to coincide, if possible, with the dark of the moon, the few days either side of the date of the new moon (see Appendix O for a table of new moon dates through the year 2062, and Appendix M for moonlight-related project suggestions).

If you use a live-trap to assess the numbers of a particular species taken at a single site over the course of a season, the question arises as to whether some of your catches are repeat visitors, previously released. One method is to kill and retain the specimens being counted, but this is wasteful and usually unjustifiable. It also obscures the question of whether the captures are residents or transients. Wing marking is an alternative that remedies these shortcomings. Marking may also be desirable for the gardener who wishes to determine the resident status of individuals.

"Sharpie" markers are the most satisfactory and are available in several colors. If your goal is merely to mark a moth or butterfly so that when next seen you will recognize it as having been encountered before, a nonspecific mark on the most easily visualized wing surface—under surface for pierids and lycaenids, upper for nymphalids, papilionids, etc.—is sufficient. For moths, choose a surface visible in the resting position. To give an insect a number for later specific recognition, follow a standard formal numbering system (Brussard 1971) allowing numbering from 1 to 999 with a single color. It is based on the numbers 1–2–4–7. One of these, alone, or in combination with one other, will represent any number 1–9, and it is registered simply by dots placed at the outer margin of the right wings, as viewed from underneath (Figure 7–16). The tens, 10–20–40–70, are handled in similar fashion at the left margin, and the hundreds, 100–200–400–700, if needed, are spotted at the wing bases. Three examples are shown.

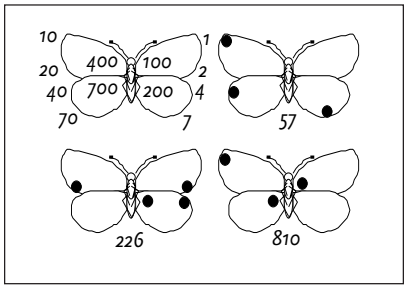

Figure 7–16. Number marking.

Mark-release-recapture surveys for studying population size and behavior within populations are somewhat complex and will not be considered here, but Brussard's article is a good introduction.

6. Collecting with Bait

The use of bait, "sugaring," probably started in the early 19th century with the observation that moths were attracted to the drippings from barrels of molasses. Recipes for homemade mixtures have been burgeoning ever since. Basic ingredients include unrefined sugar (molasses or brown sugar), stale beer (to induce fermentation—but a pinch of dry bakers' yeast will do the trick), and rotten fruit—bananas, apples, peaches, whatever is available (to provide a penetrating odor and more fermentable sugar). Black-strap molasses is very inexpensive at country feed stores and grain elevators. C. Rummel (pers. comm.) 60 years ago used only wine dregs, saved from the previous season, and the results were spectacular. Whatever the recipe, the concocted mixture should be allowed to sit for at least several days at room temperature to ferment. A cover is needed to exclude fruit flies, but the container should not be capped tightly or it may explode. Some chefs recommend adding the beer at the end of the line, or a shot of dark rum, or blackberry or elderberry wine.

Koehn (1988) indicated that addition of yeast was disadvantageous, in that it led to subsequent mold growth that diminished the usefulness of the bait. Yet stale beer as a source of yeast for fermentation may actually be an anachronism, since beer, as it is currently marketed, no longer contains viable yeast (it is filtered out and sold for animal feed: Miller Brewing Co., pers. comm.). Experiments with currently retailed beer selections indicate an absence of living yeast from all of them, with no acquisition of living yeast when allowed to sit for a month and become stale (Winter 1995). Many of us have found yeast useful, and not deleterious, in our concoctions!

Koehn (1988) pointed out that noting what fallen fruits and berries are being patronized locally by moths or butterflies, and incorporating them in your bait, can be productive. He also observed that usefulness of animal feces as bait (bear, deer, horse, raccoon) varied with the season, possibly reflecting diet variations. A mixture of sand, rock salt and urine attracted *Feniseca tarquinius, Lethe anthedon,* and *Basilarchia astyanax* in the mid-eastern U.S.

A "quick grape juice bait" bears mention (Balogh 1990). To a 2 liter (2 qt) container add 120–240 ml ($^1/_2$–1 cup) of granulated sugar, 480 ml (1 pt) dark unsulfured molasses, 1 packet of dry active

yeast, and enough grape juice to make the bottle two-thirds to three-quarters full. Store loosely capped in a warm place for two days. It should be bubbling actively within one day. Before use thicken with brown sugar.

An interesting variant was described by Gozmány (1949). On each of a dozen pieces of string 30 cm (1') long, he strung four or five pieces of desiccated sliced apples or whole small dried pears. A mixture of 1 liter (1 qt) of beer plus 0.5 kg (1 lb) each of honey and sugar (not stated whether brown or white) was heated to dissolve, then allowed to stand a few days to ferment in a ventable container. (No mention was made of reintroducing yeast, which presumably was killed in the heating process.) When collecting time came, the strings of dried fruit were soaked for half an hour in the liquid mixture, then carried in a can containing a little of the mix. The strings were then hung at a convenient height, from any object available. For night collecting, to avoid spooking, he recommended a green-filtered light, although yellow or red are more commonly recommended.

If you tend to think big, try Brou's (1987) brew. "In one 200 liter (55 gal) drum, blend 400–600 apples or peaches, 25 kg (55 lb) of granulated sugar, and 40 liter (10 gal) of beer. Ferment for two weeks, or preferably one year. Make next season's bait now."

Classical sugaring consists of applying the bait with a clean paint-brush or with a rag or sponge tied to a stick, to a series of trees, rocks or other objects, laying out a route that can be traversed repeatedly over as many hours as you wish to persist. The best situations are along edges and along woods roads, as opposed to the interior of a wood. Trunks of living trees are excellent (dead trees may be infested with carpenter ants that will monopolize the site). Judicious pruning of adjacent shrubery may be necessary to open access to the trunk. Planning the route in daylight can be an advantage. Bait should be applied just about dark, and your rounds begun in about half an hour. Results improve if you bait the same areas on the same trees night after night. Late in the season or at high altitudes, when the nights become cold, sugaring for underwings can be done in the daytime (Brower 1947).

Collecting a moth from a baited tree is best done without using a net, because the netted moth, in its efforts to escape, quickly rubs scales from thorax and wings. It is better to flick the moth into an

open jar, using the edge of the lid, as described for picking a moth off a sheet. When touched with the lid, the moth flies down into the jar (most of the time). The success of this maneuver varies with the skill of the collector and the degree of inebriation of the quarry. Cautious movements, and a subdued red or yellow filtered headlight, to leave both hands free, give the greatest success.

Incidentally, although imbibing natural or artificial alcoholic brews does indeed intoxicate moths and butterflies, the attractive component of such brews may be something other than the intoxication agent ethanol (Miller 1997).

If you are collecting primarily to obtain females for ova and don't mind scale loss, a special net design can be helpful. Tape the narrow end of a wishbone shaped wire frame to a 0.5 m (18") wooden handle, join the ends of the wishbone arms with a piece of heavy rubber band or cord, and sew on a net bag (Figure 7–17). To use, hold the net handle horizontally, with the opening up, and push the cord-side of the frame against the tree trunk so that it makes a wide arc of contact. Then flick the moth down into the bag.

If there are no handy tree trunks, fence posts, etc., fit sponges or shingles with loops of twine, anoint them with bait, and hang them on bushes.

For sugaring in treeless situations, T.L. McCabe (pers. comm.) described a procedure sometimes termed "wine-roping." It apparently originated in Scandinavia in the 19th century and has been in use in England for hedgerows and shrubby situations (Fry & Waring 1996). Take a 15 m (50') hank of cotton clothesline and boil it in two changes of water to remove sizing and preservatives. Make a thin liquid bait from 2 liter (2 qt) of cheap red wine and 0.5 kg (1 lb) of brown sugar, and soak the rope in it. The rope can be draped on a barbed wire fence on a prairie, or run over the tops of shrubs on a mountain-top. In arid climates it dries very slowly as compared with painted bait. Moths cling to the rope and spook less easily than from tree trunks. The rope can be stored in the bait container and reused repeatedly.

The usefulness of sugaring is by no means limited to the pursuit of underwing moths, even though they are the species for which we most commonly provide bait. The cuculline moths are readily attracted in the autumn, and again in the spring. Don't overlook

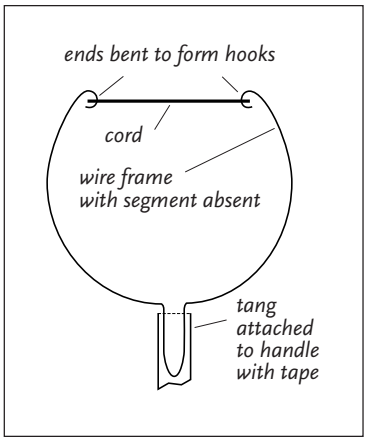

Figure 7–17 Sugaring net.

wintertime opportunities. If the temperature during the day has approached 10°C (50°F) and is likely to remain above 2°C (35°F) for an hour or two after sundown, try baiting. A drizzly night, while miserable for the collector, is also good for collecting (Nichols 1995). I have even chipped *Lithophane* from beneath ice the next morning, when the drizzle had turned into an ice-storm. "Winter moths" stop coming to our provided bait abruptly in the spring, as soon as willows and red maples begin to blossom. Western geometrids are often attracted to bait, as are nymphalids and satyrids.

Bait-traps

If you use bait-traps, sugaring becomes possible around the clock without the collector having to be in attendance. Both moths and butterflies respond. The captives are alive and well and ideal for egg-laying. Some even lay in the trap.

Various designs have been used, but the traps now most commonly used are variations on the old "Rummel Trap" (Gray undated). It was a screen-wire cylinder with a metal lid and an inverted screen funnel in the bottom. This was supported by three 2 cm ($3/4$") blocks above a small dish of bait on a platform. Insects attracted to the bait then crawled up through the funnel after feeding and were retained in the upper chamber. This trap was satisfactory for use on a season-long trap route but was not readily portable, and removing insects was clumsy. Platt (1969) described an inexpensive portable trap, suspended from a branch and having a hanging platform. The wire rings were wire coat-hangers with their handles bent into a paper-clip shape and the arms reformed into a circle that is about 28 cm (11") in diameter. A later model of Platt's trap is depicted in Figure 7–18A. He had replaced the platform with a wire ring supported by three wire radii, into which a styrofoam bait cup was placed. A layer of black plastic film can be attached to the top to keep rainwater from dripping into the bait cup and to give moths a dark place to roost.

A subsequent mutation, the installation of a sidewall plastic zipper,

opening from the bottom up, simplified removal of the catch and cleaning the trap. Metal zippers soon corrode. Velcro proves unsatisfactory for side access (Koehn 1988); it begins to deteriorate after a few seasons of exposure.

Baggett (1985) recommended a circular plywood top, with the bottom of the nylon netting cylinder attached to the perimeter of a plywood ring 5 cm (2") wide. The entry cone was attached to the inner margin of the ring (Figure 7–18C). This simplified cleaning the trap. The plywood also added weight—an asset in a windstorm, a drawback in a backpack. Koehn (1988) and Brou (1992a) gave construction details for heavy-duty models suitable for year-round use (Figure 7–18B). The platform could be round or square. In each of the above three styles, the trap is about twice as tall as wide. Actual dimensions can be figured on the basis of the materials available.

In a bait-trap with a platform, clearance between the platform and the entry ring is determined by the choice or design of the suspending hooks, from a minimum of 2 cm ($3/4$") to twice that height. The bait cup or cups (Brou showed four) can be kept from sliding about by a curb of silicone tub-caulk.

A collapsible butterfly trap for tropical collecting was described by MacDonald and MacDonald (1988). Its rectangular shape was based on the premise that this induced the entrapped butterflies to settle down instead of fluttering incessantly (Figure 7–19).

Two 25 cm (10") square pieces of 1.6 mm ($1/16$") thick plexiglass make floor and ceiling; bevel the edges and round the corners for smoothness. Using a 16-penny nail heated by a propane torch, melt a 4 mm ($5/32$") hole near each corner of each piece (drilling near the edge of plexiglass commonly shatters the material). Cut a small plastic container about 8 cm (3") in diameter down to a height of 3–4 cm ($1\,1/4$"), to use as a bait-pot. Using silicone tub-caulk, build up, on the center of the floor piece, a circular curb slightly wider than the bottom of the pot. This should be done in several layers, allowing ample time for each layer to cure.

To assemble, connect each pair of diagonally opposite roof holes with nylon cord of a length sufficient to make two equal triangles about 15–20 cm (6–8") high when suspended by their centers (using a slipknot on a third piece of cord about 30 cm [12"] long). Hang the top at about shoulder height and level it by adjusting the position of

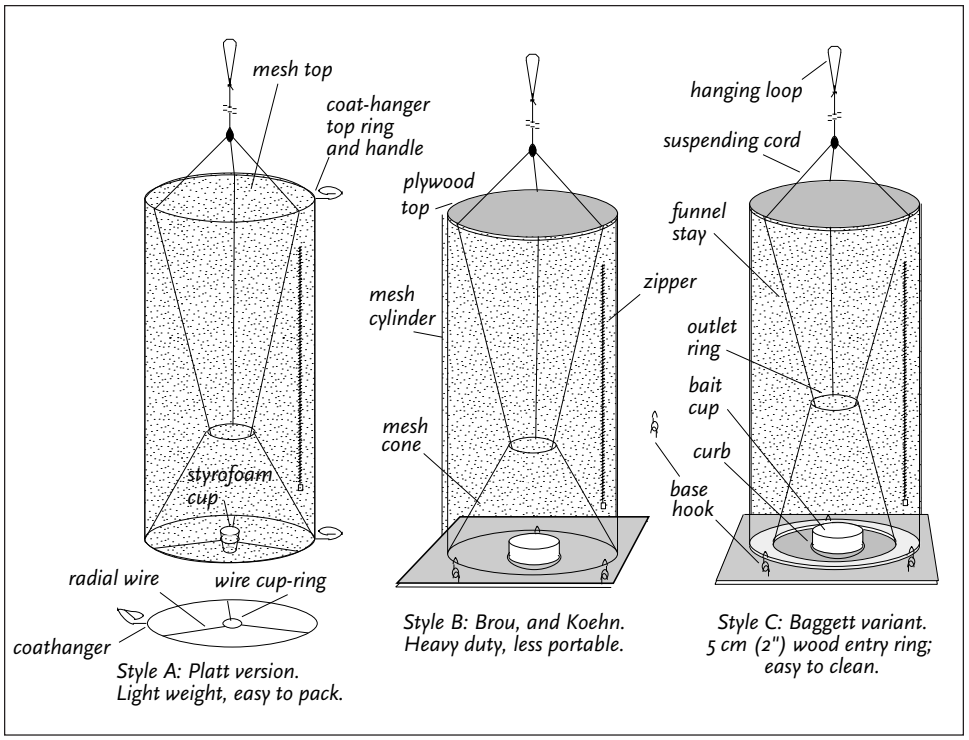

Figure 7–18. Bait-trap variations.

the diagonal cords. Now tie a 95 cm (36") piece of cord (four needed) to each roof corner hole, and tie the other ends to the floor corner holes at exactly 75 cm (30") length. Do a pair of diagonally opposite cords first, then level the floor by tying the last two cords (a small weight placed at the corner you are working on makes tying easier). When roof and floor are leveled and parallel, stabilize each knot with superglue and trim off excess cord.

In the center of an 80 x 105 cm (32 x 42") piece of heavy nylon netting sew an 18 cm (7") long plastic zipper, parallel to the short edge of the netting and oriented to open from the bottom up. Unzip, slit the netting, and trim the loose edges. Wrap the netting about the trap so that the zipper is positioned vertically in the center of one side, and the top 5 cm (2") of netting can be folded down onto the roof. Secure the netting to the corners and the center of each edge of the roof with superglue. Slit each corner of the folded edge diagonally, and cement the netting to the roof with tub-caulk. Sew the overlapping ends of netting together on the side opposite the zipper. Now sew the netting

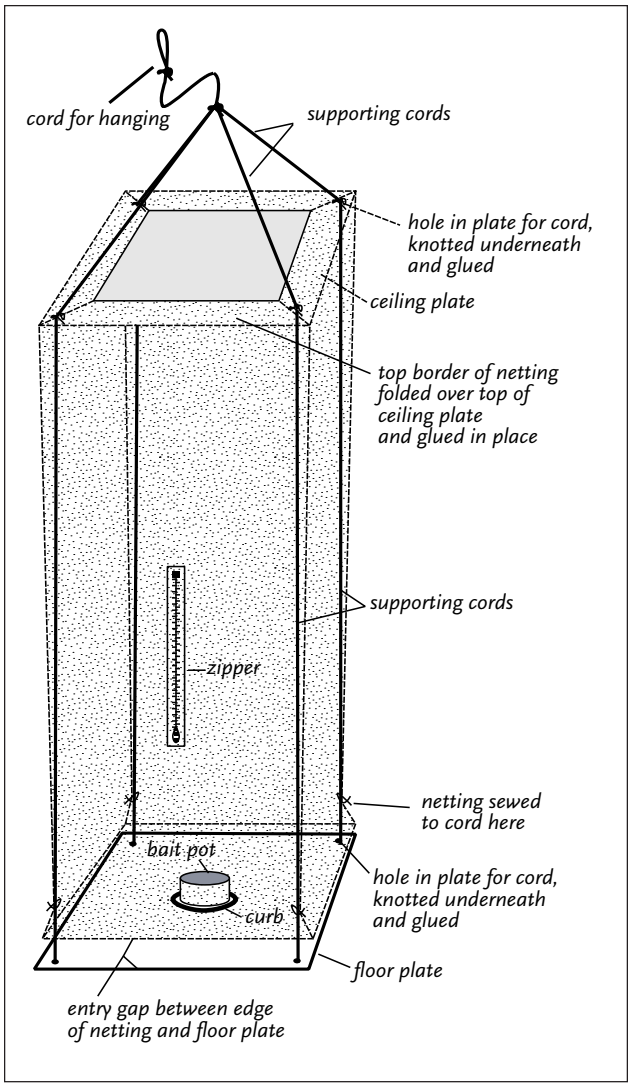

Figure 7–19. MacDonald trap.

to the vertical cord at each corner, at a point 5 cm (2") above the base. Stabilize with superglue. Trim off the bottom edge of the netting to a level 3 cm (1 1/4") above the base, and the trap is complete. "Threebie bait" (beer, banana, and brown sugar) placed in the bait-pot within the silicone ring readies the trap for action.

Baggett's bait-trap design made it possible to hang the trap inverted, with a light source suspended above the cone, converting it into a collapsible light-trap (that is, Johnson's version, described above under light-traps). Efficiency of the inverted cylinder trap, converted to light, can be increased by adding a plastic cylinder to the smaller (inner) end of the cone—a cutoff plastic bottle of diameter slightly larger than the end of the cone serves the purpose well.

Rings (1993) described a rectangular wooden trap covered with aluminum screening (again, of the platform-inverted-funnel principle), that he hung in the woods and visited about twice a month. While this was useful for census taking, the specimens were of inferior quality because of wear within the trap, and predation by

invading hornets. The traps were visited by a great variety of noctuids, particularly cold-weather cucullines.

Cylindrical traps, when made with lightweight netting, collapse into flat "pancakes" so that a considerable number can be carried in a single tote bag.

While the above approaches are effective for butterflies in the temperate northern hemisphere, Sevastopulo (1954) noted differences in tropical Africa. Using open-bottomed cylindrical mesh traps suspended with the lower edge 3–5 cm ($1\frac{1}{4}$–2") above the soil, he found "sugar" bait ineffective as compared to rotting fruit (both sexes attracted) and carrion or feces (males attracted). What kind of feces? Lion and leopard were preferred! (Might this imply any carnivore, or only felines?)

Barcant (1970) described a Neotropical butterfly "trap" consisting of a cardboard box 30 cm (1') high with a perimeter such that his net easily fit down over it. He filled it two-thirds full with rotting fruit and set it in the sun. Every hour or so he made a low-profile approach and clapped the net down over the box. The bait station worked increasingly well if he set it out in the same place day after day.

Trap Placement

Plan where to place your traps according to the Lepidoptera you are looking for—the species must be on the wing, and having their larval foodplants in the vicinity is important. Choose natural flyway sites, especially those that receive late afternoon sun—overhanging a watercourse, in power and pipeline rights of way, at forest edges, in an edge situation on a hilltop, or a hillside ravine (Koehn 1988). Observations at the Wedge Plantation (Flaschka 1989) indicated that for a pair of traps, in apparently similar terrain and only 20–30 m (20–30 yd) apart, one trap would be heavily patronized and another largely ignored. This seemed to support the "flight-path" concept.

Hang the trap so that it is well above the ground and does not brush against adjacent vegetation when breezes blow it about. You may have to do a bit of pruning to keep the trap from snagging on twigs or thorns.

Two simple tricks make hanging a trap or taking it down a matter of a moment. Select a sufficiently sturdy horizontal branch, and clip a side twig that points laterally or downward (the latter is better), to

leave a stub about 2 cm (3/4") long. Pass the supporting loop of the hanging cords over the branch and onto the cutoff stub. This method is absolutely secure, and there are no knots to untie when you take down the trap (Figure 7–20, stub hang-up). If the supporting branch has no usable side twigs, then you can pass the loop over the branch and use a small piece of a stick to secure the loop to the dependent line (Figure 7–20, stick hang-up).

If you wish to hang a trap from a branch too high for you to reach by hand, the M-hook described by the MacDonalds (1988) can be the answer (Figure 7–20). Use a long stick or hollow net handle to lift the hook with its trap up onto the branch.

Life is never perfect, and bait-traps attract more than Lepidoptera. Vertebrate varmints—squirrels, raccoons, etc.—are incorrigible, and the only solution is to repair any rips (as you would a net bag) and move the trap elsewhere. Hymenoptera are the least welcome, particularly bald-faced hornets *(Vespula maculata).* These attack from the cup as you go to replenish the bait; they enter the trap and chop up the trapped insects; and even worse, they chew holes in the tops of nylon mesh traps, allowing enclosed Lepidoptera to escape.

7. Collecting with Pheromones

The pheromones emitted by freshly emerged females are powerful attractants for males of their own species. Their effect is such that a single molecule on an antennal receptor of a male is enough to send an impulse to its brain. The use of calling females to assemble males has already been considered in Chapter 6, on rearing. Virgin females are not easily come by, but fortunately a number of useful pheromones are available commercially. Appendix H outlines the chemistry of clearwing borer moth (sesiid) pheromones, the pheromones (when known) attracting each species of North American sesiid, and available products containing the various chemicals.

Synthetic pheromones are used in agriculture to monitor moth populations. These chemicals are marketed in small objects or strips that hold their potency for years in the freezer and for a few months in the field. You can pin or tie the pheromone source to a net rim or shirt pocket. It takes a while to realize that the wasplike creature hovering and humming softly in front of you is a sought-after moth. But it takes a very quick flick of the net to capture one—they are

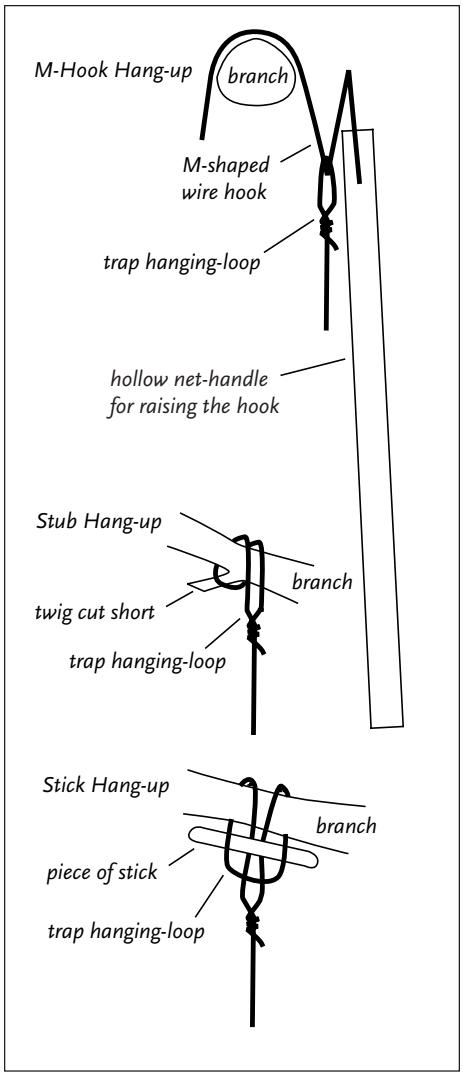

Figure 7–20. Hang-ups.

extraordinarily fast when alarmed.

Old Spice cologne lotion for men was found to be an attractant for several species of sesiids in Arizona (Wielgus 1976), but later changes in formulation rendered it inactive.

Pheromones lure only males, so sesiids should also be sought out in their natural haunts, where females can sometimes be encountered: ovipositing on the foodplant; freshly emerged from the foodplant, signalled by hovering males; resting alone or in copula on or near the foodplant; nectaring (both sexes) at flowers. While *Synanthedon acerni* is primarily taken at light, attraction of other species to light is rare and noteworthy (Baggett 1990).

Pheromone-traps

You can use these compounds effectively in pheromone-traps of several designs. It is important to handle pheromone strips with forceps (wash them with alcohol between varieties), and to wash a trap well before using it with a different pheromone (Taft & Snow 1991). A trap previously baited with "x" and now baited with "y" can mislead you if the moth you take is actually responding to "x" and not to "y." The data you record could thereby be false.

Commercial sticky traps retain the moths as they come in contact with stickum lining the interior. It is possible to release the catch with acetone or alcohol and prepare reasonably acceptable specimens.

A commercial weatherproof plastic funnel trap ("Unitrap") kills the catch with a chemical in a container beneath the funnel—Vapona

(10% dimethyldichlorovinylphosphate, dichlorvos. Alternate trade names are Herkol, "no-pest-strip," Nuvan, Vaponite). These specimens are in very good condition and easy to spread if the trap is visited daily. Males are more easily taken in black, brown or red traps than in white or yellow (Taft & Snow 1991). The dark green plastic funnel traps work well. In contrast, J Holoyda (pers. comm.) recommended "Multipher 1" traps (white with green cover) as highly effective (see Appendix L for sources of traps and pheromones).

An inexpensive salt-soap trap devised by Flaschka (1991) consisted of a flat wooden base, an aluminum pie-pan filled with a concentrated salt plus detergent solution (in water) weighted with a rock, and a wooden cover supported a few centimeters above the pan. The pheromone source was taped to the center of the under-surface of the cover. It was intended for use in remote sites with infrequent visitation. This trap has subsequently evolved (HA Flaschka, pers. comm.). A plastic milk jug is cut off at about 12 cm (5") and suspended from cords attached to its four upper corners by S-hooks. A plastic picnic-plate, with a short radial slit every 90° around the circumference, serves as a protective lid 2–3 cm (1") above the edge of the jug. Dabs of hot-melt or other glue hold the plate at the desired level. The pheromone source is fixed beneath the center of this lid. The cords are tied to a wire hook for suspension from a branch. The setup is completed by filling the jug partway with ethylene glycol antifreeze. In either version of the trap, the attracted sesiid was drowned when a wing-tip touched the liquid. Submerged, it remained undecayed for days or even weeks. The catch was removed (using a plastic fork) to small plastic vials, washed with several rinses of rubbing alcohol, and allowed to evaporate dry before spreading. Resulting specimens were of good quality and were apparently unaffected by dyes in the glycol. Flaschka noted color degradation only in the red band of *Synanthedon rubrofascia;* it fades over time in the best of circumstances.

If you are placing commercial plastic traps where you cannot visit them daily, ethylene glycol antifreeze can be used in place of Vapona. This not only gives better specimens by preventing brittleness, but also quickly immobilizes bumblebees that blunder into the trap, a common problem. The drain holes in the trap can be plugged with tightly fitting screws.

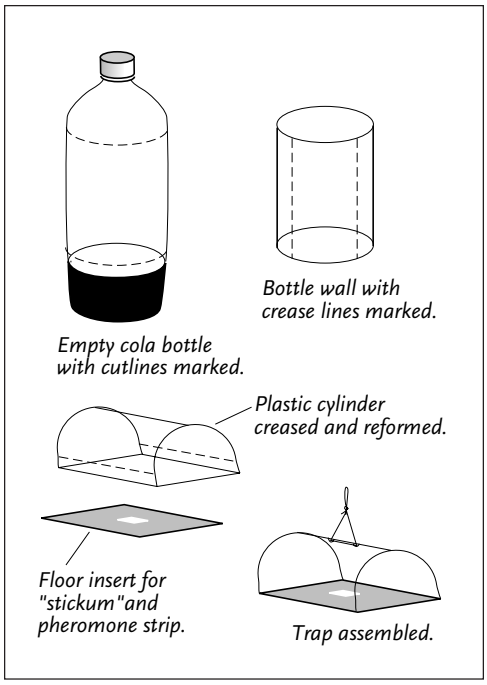

Figure 7–21. Baggett sticky trap.

A homemade sticky-trap can be made from a plastic 2 liter (2 qt) beverage bottle and a piece of cardboard or aluminum (Baggett 1986):

1. Cut off the ends to leave a clear cylinder (Figure 7–21).
2. Make two longitudinal creases to produce a rounded roof and a flat floor. How to Figure the width of the floor? Circumference divided by 2.57 gives the width. If you prefer circular measure, 360° divided by 2.57 gives 140° of arc to measure off for the floor.
3. Cut a piece of sheet aluminum or waxed cardboard to fit onto the floor of the trap, and coat its upper surface with "Tangle Trap" adhesive (available from BioQuip, Appendix L). Hold it in place with paper clips. Place the pheromone source on the center of the floor.
4. Make a pair of holes in the top for a suspending cord, and hang the trap where you wish it to collect for you.

Trapped sesiids were carefully lifted off with fine forceps, and cleaned easily by immersion in xylene (Hazardous: see Chapter 10–8, on chemical hazards, in subsection on genitalia preparations) and spread (or stored in rubbing alcohol) after the solvent had evaporated. Specimen quality was reported as "amazingly good," but not all collectors acquire the needed cleansing skill. Be careful with legs—they have features important for identification.

Trap placement and the reactions of incoming males to the traps need further observation and study. Holoyda (1990) noted that some species were best taken with the trap about 0.3 m (1') from the ground, others at 1.5 m (5'). Some came to traps amidst vegetation, others to more open sites. With two similarly designed and baited traps in apparently similar locations, one was well patronized and the other nearly ignored. Trap-switching showed that it was the site, not

the particular trap, that was preferred. Some species went directly to the pheromone and were easily trapped, others investigated very briefly from half a meter or so, then vanished.

8. Other Gambits

Heliotrope has been used effectively to attract ithomiids in the American tropics (Masters 1968). Plants of *Heliotropum indicum* and other low, shrubby species were dug up by the roots, tied in small bundles, hung upside down to dry for several days, then hung outside with leaves about 1.5 m (5–6') above the ground. Attractiveness increased with age over several months. The ithomiids settled among the lowest hanging leaves and could be found at any time, day or night. Chemical changes may occur during drying, since flowers and live plants did not attract.

Wagner (1973) reported that a "somewhat fragrant" tropical American orchid, *Epidendrum paniculatum*, attracted *Danaus plexippus* of both sexes on their southward migration through Michigan. The monarchs went to great lengths, via an open window, to locate the blossoms within a building, obscured from sight. The same species of orchid allegedly attracts ithomiids in the tropics.

Pitfall traps have been effective for taking Lepidoptera in special situations. In Arizona, a plastic container set into the ground to rim level, and two-thirds full of ethylene glycol, captured a great many *Atrytonopsis python* and *A. diva*. When these were washed in 75% ethanol and air-dried, they were in good condition and easily spread (Flynn & Nielsen 1982). Similar traps, dry or containing ethylene glycol, were used to trap flightless female geometrid "winter moths" in arid areas of California and Nevada (Powell & Ferguson 1994). The dry traps required daily checking, the glycol traps less often.

Color lures are useful for attracting butterflies. A patch of bright blue ribbon on net or clothing attracts male morphos. An orange tossed into the air will often be investigated by a fritillary before it hits the ground, and lesser fritillaries in Alaska have been taken checking out bright yellow rain gear. Small twists of crumpled wet tissue parked on a leaf are mistaken for edible bird droppings by tropical skippers. A red fish-line bob thrown up beside a fig-tree will often activate a resting *Marpesia*. A Coca Cola bottle with its orange-red label, inverted on a stick, will give pause to a passing *Phoebis sennae*.

Calhoun (1990) reported use of yellow or orange lures to attract various species of *Phoebis*. Mounted specimens or single wings (especially female) attached to the handle side of the net rim were very effective. Similarly, Evans (1952) positioned a spread male *Anthocaris sara* in the opening of his net with a pipe cleaner. Any male that came within 1.5 m (5') veered to check out the decoy. Might a life-sized or enlarged color photo work equally well?

Resting *Catocala* can be located in the daytime by direct searching. Best sites are tree trunks in mature forest stands (especially oak-hickory woodlots) with little underbrush, sides of buildings, porches, bridges and culverts, caves, cliffs, and overhanging ledges. Gently brushing a surface with a leafly branch will put to flight a moth too cryptic to discern at rest. It can then be followed to its next resting place for possible capture (Brower 1947).

The Malaise trap (named for its inventor, not for a queasy stomach) is designed for continuous day-after-day capture of all the insects flying through a particular corridor. Its conscientious use is not covered in this manual.

9. Philatelic Collecting

If you occasionally yearn for some leisurely chigger-free collecting with minimal energy expenditure and no exposure to UV, join the ranks of those who collect postage stamps and related items depicting Lepidoptera. Some countries print stamps showing butterflies that are both handsome and native. Others show species that in no way represent their local fauna and give the impression of improbable range extensions. Some display the beauty of moths.

Lepidoptera stamp collecting differs little from other specialized philately. Yet it is fair to say that there is hardly a lepidopteral philatelist alive who has not grasped a butterfly stamp carefully with his forceps and turned it over to inspect the underside wing pattern!

Sources of these stamps include dealers, your own friends in other countries, as well as a network of alerted friends with access to foreign correspondence. Hamel's (1991) *Atlas of Insects on Stamps of the World* references the stamps by country of issue and by taxon and common name of each insect. Further information can be found here in Appendix E, "Lepidoptera Philately," prepared for this book by Charles V. Covell Jr.

It is obvious that a compilation of collecting approaches and devices will never be complete, just as this one is not. When you devise or discover a new or improved method, write it up and send it to the editor of the *News of The Lepidopterists' Society,* so that it can be shared with the rest of the community of lepidopterists.

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Chapter 8.

PREPARING SPECIMENS

Moths and butterflies, when collected, require timely attention so that they can be converted into pinned, spread, and labeled museum-quality specimens that can be stored permanently and retain maximum scientific value. If same-day spreading is not feasible, safe interim storage can be followed by relaxing and spreading at a later date. The process requires specially selected pins, spreading boards constructed to particular specifications, and labels made of properly durable materials and bearing information specific to each specimen. The equipment is not complicated and the techniques are not hard to master, but the more carefully they are carried out, the more scientifically valuable (and attractive) your specimens will be.

Minimum needs

- *Simple spreading board(s)*
- *Good quality insect pins, size 3 or 4 (special purchase)*
- *Glass headed pins*
- *Paper strips*
- *Relaxing jar, for specimens dried out before spreading*
- *Protective box, for storing pinned, spread specimens*

Note: Minimum needs for Chapters 7 and 8 can be obtained as a "beginner kit" from BioQuip (see Appendix L).

1. Spreading

The basic piece of equipment is the spreading board, which consists of:

- a pair of wing boards arranged parallel to each other,
- separated by a central groove, designed with materials to hold insect pins securely in place and at uniform heights, and
- framing members that give the board a level base and complete rigidity.

Since the width of the groove must be a little greater than the width of the abdomen of the specimen being spread, and the breadth of the wing boards must exceed the span of the wings of the spread specimens, you will need boards of various widths to accommodate the many sizes of insects collected (Table 8–1).

The length of a spreading board is optional, but having them all alike simplifies handling and storage. Wing boards are commonly

Groove width	*Overall width*	*To accommodate:*
3 mm (1/8")	5.6 cm (2 1/4")	Lycaenids, small skippers, very small noctuids, geometrids, large "micros"
6 mm (1/4")	7 cm (2 3/4")	Small moths and butterflies, larger skippers
8 mm (5/16")	8.3 cm (3 1/4")	Medium butterflies, heavier-bodied moths, including small underwings
10 mm (3/8")	12.7 cm (5")	Large butterflies and underwings, small sphingids
16 mm (5/8")	17.8 cm (7")	Large sphingids and saturniids

Table 8–1. Commonly used spreading board sizes

tapered 7° from the thinner inner edge to the thicker outer edge. This gives a very slight dihedral angle to the wings of the spread specimen, so that on removal from the board they will not sag below the horizontal plane. The frame members must maintain the wing boards in precisely symmetrical position. If one medial edge is slightly higher than the other, or if the board bends under pressure, it will present eternal aggravation and prevent an optimum spreading job.

The pinning groove or channel depth is critical, since it determines the height of the specimen on the pin. The vertical distance from the upper inner edge of the wing board to the bottom of the pinning groove should be 22 mm (7/8"), so that the wings of all species, whether large or small, will be at the same height when the specimens are transferred to a storage or display case. Figure 8–1 shows basic construction details of a spreading board, with two different approaches to body-pin stabilization. Wing boards (and framing) can be made from clear white pine, basswood, or balsa. Tapered clapboard cedar siding also makes satisfactory material for the wingboards and has a proper slope. An excellent material is tulip poplar ("whitewood")—for its uniform fine-grained texture, its tolerance of repeated pinning, and its rigidity. With whitewood the only surface care needed is an occasional rubbing with 600 grade "wet-or-dry" silicon carbide emery paper to smooth any roughened areas and remove any minute protruding fibers. Then do an annual once-over with a steam iron to close up pin holes.

The first pin-support design in Figure 8–1 uses a layer of fabric

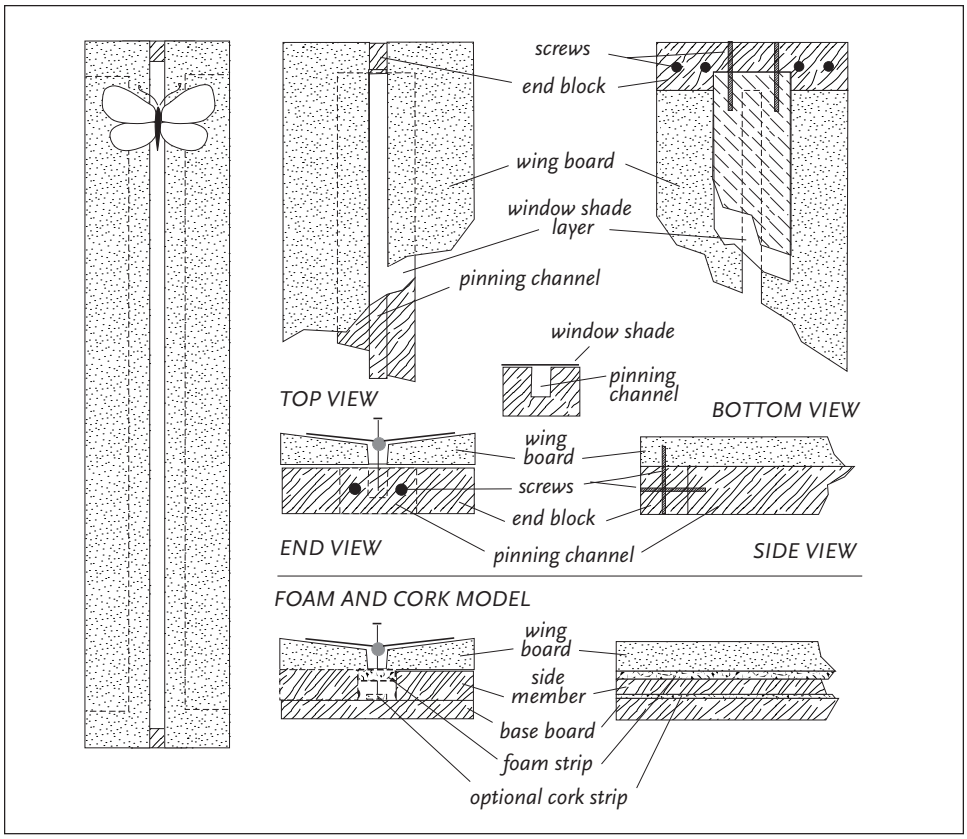


Figure 8–1. Spreading board details.

window shade glued between the under surface of the wing boards and the upper surface of the longitudinal pinning channel. No pinning material is put at the bottom of the groove.

The insect pin is thrust through the fabric precisely in the midline, and the tip is seated gently in the bottom of the groove, giving rigid support at two levels. In case of poor aim you need withdraw the pin minimally to realign it, a very simple procedure. The pinholes in the fabric are reused again and again—a set of such boards in heavy use for thirty years is still in excellent shape.

In the second design in Figure 8–1, instead of the central wood strip and two end blocks there is a plywood base board bearing two wood side members. These support the wing boards and prevent sagging. A strip of plastic foam is glued to the under surface of the wing boards, and a cork-particle strip (optional) is glued to the mid-

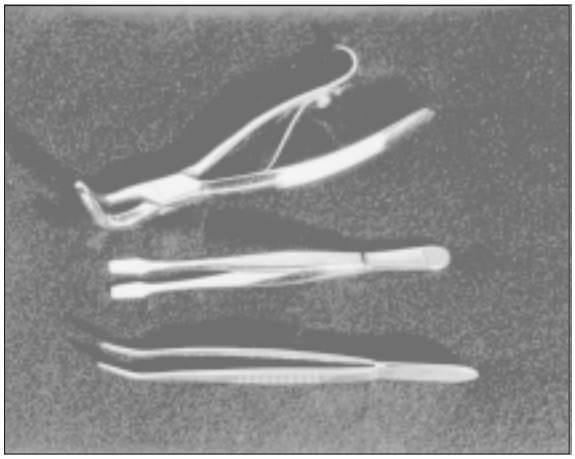


Figure 8–2. Forceps. Top: heavy "pinning" forceps; Middle: spade-tip stamp forceps; Bottom: curved tip forceps.

line of the baseboard. If you fail to place the insect-bearing pin in a position precisely perpendicular to the base board at your first attempt, you have to withdraw it almost completely to make a fresh try. This can be quite frustrating if your aim is faulty.

Lines made with a hard lead pencil and a ruler, at intervals of an inch or less and crossing the board at right angles to the groove, serve as a grid to help you align the wings precisely.

If you are not inclined to build your own, spreading boards are commercially available—wood with grooves of various sizes, or boards molded from plastic foam. The latter are very light weight, but less durable. You can also buy adjustable boards, so that you can vary the groove width to meet the need of the day. You cannot readjust the board while there is a specimen on it. Some prefer them to fixed-width boards. Covell and Cornett (1973) provide directions for building adjustable boards.

A simple but not very durable spreading board can be made quickly from a thick smooth sheet of styrofoam. Simply heat a metal rod of a diameter equal to the width of the desired groove and use it to press a groove into the surface of the styrofoam. Care must be taken to position the insect at a proper height on the pin (M. Plagens, quoted by Covell 1976b). This is a use for a pin adjuster. Spreading blocks can likewise be made from styrofoam, and using this material can be an easy way to introduce spreading to children.

Other styles of boards, as well as spreading blocks, will be discussed later in this section.

Tools and Supplies

A few spreading tools and supplies are also necessary. For han-

dling dead moths and butterflies without rubbing scales off with your fingers you will need some appropriate forceps (Figure 8–2). Some prefer spade tip "butterfly forceps" (similar to stamp forceps), about 10 cm (4") long with smooth tips. Others prefer "pinning forceps," slightly longer, with goose neck shanks and serrated tips. Both are inexpensive and available from supply houses. (These are to be distinguished from the more expensive pliers-type pinning forceps, designed for safely handling specimen pins when they are being seated into the pinning bottoms of specimen drawers or shipping boxes.)

Insect pins (Figure 8–3) are an absolute necessity and must be selected for quality. "Standard" quality are single coated for rust-proofing. Double coating gives greater protection. These pins are black and have small gold-colored nylon heads that remain well-attached. Stainless steel pins, the most expensive, are the best for very humid climates. They are immune to rust and corrosion, but their points are less sharp. Insect pins are about 38 mm ($1\frac{1}{2}$") long and are numbered from the exceedingly fine size 000, for very small moths, up to size 6, for large sphingids and saturniids. There is also a still heavier size 7 that is about 50 mm (2") long. Some storage boxes cannot accommodate pins of this length, and for most large Lepidoptera size 6 is quite adequate. The pins are sold in packs of 100 of one size, or at a lower "per thousand" rate of 10 packs of 100 each, sometimes with the option to select various sizes as you may choose.

Figure 8–3. Pinning materials. Left to right: wood pickle-fork; block for sorted pins; insect pins; rubber-band pickle-fork; "minutens;" pin adjuster.

In addition, for use with the very small "microlepidoptera," there are the *minuten nadeln* (commonly termed "minuten")—very fine

Figure 8–4. Spreading tools (clockwise from lower right): Short- and long-handled setting needles; L-shaped tool; camel hair brush; wax-headed sewing needles; #11 surgical blade; glass-headed pins.

short headless pins just under 12 mm ($^1/_2$") long and 0.15 or 0.20 mm in diameter. The minuten bearing the specimen is then secondarily mounted on a rectangle of pinning material supported on a regular insect pin—a so-called "double mount" or "staged mount." Choose minuten made of stainless steel. The enameled black ones easily corrode and break, with loss of the specimen. The use of minuten and double mounts is addressed in Section 2, below.

A pin block is a convenience, to keep pin sizes sorted. Holes 25 mm (1") deep and 12–16 mm ($^1/_2$–$^5/_8$") in diameter, drilled into a block of wood fitting snugly into a small hinged plastic box, will each hold a pack of 100 pins, readily accessible but also secure for traveling. If your plastic box is a bit too high, a layer of plastic foam glued inside the lid will keep the pins in place while in transit.

A pin adjuster is very useful—a short 6 mm diameter ($^1/_4$") dowel or plastic rod with a 1.5 mm ($^1/_{16}$") hole drilled lengthwise into the end to a depth of 13 mm ($^1/_2$"). The drilled end is then tapered slightly (a pencil sharpener works) and sanded smooth. When the insect is pinned, put the pin head all the way into the hole and slide the body of the insect up against the end of the dowel. This is useful if you are "field pinning" insects for later relaxing and spreading. It is a necessity if you use spreading boards with a pinning groove with a deep foam bottom into which a pin can be thrust to a variable depth. Without it, your spread specimens will not all be at the same height on the pin. If you use boards with a hard bottom to the pinning groove, controlling the depth to which the pin can be inserted, you don't need to use the adjuster. This tool is listed in some catalogues.

For initial manipulation of wings on the spreading board (Figure 8–4), you can employ 4 cm ($1^1/_2$") sharp sewing needles, bearing

homemade heads made from drops of melted sealing wax. For finer manipulation, it is best to use setting needles made by cementing a minuten or a 10 mm (3/8") piece cut from the end of a very fine insect pin into a lightweight wooden handle, such as a match stick or a shish-ka-bob skewer. An L-shaped tool, made similarly from a slightly longer piece of insect pin with 1.5 mm (1/16") at the end bent at a right angle, is helpful for getting errant legs down off the board and into the groove. Antennae, including the hairlike or finely feathery antennae of smaller moths, can be readily and quite safely manipulated with a fine camel hair brush dampened with water or rubbing alcohol.

To hold the wings in the desired position on the board, you will need paper or other material, and pins to hold them in place (Figure 8-5). The paper should be smooth, with no printing, and should not change its dimensions with changes in humidity. Some papers take up moisture and expand a little, so that a strip that was once stretched tautly between two pins becomes bowed and slack, and the wing beneath it slips down. A bit of trial and error, moving some sample pinned strips from a dry workplace to a humid bathroom, will help you identify a reliable grade of paper. Glassine paper is very satisfactory. Strips can be cut from the 24 x 27 cm (9 1/4 x 10 1/2") glassine envelopes made for holding whole sheets of postage stamps, if you cannot find plain glassine sheets. Many people prefer to use strips of acetate tracing film or similar material, best used with the shiny side down against the wing. This material is not affected by humidity, and its near transparency makes it possible to try to identify specimens without having to wait for them to come off the board. These strips can be used only once, however. The pin holes leave a sharp burr on the under surface that will damage wings if the strip is reused. Mylar drafting paper (J.M. Johnson, pers. comm.) is tough, stable, and transparent, and the strips can be reused several times. This material can be found in the engineering section of college book stores. A paper cutter is a good investment, to avoid the aggravation of trying to cut uniform straight strips with scissors.

Never use waxed paper; a temperature rise can transfer the wing-scales to the paper *en masse.*

Pins for holding the paper strips in place should be chosen with several criteria in mind. Because you will be using them a lot, they should be comfortable for your fingers. "Common pins" fail this test,

while pins with round glass heads can be excellent. These are about 30 mm ($1\frac{1}{4}$") long, are sold for dressmaking and should be rust-proof. Before buying, test the point by running it point-first, parallel to the skin, along your finger tip. A good point should snag easily. Some pins advertise "silk points," slightly rounded so they won't snag in fabrics. Such pins are to be avoided, as they can loosen and pop out of the spreading board. Insect pins are effective for holding strips, and they are less likely to vibrate out of boards carried in bouncing vehicles. You may find them inconveniently long, and uncomfortable for your fingers. Insect pins used for holding strips should not be reused for pinning insects. Keep a set separate for strips.

Spreading Procedure

The spreading procedure is perhaps the most important technique connected with the collection of Lepidoptera. Mastering it early gives great satisfaction, saves time, and enhances the appearance and scientific value of your specimens. A conventionally spread specimen has the pin inserted through the back of the thorax, centered and oriented to be at a right angle to the long axis of the body and to the plane of the outspread wings. The forewings are arranged so that their hind ("inner") edges form a straight line at right angles to the long axis of the body. The hindwings are drawn forward so that their anterior margins are just covered by the inner margins of the forewings. The plane of the wings is 22 mm ($\frac{7}{8}$") above the point of the pin, as determined by the depth of the spreading board groove. The antennae are spread in the same plane as the wings and parallel to, but not touching, the costal (leading) forewing margins. Legs should be in the groove, trailing backwards as much as possible. The approach is as follows:

1. Hold a fresh or properly relaxed (see Section 8–5) specimen from beneath by the thorax, using thumb and forefinger or pinning forceps, with the wings folded partway up over the back, and pass the pin through the thorax from the upper side, so that it tilts neither fore nor aft, right nor left. The pin should come through in the midline, between the second and third pairs of legs. If the pinpoint strikes a trochanter or femur, it will usually remove the leg, and some diagnostic features may

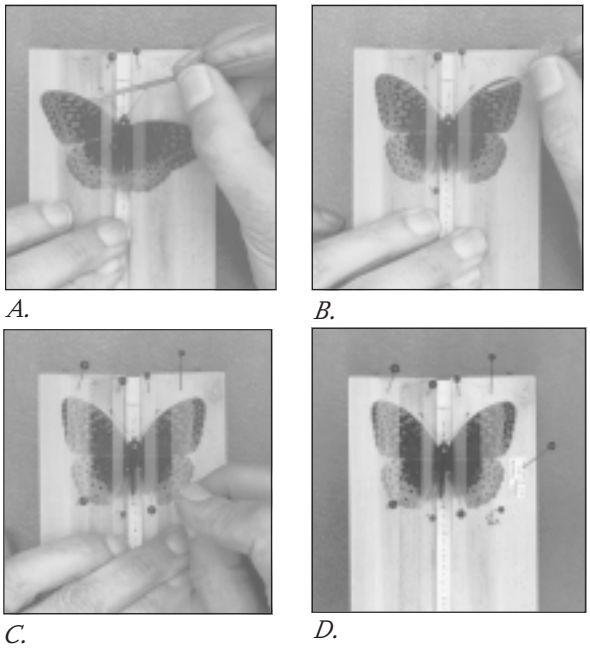


Figure 8–5. Spreading procedure.

be lost. Push the pin through until about 2.5 cm (1") is exposed. Older lepidopterists may experience difficulty focusing sharply enough to insert the pinpoint exactly in the center of a dark thorax. Holliday (1990) suggested coating the point with a minute bit of white "correction fluid," making the location of the tip easy to see and control. He also recommended using a pair of curved-tip forceps, the inner faces padded with narrow patches of adhesive-backed insulating foam, to grasp the lower half of the thorax for pinning. The padded forceps have to be opened widely for use, so they act as a clamp to hold the insect firmly and leave both your hands free. You hold the wings apart with a second pair of forceps with one hand, and insert the pin with the other.

2. Insert the pin vertically into the center of the groove, so that it has no sideways lean when you look down the length of the groove, and no fore or aft lean when you look at it from the side. Holding the pinning forceps on the bases of the forewings at the sides of the thorax, push the thorax carefully downward until the wings are exactly at the level of the board on each side. If at this point the wings are in contact with the board on one side, and not on the other, the pin is crooked in the groove or in the thorax. You need to try again.
3. Place a strip of paper down each side of the board (A, in Figure 8–5), with its inner edge about 3 mm (1/8") from the inner edge of the wing board. Secure the top ends with glass headed pins. Some collectors use single strips wide enough to cover the entire breadth of the wing. Others use the narrow inner strip

and an outer, broader one, offset about the same amount from the inner one, to cover the balance of the wing. At this time try to move the antennae tentatively into place beneath the strips.

4. You may wish to place an additional insect pin temporarily in the groove, in contact with the rear edge of the thorax or anterior abdomen on each side, to prevent rotation while the wings are being moved upward into place. These pins can be removed when spreading is complete.
5. Hold the inner paper strip taut and in contact with the board by curling the end up between your second and third fingers. Use a setting needle, with the point placed just behind the costal margin, to move the forewing up and forward until the inner margins of the wings form the previously described straight line across the board. Keep in mind that any point, as the wing is moved, follows the course of an arc, not a straight line. If you move your setting needle in a straight line, the wing will be torn. Keep continuing tension on the paper strip, and move the hind wing (with the tip of the setting needle behind one of the heavier veins near the front of the hindwing) up into proper position and pin the strip tightly in place. If you push the head of the pin slightly towards the foot of the board as you insert it you will put and maintain strong tension on the strip. Repeat the process for the opposite side (B, in Figure 8–5). If you are using two strips on each side, the outer two should now be pinned in place, making final adjustments of wing position as you do so (C, in Figure 8–5).
6. Using the dampened brush or a setting needle, coax the antennae into proper position. It is sometimes helpful to raise the paper strip slightly by temporarily slipping an insect pin horizontally beneath it, to give a little clearance for moving the antenna.
7. Pin the individualized pin label (to be described in Section 3) to the board beside the specimen. Write the spreading date on one of the paper strips (D, in Figure 8–5).

Special Considerations

If the outer pinning strip does not cover the entire outer margins of the wings, the edges of the wings may curl upward as the specimen

dries. This makes a less attractive specimen, but does not diminish its scientific usefulness. An alternative to a full-width strip is a piece of index card laid on any protruding wing margins or tails. Bevel the margin by running the side of a pin along the edge, to remove any roughness. The weight of the card is enough to control curling. Pin the pieces in place if you store your boards vertically.

If you are spreading very large specimens (*Ornithoptera,* Saturniidae, etc.) and don't want to buy or build an oversize board, you can prevent drooping and curling by placing an 8 x 13 mm (3 x 5") index card beneath the wings that extend beyond the edges of a regular board. Another card placed over the extended wings and held in place with a paper clip will keep them flat while drying.

Some specimens are very recalcitrant when it comes to having their wings brought up to proper position, even when they are quite fresh. This is particularly true of the "branded" skippers. A small incision with the point of a piece of broken razor blade or with a No. 11 surgical scalpel blade, horizontally just below the insertion of each wing, can loosen things up. *Caution: too vigorous or too high a cut can remove wings.* Similarly, using a pair of small fine-tipped "needle-point" steel cuticle scissors inserted (with the points slightly open) into the thorax below the hindwing at the correct angle and depth, you can sever the heavy muscle in one easy cut. This move takes practice, but it is very rewarding once mastered (S.S. Nicolay, pers. comm.). Nielsen (1980) recommended a more conservative variation with the same purpose. With a "minuten" pin mounted in a match stick, he made multiple punctures through the sides of the thorax beneath the wing bases, to "tenderize" the thoracic muscles. He used this for fresh or relaxed specimens of any family. A 000 insect pin also works well.

In many moths, especially medium sized noctuids, and also skippers, if you move the forewing up very far ahead of the hindwing, the base of the hindwing will dislocate and overlie the forewing. To correct this, move a horizontally held insect pin medially beneath the forewing and above the hindwing, until the latter pops back underneath. Then resume spreading, but move the wings forward cautiously and alternately, a little at a time.

Some descriptions of the spreading procedure call for routine impinging of the wings, through the pinning strips, with pins of

considerable size, to prevent slippage. This is to be avoided. When direct pinning is necessary because of insistent slipping use only a No. 000 or 00 insect pin or a matchstick-mounted minuten setting needle placed directly behind a vein in an area not covered by a strip.

The abdomen of a freshly spread moth or butterfly may sag if the spreading board is stored horizontally. An "X" made by crossing two pins beneath the abdomen holds it in line with the body axis and avoids this problem. Another approach (Dodge 1985) uses a narrow wedge of balsa wood slipped beneath the abdomen to raise it into position. The wedge should be pinned in place with an insect pin.

An alternate solution is to hang the boards vertically in a case on a wall. Gravity keeps all the abdomens in line as they dry. Such a case (Figure 8–6), with a carefully fitted screen door, provides good ventilation for drying and at the same time protects the specimens from depredation by mice, roaches and ants.

The inside dimensions of your case should be enough to accommodate the boards you use regularly, with a small amount of extra space to allow easy removal of one board without disturbing others. Leave room also for the screw-eyes from which the boards will hang. Don't forget pin height when measuring for box depth. Ordinary 2 cm (1") stock will do for the box and door frame, and 1 cm (3/8") plywood for the back.

On a flat surface position the door frame precisely on the box and hold it in place with tape. Install two hinges on one side, two flat cabinet hooks on the other.

The important feature, to keep the lightweight door frame from sagging out of shape, is the positioning peg in the inside of the lower corner opposite the hinges. This closes into the alignment hole in the corresponding corner of the box. Drill a single hole through the taped frame and into the edge of the box to get perfect alignment, then remove the tape and glue a dowel into the hole, leaving about 1 cm (3/8"), with the end rounded, protruding toward the box.

Staple window screen (or finer screen, if you can find it) to the front of the door, leaving the outer two-thirds of the frame uncovered. Strips of thin wood, for facing, can be used to cover the rough edges and the hole you drilled for the dowel.

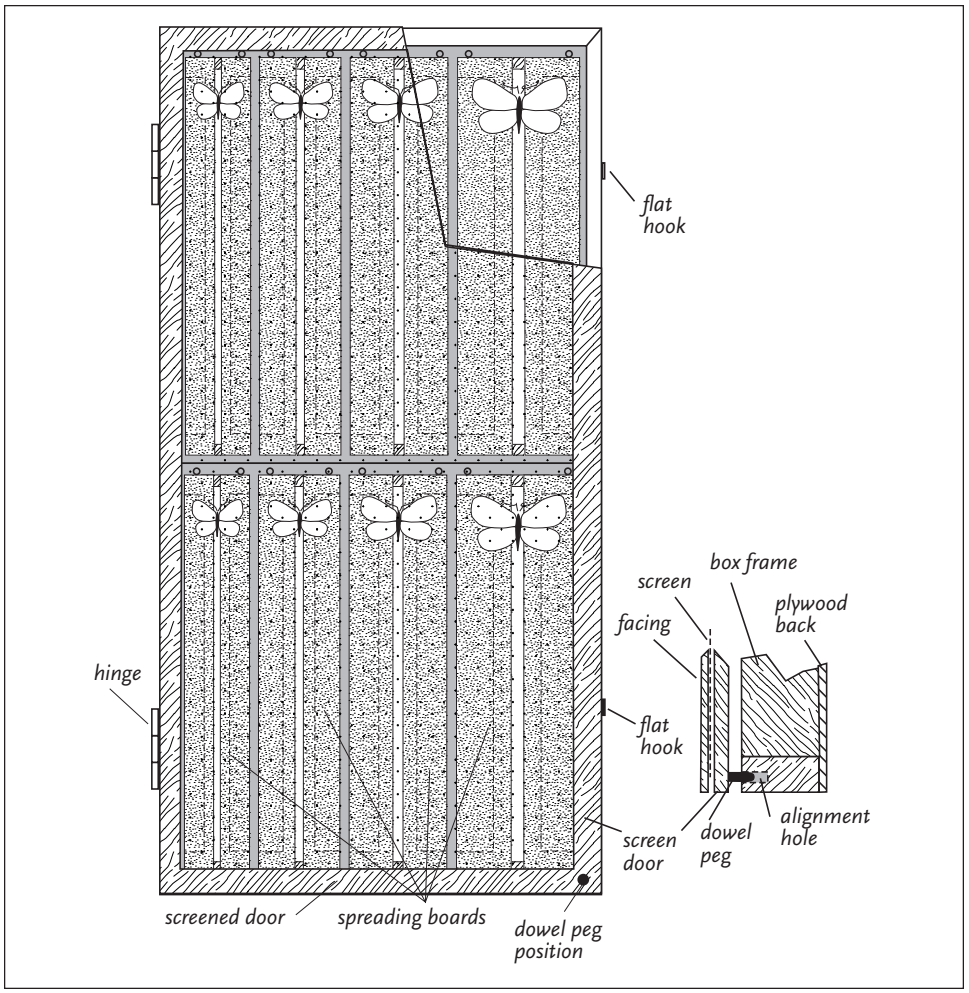


Figure. 8–6. Wall box for spreading boards.

Drying Time

Drying time is a function of temperature, humidity, and the size of the insect body. In moderate relative humidity (50–75%), drying may be complete in three to four days for small insects, eight to ten days for large. A simple test is to apply gentle pressure to the side of the abdomen with the end of an insect pin held vertically. If the abdomen seems rigid, the time is up, but any suggestion of flexibility calls for further drying. Do not apply excessive pressure, or a small abdomen may be snapped off. Drying time for specimens spread while fresh is

significantly longer than for previously dried specimens spread after relaxing.

In very humid situations you may need to use a drying oven—a box warmed with light bulbs and monitored with a meat thermometer. A kitchen oven with a gas pilot light can also serve. Try to use a temperature of ca. 45°C (110°F). Some collectors allow specimens to dry in room air, then place the boards in the drying oven for 24 hours. If oven drying is continued too long, or at too high a temperature, wings tend to revert to their pre-spreading position.

As you remove the paper strips from a sufficiently dried specimen, it is a good idea to use the tip of the pinning forceps to hold down the paper strip beside each pin as it is extracted. Otherwise the strip may cling to the pin and be yanked up unexpectedly, causing damage to wings or antennae.

Leaving specimens on the boards longer than necessary for adequate drying increases the opportunity for infestation and damage by dermestids and other museum pests (as discussed in Section 7). These creatures are not excluded by the screened case described above, and it is doubtful whether perfect physical protection could be devised. It is therefore good practice to put all freshly removed specimens, before adding them to a main collection, into "quarantine boxes" handled by the freezing regime described in Section 7.

Other board styles

A magnetic spreading board (Dorfman 1979) is made by cementing a sheet of galvanized steel, the upper surface polished with emery cloth, to each wing board of a standard spreading board.

1. Tack a broad strip of glassine paper to each side of the top end of the board, long enough to reach the end of the board. Cut rubberized magnetic sheeting 1.5 mm (1/16") thick into rectangles adequate to cover each pair of wings of an insect the size you are spreading. (Metal magnets are too strong and can damage the wing veins.)
2. Place a pinned insect in the groove in conventional fashion and spread the wings flat beneath the glassine paper. Then move each wing into the desired position with a setting needle, and as you go along apply magnetic rectangles over the glassine strips to hold the wings firmly in place.

3. Dry in a 65°C (150°F) oven for one to two hours, followed by a six- to eight-hour cooling period. Higher drying temperatures resulted in shrinkage of the glassine paper and distortion of the wings.

I have tested this method, using a board provided by Dorfman. It works, and it may appeal to some collectors.

Another variation of spreading technique is the monofilament method (Dodge 1985), using a standard spreading board (maximum advised length 30 cm [12"]) and 5 kg test (10-12 lb.) monofilament fishline. (This has some similarities to Nicolay's block method; see below.)

1. Stick a pair of thumb tacks into the framing piece at the head of the board, each set outward from the edge of the groove by a distance equal to the groove width.
2. Tie a loop in one end of each of two pieces of line and hang one over each of the tacks.
3. To the other end of each tie a 30 g (1 oz.) pyramidal lead sinker, adjusted to hang over the bottom end of the board and a little below the work surface of the table. Cut small notches into the ends of the board to keep the line properly distanced from the edge of the groove.
4. Pin the insect and place it on the board. Spread the wings beneath the weighted monofilament lines; move and pin them into place with spreading needles, then hold them with pinned paper strips. Hold the antennae in place with crossed pins.
5. Then move the lines and weights sideways onto the table, in anticipation of spreading the next specimen.
6. When the board is full (all in one session), remove the weighted lines and use them on the next board.

This method had some similarities to and some of the merits of the block method (see below), but in my admittedly limited trial it did not surpass the blocks in speed or flexibility.

Flaschka (1990) described a method for spreading clearwing borers belly-up, to allow legs to be arranged successfully.

1. Prepare a block of styrofoam at least 2 cm ($3/4$") thick.
2. Place on the block, 2 cm ($3/4$") down and in from a top corner, a patch of smooth plastic adhesive tape broader than the wing area of the specimen being spread.

3. Press a groove down the center of this patch with a fingernail or a coin. The groove should be broad and deep enough to accommodate the dorsal thoracic hump of the specimen being spread.
4. Place the specimen upside down on the block, thorax in groove, pin perpendicularly through the ventral side, and push the pin on into the block only far enough to hold the specimen firmly in place.
5. Move the antennae into place and hold with crossed pins, as near the antenna bases as possible; stabilize the body with a pin at each side.
6. Move the wings into place using No. 000 pins; anchor with paper strips passing over wings and antennae and converging above the head to form an inverted "V."
7. Move the fore, middle, and hind legs into position and hold them, and the abdomen also, with pairs of crossed pins. Remove the leg pins as soon as the legs are dry, but keep the wing strips in place through a normal drying period.
8. The specimen pin should be pushed a little farther into the styrofoam every day or two (after just a few hours with very small specimens) to keep it from becoming "glued" in place.
9. When the specimen is dry, first remove the wing strips, then, holding the thorax carefully against the block with very fine forceps, extract the specimen pin and set it aside.
10. The specimen is then turned over and the specimen pin reintroduced through the dorsal surface, using the same pinhole. A droplet of glue at the under surface of the thorax may be necessary to keep the specimen at the proper height on the pin (Flaschka found that clear nail polish was good and tolerated any subsequent degreasing procedures that the clearwings often required).

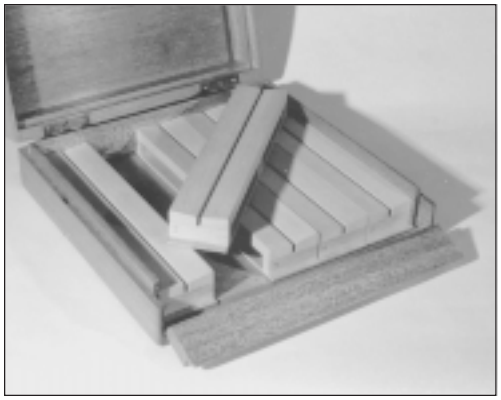


Figure 8–7. Small carrying case for spreading boards.

It is often an advantage to carry spreading boards along while you travel, so that you can spread smaller specimens promptly after capture and avoid the need to relax

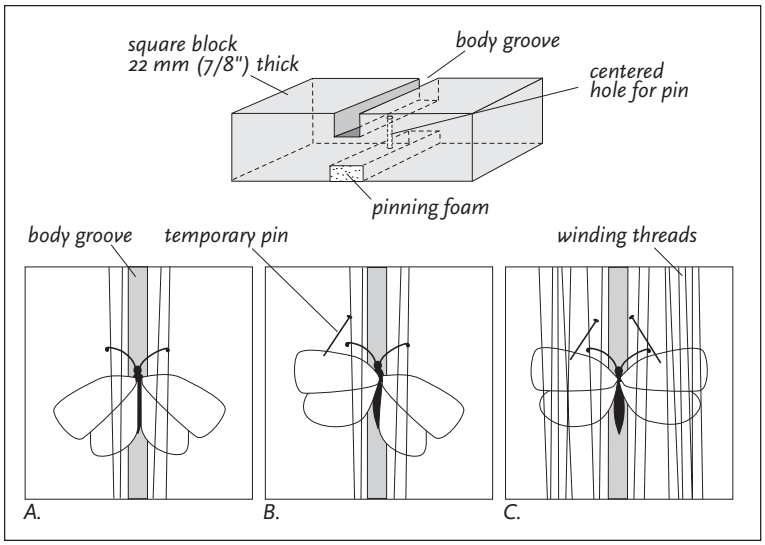


Figure 8–8. Spreading block.

them later. A carrying case can be made to support your regular spreading boards in a stable horizontal or vertical position. If you paint the case black and provide it with a few ventilation holes (screened against mice and ants) it can perform nicely as a drying oven when placed in a closed car in the sun. You can also make special small boards to carry in a compact case that will fit under an airline seat. This is very useful if you collect lycaenids, small skippers, and small geometrids, particularly the greens (Figure 8–7).

Spreading blocks

Spreading blocks (Miller 1971) have the same widths and grooves as spreading boards but they are square and each block accommodates but a single specimen. Choice of wood is as for spreading boards, with clear pine or whitewood serving best. Regardless of block size, all are 22 mm (7/8") thick (bear in mind that nominal one-inch lumber is only three-quarter inch thick, so you may instead need to trim down "five quarter" or other oversize stock, or get a special order). To make a block (Figure 8–8):

1. Cut square blocks of wood, in varying sizes from 3.5 cm to 15 cm (1 3/8 to 6").
2. Down the center of the top of each block cut a groove (running with the grain) 3–16 mm (1/8–5/8") wide, and 4–8 mm (3/16–5/16") deep, depending on the size of the block.
3. Down the center of the bottom of the block cut another groove, about 1 cm (7/16") wide and 5 mm (3/16") deep (same size

bottom groove regardless of size of block). If the nature of the wood you are using makes you concerned that the block might split where it is thinned by the opposing grooves, the bottom groove can be run crosswise and will serve just as well.

4. Drill a very small vertical hole at the center of the upper groove through into the lower one.
5. Fit the mid-portion of the lower groove with a strip of pinning material, so that it underlies the hole: use balsa wood, cork, or pinning foam. It need not run the full length of the block.
6. With a sharp knife blade, cut a number of notches in the end grain at the near and far edges of the block, to accommodate the threads that will be wound about the block to hold the wings in place.
7. Knot the end of some size 50 cotton or polyester thread and wedge the knotted end into one of the notches on the block; wrap the thread around the block 12-15 times, and the block is ready to use.

Pin a fresh or thoroughly relaxed specimen in the same manner as for a spreading board, and pass the pin through the hole in the block and through the pinning material in the bottom groove to the level of your work table.

1. Using forceps, adjust the height of the body on the pin so that the wings can be laid flat against the upper surface of the block.
2. Take two turns with the thread around each side of the block to hold the wings tentatively in place. These turns should be run from the top downward (A, in Figure 8–8).
3. Slide a pin beneath the threads on one side to raise them slightly; move the wings forward into place with a setting needle, and pin them temporarily with No. 000 insect pins. Remove the pin that is beneath the threads. Repeat with the opposite wings (B, in Figure 8–8).
4. Now wrap four or five additional turns of thread about each side of the block and secure the end in another notch. Continue to wrap any remaining thread about the block, and secure it, so that it will not dangle and tangle (C, in Figure 8–8).
5. Arrange the antennae beneath the threads, and attach the pin label to the block.
6. The temporary pins can now usually be removed, but for some

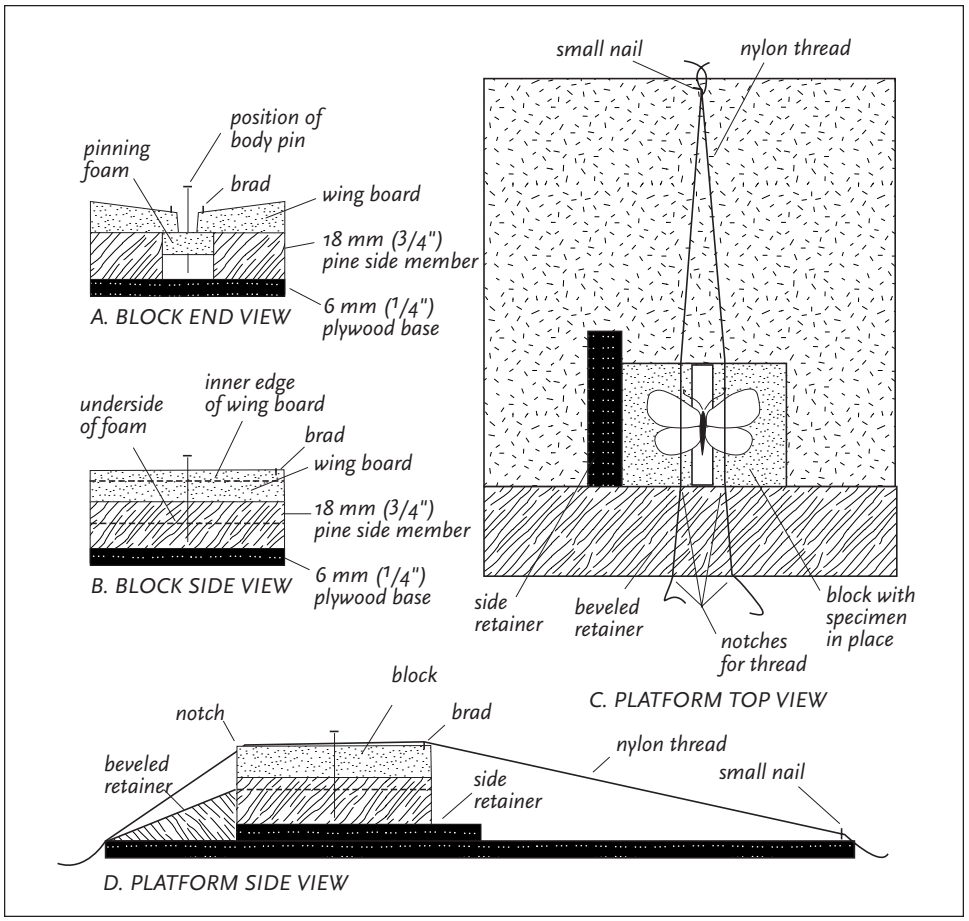


Figure 8–9. Nicolay's spreading block.

robust species they may need to be left in place, especially for the hindwings, until the specimen has dried.

S.S. Nicolay (pers. comm.) has a different design for making and using blocks. Figure 8–9 shows the construction details:

1. Make ordinary spreading boards (A, B, in Figure 8–9) in widths to meet your expected needs, gluing the parts together with strong wood glue. (Glue the pinning foam in place later, after the individual blocks have been cut.)
2. Then cut the boards crosswise into blocks large enough to accommodate one specimen each.
3. Carefully drive a short thin brad (or sequin pin) into the upper surface of the block near each side of one end of the groove.

Make a fine notch in the other end of the block at each side of the groove.

4. Make a working platform as diagrammed (C, D, in Figure 8–9).
5. Rig the platform with two nylon threads (no other material will do!) tied to the nail at the far end and several inches longer than the platform.

You then use the blocks in the following manner:

1. Pin the specimen in normal fashion, set height with a pin adjuster, pin it into the groove with wings at proper level for spreading, and position the block on the platform (C, in Figure 8–9). Lay one of the threads across each side of the block, outside the brads The brads keep the thread from slipping into the body groove.
2. With one hand on the beveled retainer that serves as a hand-rest, and holding one thread under tension across the wings on that side, use the other hand with a setting needle to move the wings into proper position. Secure the thread by pulling it down into a notch at the near edge of the platform. Repeat for the opposite side.
3. Hold the wings in position with paper strips and pins, as you would on a board. Release the threads and set the block aside for drying.

At first thought, the idea of using blocks instead of boards may seem clumsy and disordered, but those who use them, especially those who spread large numbers of specimens, find blocks to be major time-savers. Removal time is also shortened, and if you drill into the corner of each block a small hole precisely 13 mm ($^1/_2$") deep, this can simplify placing the pin label at the proper height. These blocks can be cut to include the 7° dihedral used on spreading boards.

2. *"Microlepidoptera"*

The moths known as "microlepidoptera" are in some instances as large as or larger than some of the "macro" moths, and they are handled and spread in the same fashion. But when you begin to deal with smaller and smaller moths, it becomes necessary to use considerably different techniques to capture, prepare, and preserve them satisfactorily. For this reason, and because many "micros" are not readily identifiable through being obscurely marked or even

undescribed, many lepidopterists simply walk away from the micros. As a result, in North America at least, they have received much less attention than their intricate life cycles and microscopic beauty deserve.

In an effort to reverse this situation, Landry and Landry (1994) published an article providing all the details necessary for you to understand the techniques for dealing with "micros" and making their collection and study routine and practicable. Any attempt to condense that article would emasculate it. It is therefore included in toto as Appendix F. Elements that differ from dealing with "macros" are summarized here:

- The manner of collecting.
- The manner and timing of killing specimens to be spread.
- The importance of waiting 24 hours before killing and spreading reared material.
- Special spreading equipment, and techniques, suitable for use in the field as well as at home. This equipment is simple and inexpensive, and is mostly homemade.
- Understanding the benefits, and the simplicity, of making "double mounts" for these very small moths. (Wright's 1995 procedure for cutting dense polyethylene foam into uniform strips for double mounts is reproduced in full as Appendix G.)
- The fact that undiscovered and undescribed species abound, even in long-studied areas.

Another detailed consideration of all aspects of collecting and preserving microlepidoptera can be found in an Amateur Entomologists' Society booklet on the subject (Sokoloff 1980).

3. Label Content

A spread specimen, with not a scale out of place, wings in classic position and antennae perfectly aligned, may be a thing of beauty, but without its individual "ID card" it is of no scientific value. Each specimen must carry on its pin a small permanent label bearing selected information from your field and rearing records (two labels in the case of reared material). Primary labels include the following four elements.

First is the origin of the specimen. Specify where it lived in the wild: state, province or comparable political subdivision; county, if

they exist; nearest municipality, with indication of direction or distance therefrom, if the habitat is varied (for example, rte 47, 2 km sw of Booneville), or in very sparsely settled areas, location in relation to named topographical features, intersections of roads with watercourses, etc. Notation of altitude is important in mountainous areas. A habitat note (salt marsh, upland hardwoods) can be useful. All locality names utilized should be taken from standard topographical maps or road maps that are widely available. "Uncle Joe's farm" is not a location identifiable by the general public! Obviously, the nation of origin should also be included, but it is unfortunately the habit of most collectors in the U.S. and Canada to assume that the whole world will know where Rhode Island and Prince Edward Island are located. Abbreviations should be those recommended by the postal service or found on maps.

With regard to highway route numbers, these are not as reliable as they might seem. For reasons not related to Lepidoptera, a few states in recent years have chosen to renumber their highways. Future interpretation of some data labels may require the services of a historian as well as a geographer!

In the U.S. many collectors use the township, range, section (TRS) information from US Geological Survey maps. These data are also available on most US Forest Service maps, Bureau of Land Management maps, etc. In fact, for those areas it is easier to find maps with this TRS information than maps with latitude-longitude information. Each township is named on the map according to town lines (north or south of a base line), and range lines (east or west of a meridian line), and each contains 36 sections. These sections are numbered in horizontal rows, starting with section 1 in the upper right-hand corner and "snaking" left to right, then right to left, downward to end with section 36 in the lower right-hand corner. Since the sections are one mile on a side it is rather easy to locate a site accurately, for example: T15N, R20E, SE $^1/_4$ of S19, indicating township 15 north, range 20 east, southeast quarter of section 19, defining the locality as a specific piece of land one-half mile square. Added to state and county, this unambiguously locates the collecting site.

An ideal goal, to be in keeping with international practice, is to pinpoint all locations by coordinates of latitude and longitude. Eventually, as the prices come down on the satellite-based Global

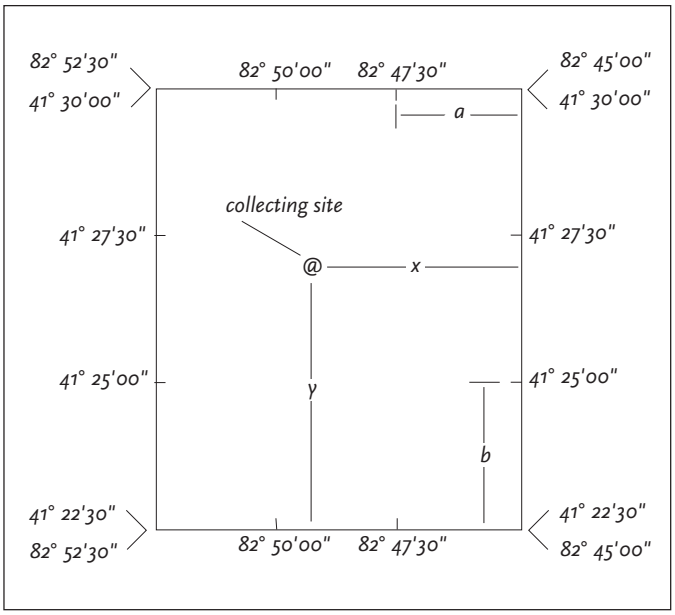


Figure 8–10. Calculating coordinates on a 7 1/2 minute quadrangle.

Positioning System (GPS) receivers, the average photographer or collector will be able to do this with ease. An article by Babcock (1996) gave a glimpse into the scope of this system, in terms of expenditures and rewards. Check electronics and sporting goods catalogs as equipment evolves.

In the meantime, Wright (1992) gave a procedure (modified here) for determining coordinates from US Geological Survey maps, and adaptable to any detailed map bearing coordinates (Figure 8–10). Determine from the information in the margins of the map whether you are dealing with a 7 1/2 minute or 15 minute quadrangle, or some other scale.

For a 7 1/2 minute quadrangle, use the following procedure:

1. Pinpoint your collecting site on the map.
2. Measure in millimeters the distances "a," "b," "x," and "y" as depicted in figure 8–10.
3. Then use the formulas below to calculate the increments for your site's coordinates:

   longitudinal displacement:

   $$\frac{\text{"x" mm (2.5 min)}}{\text{"a" mm}} = \frac{41 \times 2.5}{24} = 4.27 \text{ min} = 4'15"$$

   latitudinal displacement:

   $$\frac{\text{"y" mm (2.5 min)}}{\text{"b" mm}} = \frac{57 \times 2.5}{32} = 4.45 \text{ min} = 4'27"$$

4. Convert the decimal increments to minutes and seconds.
5. To the latitude and longitude bearings of the lower right corner

of the quadrangle add the respective calculated increments, and you have the bearings of your site.

Because the meridians of longitude converge towards the poles, it is necessary to do the edge measurements for each quadrangle as you use it. Even the right edge measurements cannot be safely carried over from one quadrangle to another; trial comparisons have demonstrated discrepancies. "Lower right corner" for base bearings applies only to north latitude, west longitude. In other parts of the world, east longitude requires use of a left corner, and for south latitude, a top corner. Appropriate base lines and vectors must then be chosen for measuring the "x" and "y" coordinates of your site.

If you include coordinates on your labels, they are not a substitute for conventional locality data, but an elaboration and refinement thereof.

Customarily, when defining a location (as in addressing a letter), we progress from the smallest to the largest element, ending with state and nation. It is preferable, on a data label, to progress from largest to smallest, putting first the nation and state. This simplifies the task of sorting specimens geographically in large collections.

A note of caution: the locality data on a reared specimen must be that of its place of origin *in the wild,* not where the adult happened to be when it emerged from the pupa. Errors of this sort, committed by otherwise competent lepidopterists, cast doubt on the validity of their labels. Place of origin of the foodplant utilized should also be recorded.

The second label element is the circumstance of collecting the specimen, unfortunately commonly omitted. A statement as to how the specimen was collected is very valuable, especially for moths. Differentiate between incandescent light (IL), black light (BL), and mercury vapor light (MV), since different species, and even different sexes, can be taken more commonly at one source than another. Time ("1730MST"—24-hour clock, zone and standard time) and situation ("at bait," "at sap-flow," "at carrion," or "resting," "flushed") pinpoint partitioning of the day or night or atypical flight times as well as providing behavioral information.

Reared material should be so indicated: "ex ovo," "ex larva," etc. Larval foodplant should be carefully noted. "Ex larva *from* foodplant X" indicates that the larva was feeding on that foodplant in the wild.

"Ex larva *on* foodplant X" indicates that the larva accepted and matured on that foodplant when reared in captivity.

Date of capture, or of preservation of a reared specimen, is the third item. As noted under labeling photographs in Chapter 2, this is best done in a form such as "4 Sep 1996" in ascending sequence of units and with the month spelled or in Roman numerals.

Fourth is the name of collector, and in the case of reared material, credit should go to the original collector of the livestock, rather than to the rearer, if different. The name should include the collector's initials. The form "leg. J.B. Slugg" was recommended by Remington (1958), as "leg." (from Latin *legere,* to gather) indicates "collected by." The commonly used expression "J.B. Slugg, coll." can imply "collector" or "from the collection of," the latter being the customary European interpretation. "J.B. Slugg, collr." is also unambiguous.

Reared material requires further detail, so an additional label is advised. Various forms and content are described under "Rearing Records" in Chapter 6.

It is often useful to add, below the data label, a secondary pin-label indicating the name of the specimen. It may contain a number (such as the MONA number), genus, species, and species author, and a line "det. (by whom) (what year)" (Covell 1993b). Specimens sent to experts for determination usually come back bearing such labels. The name of the species author will be open if the species was described in the same genus in which it now lies, but if the generic name has been changed, the name of the author of the species is enclosed in parentheses—for example, *Hemileuca lucina* Hy. Edw., 1887, vs. *H. maia* (Drury, 1773). The date is the year of the original description. The determination label should always be lower on the pin (and replaceable), because the initial determination could be incorrect, or some industrious taxonomist may see fit to change the name or relate it to a different genus. The upper data label (and any rearing label immediately below it), on the other hand, contain irrevocable information that should never be allowed to become separated from the specimen.

What is the meaning of the word "type" and all its subdivisions?

The term "type" connotes "the single specimen or any one of a series of specimens from which a species is described..." (Torre Bueno 1962). "Holotype" indicates "the single specimen selected by

the author of a species as its type, or the only known specimen at the time; the type." "Paratype" indicates "...any specimen in a series from which a description has been drawn up..."

If you have the good fortune to have some of your specimens used in the original description of a new species, their pins will be provided with a further label indicating "Holotype" or "Paratype." A holotype should be deposited in an institution, and not retained in a private collection. If you have specimens collected at the precise locality from which the species was originally described, you can properly label them "Topotype."

An optional lowest and smallest label, sometimes applied by collectors or by museum curators on acquisition of a private collection, indicates "Collection of J.B. Slugg."

A recent development, already in place in some of the larger museum collections, adds specimen-specific bar coding on the underside of a lowest label in the pin. It can greatly simplify accessing information in a large computerized data base (R.W. Hodges, pers. comm.). How soon it will be possible or advantageous for the average collector to use this technology is unknown!

4. Label Making

While it required over two dozen paragraphs to describe the content of the data labels, the space allotted is usually four or five lines on a small paper rectangle measuring about 8 x 18 mm ($5/16$ x $7/8$"), and a maximum of 7 x 15 mm ($1/4$ x $6/10$") for "micros!" Judicious use of standard abbreviations and omission of unnecessary articles and spaces is helpful. "Location" may use part or all of the first two lines, or spill over into three. "Circumstances" may fit into the balance of a locality line, spill over into the next, or use part of the "date" line (which is usually next to last). The last line is normally reserved for collector. The size of the label is not absolute, but it should be as compact as possible while retaining legibility. A second, underlying label is the solution when details are numerous, as with reared material (see above). The criterion that must never be compromised is durability, in terms of hundreds of years.

Paper quality is therefore paramount. Ordinary pulp paper has a high acid content, and over a period of just a few decades it can discolor, crumble and disintegrate even when stored untouched in the

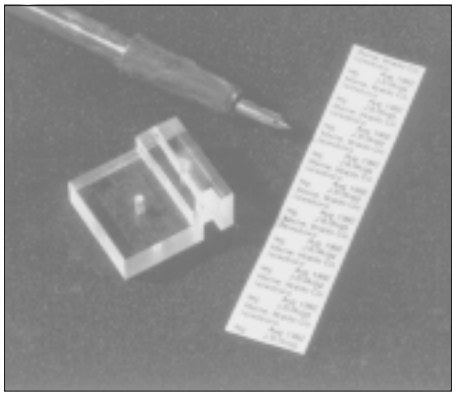


Figure 8–11. Accessories for labeling.

dark in tightly closed boxes. Paper selected for labels should have 100% rag content and be designated as free of acid, rosin, and alum, buffered to maintain pH above 7.5 in storage. Such paper is made for the printing trade and can often be obtained through a local printer. Ordering from the manufacturer is impractical, since they set the minimum order at hundreds of dollars. Papers made for legal documents and ledgers can also meet the requirements. The paper can be "text," "cover," or "index" stock. "Eighty lb. Mohawk Superfine cover stock" is excellent (Webber 1993). It should be heavy enough so that it will grip the pin well, and not curl or tilt, but not so stiff as to damage the points of lighter weight pins. "Alkaline papers" have calcium carbonate filler, and are not as durable as 100% rag. Biological supply houses can also provide appropriate paper, and "archival" paper is now sold widely for laser printers.

The paper must be compatible with the manner of printing you use. Some papers will perform well with laser printers and printer's ink from a hand press, but ink from a pen "bleeds" into the fibers and becomes illegible. Others accept all three pigments. These details should be checked before making a final paper choice.

Hand-lettering is fine for single or small numbers of labels, or for adding a few characters to preprinted labels, but for voluminous collecting or for season-long collecting at one site it is extraordinarily tedious. Ball-point and other ordinary pens are unsatisfactory. You cannot print small enough, and the ink may fade in just a few years. Drafting pens, such as "Mars" or "Rapidograph," are very useful if you will be using the pen on an almost daily basis. However, the fine "000" or "0000" point that must be used is very difficult to keep in working order and expensive to replace. An inexpensive, old-fashioned fine-pointed steel nib in a wooden penholder, wiped clean after each use, is still the best. These are available as "crow quill pens" from some biological suppliers. Williams and Hawkins (1986), reviewing ink characteristics necessary for acceptability in vertebrate

collections (which appear to receive rougher treatment than insect collections), found that inks meeting their criteria were "Rotring 17 Black," "Hunt Speedball Super Black India," "Pelikan 17 Black," "Pelikan 50 Special Black," and "Higgins T-100." It seems reasonable to assume that these products would meet the hand-lettering requirements of lepidopterists.

A disposable Japanese pen, "Pigma," allegedly has satisfactory pigment durability. Available at art and drafting supply stores, it comes in several widths and colors. Use only black for labels. It works well for filling in the date blanks on printed labels, and it doesn't clog or quit. On plastic frames of color transparencies even the coarser sizes do not show up as well as liquid inks. Size 005 (0.20 mm, item XSDK005) is good for pin labels.

An excellent but tedious method of producing labels uses a hand-run printing press and 4- or 5-point type (a point is 1/72"). The type is hand set, and a strip of labels is run off at one stroke per label, manual advance! It now seems archaic, but thousands of labels out there were so produced.

Stanford (1985) advocated typing the labels in the desired form, using reducing xerography to bring them to the desired size—"50% (area) reduction," performed twice, gives labels one-half the dimensions of the original (all this on ordinary xerography paper)—then copying the reduced set onto proper label paper. The results look satisfactory to the eye, but there is concern regarding the durability of the pigment and its adhesion to the paper. Information from the Institute of Paper Science & Technology (F.W. Preston, pers. comm.), indicates that toner will adhere better to the slightly rougher surface of 100% rag paper than it will to the calcium carbonate filled "alkaline" papers. But in contrast to labels penned with permanent ink, xerographic labels can be damaged by rubbing.

The general principles of xerographic and laser printing are similar. Both processes use light to produce an electrostatic charge on a drum. The toner is attracted to the charged areas, then heat-fixed onto the paper. Plastic in the toner serves as a bonding agent. A laser printer merely uses a more intense light.

The inks provided for some ink jet printers definitely are nonpermanent, and colored inks must certainly be avoided. It is a sound move to inquire about pigment longevity from the vendor of any ink

jet printer being used to produce labels (F.W. Preston, pers. comm.).

With the advent of the word processor and the laser printer, creating and reproducing labels has been greatly simplified. Simply set up the label text for the most economical use of space, reduce the text block to the desired size, whether 4 or 5 point (5 point is too large for most work), duplicate the block to as many labels as you need, set up in columns (making sure column breaks do not occur in the middle of a label), and print on a laser printer. Experiment with different type faces to see which is most readable for you when reduced—some prefer a serif font, some a sans serif. If you look at xerographic and laser printing on rag paper under a moderately strong lens, you will see that the margins of the letters are somewhat diffuse, as compared with the sharp margins of press-printed letters. For this reason, a sans serif type face is slightly easier to read if you are using a laser printer. The dpi (dots per inch) resolution of the printer also affects clarity. Line spacing can be manipulated to save space or to enhance readability. There is no need to separate the individual labels by a space, since normal line-spacing leaves enough room to cut the labels apart. When you are satisfied with test results on ordinary paper, then do a final print on proper label stock. Store the labels as closely trimmed single column strips, then cut off individual labels one at a time as needed.

If you are doing season-long collecting at a single site, as with a backyard moth trap, it is convenient to set up columns of labels dated for each month plus the year, but leaving a blank space for the day (to be filled in with the quill pen as you use the labels). It is easy to print more if you run short, and the following spring a few key strokes with the "replace" command will update the year for printing a new set.

Label placement can be done with the aid of a pinning block (Figure 8–11). This is a small hardwood or plastic block 23 mm (7/8") wide and 12 mm (1/2") thick, with a step cut in one end to reduce the thickness to 7 mm (1/4"). Small holes are drilled vertically through the center of the top and of the step. Insert the point of the pin near the center of the data label—but where it will not impinge upon a letter—and push it down through the upper hole. Pin the determination label through the hole in the lower step in the same manner. If the block is made of lucite and illuminated from the side at a low angle, the position of the hole in the block is visible through the label

as a bright spot. Use of the pinning block gives uniform label placement at readable levels. A simpler device consists of two small vertical holes, 13 and 6 mm ($^1/_2$ and $^1/_4$") deep, drilled into the edge of your work table (Covell 1993a).

Because setting labels at rigidly uniform heights sometimes breaks legs off specimens, some collectors prefer positioning labels by eye. In this case a transverse slice from a yucca stalk or a pad made from a rolled strip from the bottom edge of a phone book, bound with tape, serves well.

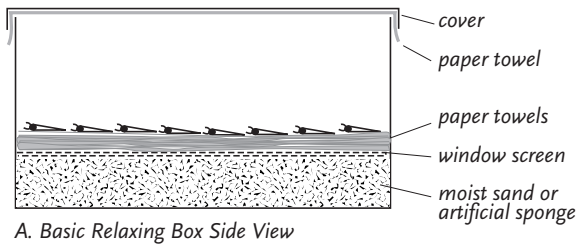
5. Relaxing

Specimens that have been stored in the field in glassine envelopes or paper triangles and allowed to dry, or specimens that have been field-pinned and dried, require relaxing before they can be spread. This is necessary whether the delay has been a day or so or many years. Attempting to spread a specimen whose wings are not freely moveable results either in wing damage during spreading or gradual return to the specimen's unspread posture after removal from the spreading board. Geometrids are particularly prone to the latter reaction.

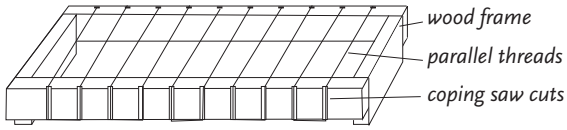
Relaxing involves rehydrating the thoracic muscles and the structures controlling the antennae, so that the wings, legs and antennae can be moved easily and without breakage. The specimen is then spread in the usual fashion and allowed to dry again. Drying after relaxing does not take as long as for a spread fresh specimen.

The process is classically carried out in a "relaxing box," a broad shallow closed waterproof jar or other container with a layer of moist sand or artificial sponge in the bottom, then a few layers of paper towelling (Figure 8–12). A layer or two of window screen placed between sand and towelling keeps the latter from becoming too wet. A freshly prepared relaxing container should be allowed to sit for a day or two before use. If the inner surfaces show excessive condensation they can be wiped dry and the container aired for an hour or two. The goal is 100% humidity, drip free (Covell 1990). A layer of paper towel or cloth between jar and cover will prevent dripping of excess condensed moisture—while the specimens need a highly humid atmosphere, they should not be allowed to get visibly wet.

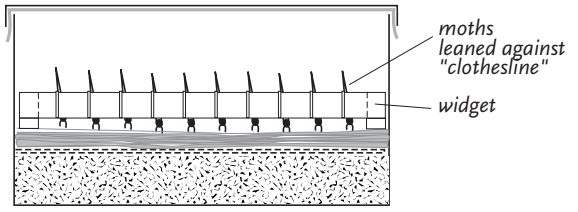
Because mold spores are everywhere and germinate and thrive in a



A. Basic Relaxing Box Side View



B. Preston's Clothesline Widget



C. Relaxing Box, Widget in Place

Figure 8–12. Relaxing box elements.

humid environment, it is necessary to add a mold inhibitor to the relaxing box. Mold on the surface of a specimen is unattractive and very difficult to remove. A specimen invaded by mold filaments quickly disintegrates and is beyond recovery. A few drops of phenol (carbolic acid) solution on the sand or toweling is a very effective inhibitor. Also useful are thymol, Lysol, and chlorocresol. Paradichlorobenzene and naphthalene are used by some, but toxicity (Chapter 10, Hazards) puts them low on the list. The green colors of moths, particularly the geometrines, can be quickly degraded by some of the inhibitors, so their exposure should be as brief as possible or other methods described below should be used.

The dried specimens, removed from their envelopes, are laid on the towelling not overlapping one another. Care must be taken that the collecting data and the specimens do not become separated or confused. Some collectors leave the specimens in their triangles and cover them with a couple of layers of damp paper towels. If you wish to relax specimens within glassine envelopes, carefully trim off the side folds and top fold of the envelope with straight sharp scissors, to allow humidity to penetrate. If the envelope flap contains data, it can be slipped within the remaining envelope fold. If you are relaxing pinned specimens, do not leave them more than two days, or black pins may start to rust. Stainless steel pins avoid this problem.

The average butterfly or medium-sized moth will be properly

relaxed in 48 hours in a well balanced relaxer. Smaller sizes need only one day; oversized moths may require three. Take care to relax only as many specimens as you will be able to spread in a timely fashion. Excessive time in the relaxing jar can lead to wetting, rotting, or other deterioration. Close the container immediately each time you remove a specimen, so that humidity will not be lost.

A rapid method of relaxation calls for a thirty-minute stay between previously moistened layers of paper towels in a container with a tight-fitting lid. Then gently inject a few drops of warm water into the side or bottom of the thorax, using an insulin syringe and a 26 or 30 gauge needle, until beads of water start to come out of the thorax. Put the specimen back in the jar, on top of the toweling, and it is ready to spread half an hour later (Freeman 1985). This does not work well if the thorax has been previously pierced with a pin. The water escapes too easily through the pin holes. Water injection is often useful to complete relaxation in a specimen that was not quite loose enough when removed from the box, especially skippers and large-bodied moths.

Fine gauge hypodermic needles clog and their syringe plungers "seize up" if you try to store them dry after use. Put the combination away filled with water, with enough water in the protective plastic needle cap to keep the point immersed.

Yellow pierids may develop unnatural and permanent green spots on the wings if they become actually wet during relaxing. F.W. Preston (pers. comm.) described a small rectangular wood frame bearing parallel thread "clotheslines." This unit went into the relaxer and the specimens were leaned against the lines with only their bodies touching the damp paper toweling (B, C in Figure 8–12). Cut a series of parallel shallow coping saw cuts vertically on the outside long edges of the frame. Tie a piece of thread around one of these cuts, then run it back and forth across the top, securing each lap beneath the frame via a pair of cuts, to produce the result pictured.

You can advantageously use a frame of this sort for relaxing any specimens removed from their envelopes. To raise the height of the lines to accommodate larger specimens, raise the frame on small wooden crossbars placed beneath the ends.

Retention of green colors requires special mention. Certain notodonts and noctuids, that have a greenish cast when captured, will

lose the green within a few weeks no matter what steps you may take to avoid this. A color photograph, taken promptly, gives the only sure color record. Greens in other noctuids, and in the geometrine inchworm moths, are colors that are very susceptible to degradation by routine relaxing techniques.

The quick method described above is particularly useful for green moths that may otherwise permanently lose their color. Still better for greens is to avoid the need for relaxing: freeze specimens immediately after killing, kill by freezing (or store in an ice chest until freezing is possible), then spread at once after thawing. The manner of storage is important: in a plastic container with tight closure lay a few layers of toilet tissue, a layer of moths, not touching one another, another layer of tissue, with continuing "sandwiching" until the container is just full enough so that the moths will not jostle each other but will not be crushed by overfilling (Metzler 1989).

Another accelerated method works well with rather small butterflies (wings folded up over the back) and requires a small pot of gently boiling water, or better, a mug of water with an immersible electric cup-heater. Installing a rotary switch in the heater cord is an added convenience. Start the relaxation as in the paragraph above, then pin the specimen in conventional fashion. Holding the upper part of the pin firmly with forceps, turn off the heat so the water surface is quiet, and immerse the pin in the water to a depth such that the water just touches the lower surface of the thorax, but not enough to wet the wing bases. The hot water soaks up along the surface of the pin and into the thorax. Hold it there for a count of five to ten seconds, set it aside pinned to some foam for a few minutes, and the specimen should be soft enough to spread. Only occasionally will a second treatment be necessary. This method is particularly useful for specimens too small for easy injection of water and is a modification of a description by Barcant (1970).

6. Storing Specimens

Once specimens are collected, definitive steps must be taken to place them in safe temporary storage, long term storage, or permanent retention as spread, fully prepared specimens. You will usually be able to spread directly, within 24 hours, the catch from a brief local day trip or moth trap in home territory. Specimens in the killing jar

can go directly to the spreading board, with or without a preliminary hour or two in a relaxing box, as necessary. Specimens brought back alive in individual vials can be killed in the freezer, thawed after a few hours, and promptly spread. Pinched specimens in envelopes sometimes need a period of relaxing prior to spreading.

When prompt spreading is not possible, as on an extended field trip or because of the volume of material to be spread, proper storage is essential. Specimens in envelopes can remain as such but must be sufficiently labeled to define the date and place of capture. Jarred specimens are transferred to labeled envelopes. You can speed this process greatly by using a "pickle fork" (as described in Chapter 7) to aid in folding the wings over the back.

Thick-bodied specimens that are placed in envelopes, then stored in boxes while fresh, will often have their abdomens become flattened in a vertical plane. Once they are dry, reshaping is usually impossible, even after a prolonged stay in a relaxing jar. To avoid this, place a wad of cotton in the bottom of the envelope before inserting a thick specimen.

For a different approach (Figure 8–13, A–C), introduce the specimen, wings folded down over the legs (A), into the envelope with the abdomen first, sliding it down to the first corner (B), rotating it 90° to advance the abdomen along the bottom fold until the specimen comes to rest in the second corner, head down, antennae trained along the forewing costal margins, and the abdomen pointing upward (C). The envelope is then left standing on its bottom edge, flap still open, until the body is dry enough so that it no longer deforms if gently squeezed (Flaschka 1992). Because the abdomen stays rounded, a bit of tape may be necessary to hold the envelope closed after the drying is complete. If you are using "end open" envelopes, choosing a larger size makes this maneuver still possible.

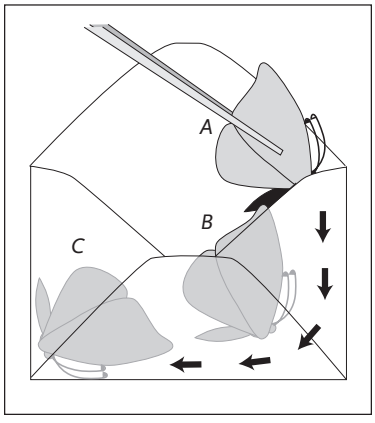


Figure 8–13. Papering thick-bodied specimens.

As a substitute for envelopes, you can use homemade paper triangles. Fold a rectangular piece of paper to the inside along lines a–d, a–b, and c–d in figure 8–14. Write the specimen data on the outside of either of the

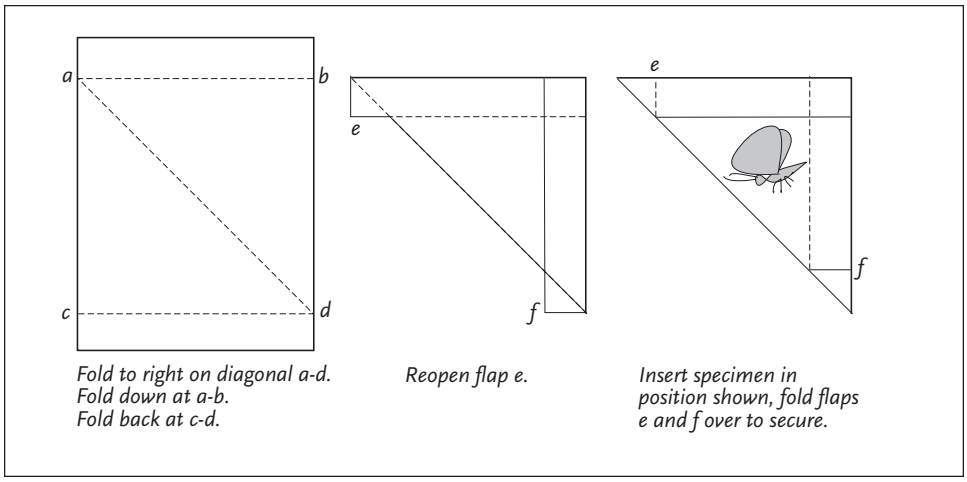


Figure 8–14. Paper triangles.

narrow rectangular flaps so formed. Open flap "e" to form a pocket and place the specimen inside with the wings folded over the back, the antennae parallel to the forewing costa, and the costa lying in the hypotenuse fold of the triangle (Preston 1986). Refold this flap, then fold over the corners "e" and "f" to secure the triangle. Triangles should be made in various sizes to accommodate the specimens at hand, and should be roomy enough so as not to catch wing margins in any of the folds.

It is interesting to note that a rectangle whose edges have the ratio of 7:10 units can be folded in half and cut repeatedly, with each smaller rectangle retaining almost exactly the same proportions as its predecessor. This is worth keeping in mind when you prepare multiple rectangles of various sizes. The flaps come out to be a convenient width for the triangle size.

When extra protection was desired, Preston recommended placing the glassine envelope inside a slightly larger paper coin envelope, or placing the specimen within a fold of tissue or paper towel within the glassine envelope.

Once papered, whether in envelopes or triangles, the specimens need further protection in boxes to prevent damage from jostling or crushing. Plastic sandwich boxes (Tupperware or the like) are convenient. Freshly papered specimens packed snugly but not tightly in a sandwich box, protected against molds by the addition of $^1/_2$ tsp. of chlorocresol crystals and stored in a freezer are protected from all

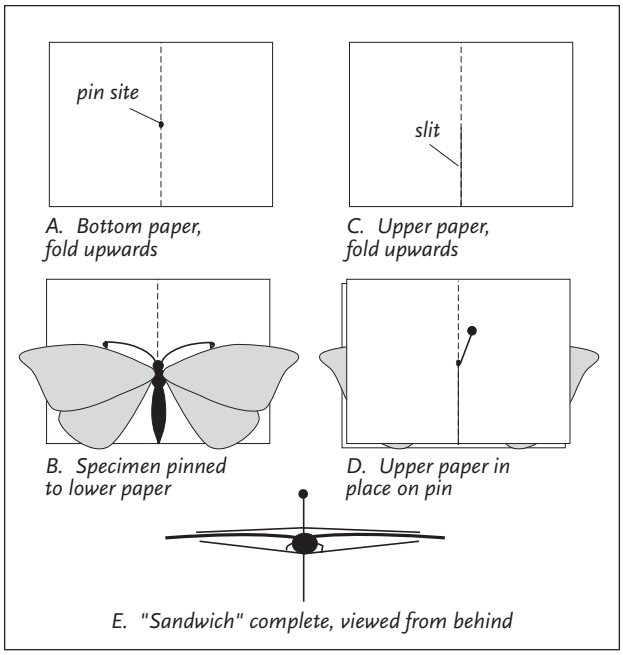


Figure 8–15. Flat papering.

hazards. They often can be spread many months later without need for relaxing. Even without freezing, the chlorocresol box can maintain pliability for months if it is filled to capacity with medium to heavy bodied insects and tightly sealed against moisture loss. Most significantly, it protects against molds.

Tindale (1961), who originally recommended the use of chlorocresol (elaborated on by Fisher 1973), did not paper his specimens, but prepared the box with the chlorocresol crystals on the bottom, a thin layer of cotton to keep this in place, then a layer of tissue. Moths were laid out on the tissue out of contact with each other, covered with another layer of tissue, and successive layers were built up until the box was full. Sealed boxes remained fresh and spreadable for months even without refrigeration. This is a particularly convenient approach for dealing with large numbers of moths. Labeling the layers (lead pencil on paper) is essential. When you are spreading the specimens, keep the excess in a relaxing box so they won't dry out while awaiting their turn! All containers used should be plastic, as chlorocresol corrodes metal.

Meyer (1988) thought that most of the problems with relaxing and spreading dried butterflies related to the wings-over-the-back position. He devised a method for drying and storing with the wings in the horizontal plane:

1. Cut two rectangular pieces of medium-weight paper two-thirds the size of the butterfly's wing span and fold each to make a crease down the shorter midline (A, C in Figure 8–15).
2. Pin a fresh butterfly (at proper pin height) to the center of one

rectangle so that the body and legs lie in the gutter fold (B in Figure 8–15).

3. Cut a slit through half the length of the fold in the other rectangle, and slip it onto the pin, tented upwards, from the head end of the butterfly, to sandwich the wings and antennae between the two rectangles (D in Figure 8–15).
4. Pin the resulting papered specimen (E in Figure 8–15) into a field box, shingled (see below) to save space, and allow it to dry.

Meyer found that relaxing time was reduced to 12–24 hours, and results were excellent. Skippers did not respond easily to this technique.

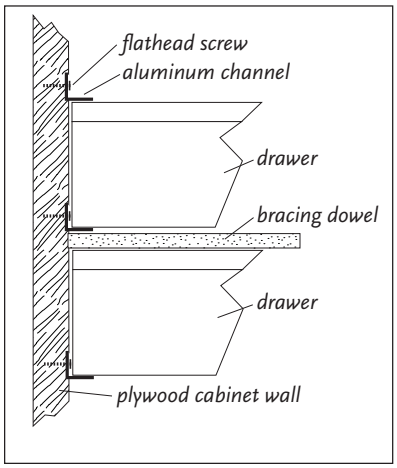


Figure 8–16. Aluminum-channel drawer supports.

Collectors who wish to minimize preparation time in the field often use field pinning as a method of interim storage. The specimens are removed from the killing jar, pinned as for spreading, with the height on the pin standardized with a pin adjuster, then pinned in close array in a field box—a sturdy, latched box with a pinning bottom of material that holds the pins very securely. This approach is particularly useful for thick-bodied moths, since it allows tufts and tegulae to remain in normal position. It also simplifies sorting specimens before spreading (Forbes 1947). Care must be taken that the collecting data are kept clearly associated with the specimens. Back home, when time allows, the specimens, still on their pins, are relaxed and spread in the customary manner.

Unspread specimens, if not in a freezer, must be protected from museum pests and other damage in the same manner as spread specimens (as detailed in Section 6).

The commonly used method of storing spread specimens is in standard insect boxes (the "Schmitt box") measuring 22.9 x 33 x 6.4 cm (9 x 13 x $2^1/_2$") overall, or in glass-topped museum drawers. Two common drawer types are the Cornell drawer, 48.3 x 42 x 7.7 cm (19 x $16^1/_2$ x 3") and the California Academy of Science drawer, 48.3 x 43.2 x 6.4 cm (19 x 17 x $2^1/_2$"). The former has

enough head room for No. 7 insect pins. These drawers can be purchased ready-made or in the form of precut kits to assemble yourself (at little more than one-third the price of the ready-made product). A third type, the U.S. National Museum (USNM) drawer is 45 x 45 x 7.3 cm (18 x 18 x 2 7/8"). Insect boxes and museum drawers have certain construction features in common: the lower sidewalls bear an inner shoulder onto which the cover fits snugly, to exclude insect pests; there is a hard bottom bearing a layer of pinning material that accepts and holds pins firmly; and the covers have hooks to hold them securely in place. The Schmitt boxes, in addition, usually have hinged lids.

Drawers are usually stored in cabinets fitted with grooves or flanges to support them. The cabinets are expensive to buy but are easy to build out of plywood. Figure 8–16 shows a way to make space-saving drawer supports using 1.3 cm (1/2") aluminum channel 1.5 mm (1/16") thick, held in place with flathead screws. Cut the dadoes for the flanges a little deeper than the thickness of the metal, to give a bit of clearance for any screwheads that are not perfectly aligned. Allow a bit of clearance at the top and each side of the drawers, and you may wish to add a front cross-brace every 6–8 drawers to prevent cabinet bowing in humid weather. A piece of 1 cm (3/8") dowel glued in place will suffice. If you have the shop equipment to build drawers from scratch, Brou (1992) provided detailed instructions for making your own Cornell-style drawers.

Holliday (1988) outlined construction of very simple boxes made from 65 mm (2 1/2") wide tongue-and-groove red cedar of the sort used for lining clothes closets. From the groove edge he ripped a 16 mm (5/8") strip, placed it upon the tongue edge, and taped it in place with masking tape. The strip served as edges for the cover, and the balance as sides for the box (Figure 8–17). Tops and bottoms were made from 3 mm (1/8") thick mahogany panelling he had on hand. Ends and sides were cut to the desired lengths (without removing tape), and the box assembled using white glue and finishing nails. He used square corner joints, but mitred corners look better and hold well if joined with glue and finishing nails. Square corners were assured by equalizing the diagonals and holding the shape temporarily with tacked-on diagonal strips. He advised caution to avoid gluing top to bottom! An interspersed layer of thin plastic film

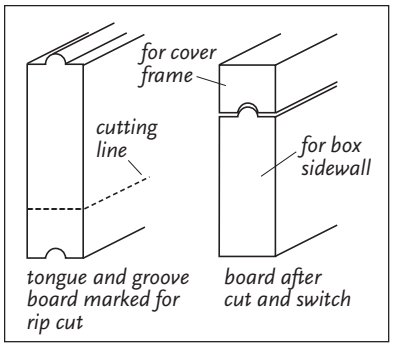


Figure 8–17. Insect box construction detail.

reduces this risk. For pinning bottoms he used 6 mm ($1/4$") thick cork sheeting, obtained from building supply stores. Plastic pinning foam may be less expensive. He finished off with two coats of polyurethane finish, and two pairs of brass hooks to secure the cover. Any defects or holes in the woodwork were reformed with plastic wood, and a bead of white glue sealed all interior joints.

Insect storage boxes may be stacked flat, but are easier to access if they are lined up on a shelf, resting on their front edges. An end label, made out with lead pencil, indicates the contents and can easily be erased and revised. If the boxes are also numbered and stored sequentially, it is easy to make an index for locating particular groups or species.

Pinning surfaces have evolved over time, from layers of cork covered with white paper, through various types of soft fibrous composition board, to several sorts of plastic foam now in use. These include 8 mm ($5/16$") thick polyethylene foam that has a rather open texture but holds pins well, and 9.5 mm ($3/8$") thick plastazote foam, that has a very fine texture and holds pins even better. Both are chemically similar, and in both the pinholes tend to disappear after pins are removed. While the plastazote costs 70% more than the polyethylene, its brighter whiteness, finer texture, and softer "feel" for pinning makes the extra cost worth considering. It readily accepts even No. 00 pins.

It is often possible, at considerable saving, to acquire used insect boxes or museum drawers that are being "retired" by collectors or museums. These may have deteriorated pinning surfaces that need to be upgraded. Foam sheets can accomplish this nicely, but check first to make sure that adding a layer inside the bottom will still leave adequate head room for your pins. Suppliers carry special glue that they recommend for holding the plastic foam sheets in place. Water-based household glues, such as Elmer's, are satisfactory, but cements based on volatile hydrocarbon solvents must be avoided. They can cause disastrous buckling, shrinkage, or other distortion of the foam sheets. "Arlene's Original Tacky Glue" (from dry goods or fabric

stores) has been reported as particularly satisfactory for installing pinning bottoms (McMahon 1990).

Collectors who acquire more and more species, especially of small and medium sized moths, are often frustrated by the difficulty of placing an additional species in proper taxonomic order in an already crowded drawer. It can involve moving large numbers of the present residents of the drawer, forcing overflow into the next drawer, with more large-scale moving, and so on. One approach is to place specimens so as to leave gaps for additional species that you have a reasonable expectation of acquiring in the future.

Unit trays are a more flexible (and expensive) solution. These are little topless cardboard boxes with pinning bottoms, sized so that rows of equal or mixed sizes will snugly fill the drawer. A single species is put into each unit tray, with the size of tray selected according to the species' size and the number of individuals you need to accommodate. Rearranging involves moving trays, not individual specimens. You save time and avoid damage. Each tray can be marked with the scientific name and catalog number of the species, typed on a gummed label placed inside the top edge. *Caution:* the dimensions of the unit trays differ just enough between the Cornell, California

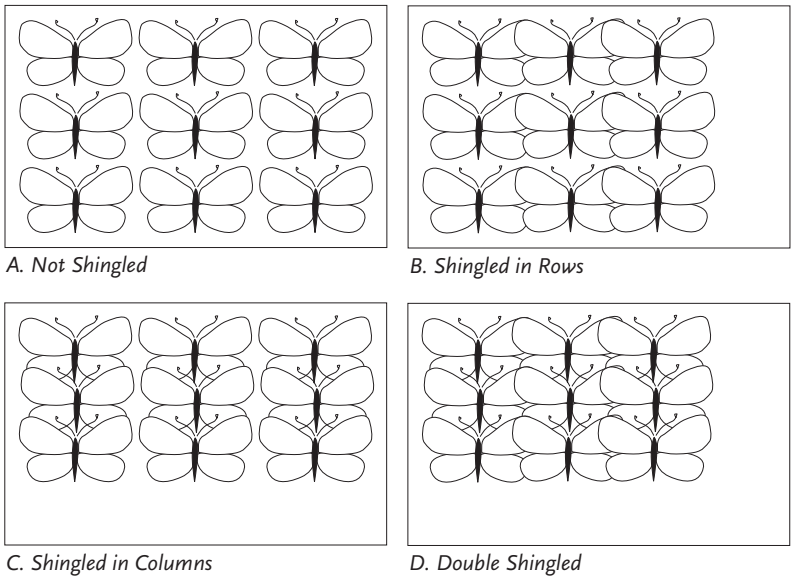


Figure 8–18. Shingling specimens.

Academy, and USNM systems so that the trays are not interchangeable. In addition, the height of the trays is such that they need to be used in drawers with hard bottoms and no pinning material.

Shingling is a means for storing increased numbers of specimens in limited space (Figure 8–18). If you are arranging in vertical columns, lean the pin obliquely towards you about 10° as you pin it into the box, so that the front of the body is higher than the rear. Subsequent specimens are pinned at the same angle, overlapping from the rear. If you are using horizontal rows, insert the pins tipped to the right, with the left wings of the second specimen overlying the right wings of the first. For double-shingling, insert the pins leaning both towards you and to the right, so that overlapping can be done in both directions at once.

The drawback of shingling is that access to individual specimens, except those at the end of a row or column, becomes quite difficult, and great care (or moving other specimens) is necessary to avoid damage. Data labels may be impossible to read. Shingling is perhaps best reserved for specimens that have been completely curated and inventoried and will seldom need to be taken out for examination.

Makeshift boxes are a trap for most beginning collectors and should be avoided whenever possible. The traditional shoe or cigar box paved with corrugated cardboard does indeed hold pinned specimens securely so that they do not jostle each other, but such boxes are no defense against foraging museum pests. Repellents that would ordinarily be useful dissipate so rapidly as to offer little protection. Early investment in a few proper insect boxes can prevent major disappointments. Perhaps the only way to maintain a collection in makeshifts is to seal the boxes in plastic bags and store them in a freezer—if you can negotiate enough space!

7. Protecting Stored Specimens

Every niche on this planet is capable of being exploited by one organism or another, and the lepidopterist's collection is no exception. The main threats to preserving a collection are museum pests, molds, humidity, light and direct physical damage. Defenses can be physical, chemical, thermal and combinations thereof.

The most actively intrusive museum pests are the unobtrusive dermestid beetles known also as carpet beetles and buffalo bugs (the

larvae of carpet beetles). While several species are involved, they all operate in similar ways, and accurate distinctions are unnecessary. An excellent atlas for identification is available, however (Kingsolver 1988). The larvae devour the dried specimens, quickly reducing them to piles of dust, and given time, they will leave nothing but pin and label. The first sign of damage may be a separated head or abdomen or a drooping wing on a previously intact specimen, or a pile of dust obscuring a label. Cast larval skins may float around in the eddy currents created when you take the top off a drawer. Eggs are laid on specimens on the spreading board, or loosely built boxes are invaded by wandering larvae. Specimens acquired from other collectors may already be infested, even though no damage is yet visible. The creatures seem to have the knack of selecting your most favored specimens and saving the more ordinary ones for later! Infestation by this group of insects is an almost universal threat.

Psocids, or booklice, are so small as to be barely noticeable and feed as nymphs and adults on molds and dead insects. Their appetites and activity are greatest at moderate humidity and elevated temperatures. Their damage is more subtle, consisting at first of lost fringes, or lost patches of scales, with piles of dust so small as to be easily overlooked. Adults, with bigger mandibles, do greater damage, and total loss can occur if the problem is not treated (Welling-M. 1983).

While cockroaches can dine happily on specimens, their size makes it simple to exclude them by the use of tight boxes and drawers. If they are an annoyance in your workspace, conventional methods for dealing with household infestation should suffice.

Quite common are the thysanurans or silverfish. These are better classed as a nuisance, rather than a pest. They eat starch and wood pulp, but not rag paper, and hence are attracted to pastes in books or wallpaper and the starch sizing in some papers. Damage to collections is more likely to occur to libraries and box labels, rather than to the specimens themselves.

If ants invade the area where you store or work with your collection, they can be extremely damaging scavengers, especially to material still on the spreading boards.

The first line of defense against marauding arthropods is physical. The importance of tight box and drawer construction has already been

noted, as has been the use of a tightly crafted screened cage to exclude ants from spreading boards. Wood (1971) recommended application of a residual insecticide (not named) to all the side surfaces of the spreading boards. Because these are the surfaces by which boards are commonly handled, the possibility of finger-to-mouth transfer of the chemical agent is worrisome, and this cannot be recommended. The time-honored method of setting each leg of a tropical work table in a bowl of kerosene to exclude ants still works. "Combat," with a chemical name longer than a battalion of ants, is an effective product for eliminating indoor ants in your home (American Cyanamid; a commercial preparation of hydramethylnon or [tetrahydro-5,5-dimethyl-2 (1H)- pyrimidinone (3-[4-(trifluoromethyl) phenyl]-1-(2-[4-(trifluoromethyl) phenyl]ethenyl)-2-pro-penylidene) hydrazone]).

Physical barriers seldom seem to provide absolute protection against dermestids and psocids, yet many failures can be related to sloppy practices in handling a collection. Since a great many infestations start on the spreading board, specimens removed from the boards need isolation and freeze-treatment in a "quarantine box" before they are merged with other specimens. The same is true of specimens acquired from other people. These might be already infested before they reach you. Leaving specimens outside closed boxes when they are not actually being worked with allows access by pests. Simple poor housekeeping—allowing dead flies and wasps to accumulate on inside windowsills or in light fixtures—helps to keep a domestic dermestid population thriving. Pheromone traps can be used to monitor wandering dermestids in your work area. It is important not to use traps aimed at the dermestids that infest stored food; these are a different group from the museum pests (Alpert & Alpert 1988). Sticky traps baited with a dead insect are also useful monitors.

Collectors have long used chemical agents as the next line of defense. Each agent has its limitations, and each has its particular toxicity to humans. As a lepidopterist, your work space is customarily within arm's reach of your collection, so you (and perhaps your family) are at risk of long-term subliminal exposure to any chemicals that you use. These agents are included here, and their manner of use described, because they are in widespread use and are readily available. It is important to be aware of what they cannot and can do

for you, but more significantly, what they can do *to* you. Before deciding to use or to continue to use any of these chemicals, you should *read the information provided on each in Chapter 10, Hazards.* Since the susceptibility to ill effects from these toxins can vary greatly from one individual to another (especially in relation to preexisting health conditions), you should seek the advice of your physician.

Naphthalene, as crystals or "mothballs," has long been used as a repellant and is fairly long lasting (up to a year). This chemical does not kill larvae in an already infested specimen, so newly introduced specimens may carry a risk; the larvae they bear can go on to mature and reproduce. Eggs are not killed.

The crystals are placed in a small screen-covered fumigant box, in a receptacle made from cardboard tubing glued into the corner of a drawer, or in a small unit tray if you use that system. A "pin ball" made by plunging the heated head of a common pin into a mothball is another way to apply naphthalene. A couple of these mothballs are then pinned into each specimen box. Note that as the naphthlene gradually evaporates, the remainder of the moth ball can separate from the pin, leaving a potentially damaging missile loose in the box. Never include pinned mothballs when shipping specimens.

Paradichlorobenzene (PDB) is repellant and lethal to larvae and adults. It is placed in the boxes and drawers using the same methods as for naphthalene (except that it cannot be mounted on pins), but because of higher volatility it needs replenishing every few months. Dermestid eggs are not killed by PDB, and even larvae are sometimes resistant. Eggs hatch in 14 days or less, depending on the temperature, so three weeks of strong exposure is a bare minimum.

When used in high concentrations or for long periods, both these agents sometimes crystallize on the bodies of specimens, producing deleterious effects. Growing crystals disrupt the hair and scales, damaging tuft patterns that are of diagnostic significance. "Greasing"—escape of body fats to the surface, where they mat wing and body scales and obscure colors—seems to be accelerated. Pins sometimes corrode (except for stainless steel).

A useful insecticide is Vapona, trade name for dimethyldichlorovinyl phosphate (DDVP or dichlorvos, also sold under the names Herkol, Nuvan, No-pest-strip and Vaponite). It is marketed as an

impregnated plastic strip that can be cut into small squares and pinned into the box or drawer that is to be protected. A 1.3 cm ($^1/_2$") square is usually enough for a Schmitt box. Yearly renewal seems to be sufficient, but since the plastic strip does not change in appearance, and since cutting the strip speeds up the diffusion rate, it is hard to know when the fumigant is exhausted. It does not kill eggs. If the strip is in contact with the bottom or side of the box, it can make unsightly stains. Too liberal use results in an oily film on the inside of glass drawer tops, and potentially on the specimens as well. The agent is most effective at temperatures of 15–18°C (60–66°F). At higher temperature and humidity it starts to break down and become corrosive; at lower temperature effectiveness diminishes (Williams & Walsh 1989a). Vapona can degrade some lacquers and accelerates corrosion in steel. Pins can suffer. Its effects on museum specimens are not completely known, but it is oil and fat soluble and its breakdown products are strongly acidic. Museum specimens exposed to insecticidal levels of DDVP, after removal, are readily consumed by dermestids, albeit somewhat more slowly (Williams & Walsh 1989b).

Some of the plastic foam pinning surfaces react adversely to the above chemicals, developing shrinkage from vapors or holes from contact with dropped liquid or crystals. Glues used to hold the pinning bottoms in place may also be affected. The advice of the suppliers of these pinning surfaces should be respected. Aluminum also reacts adversely: perforated foil, used to keep Vapona from direct contact with pinning foam, can be reduced to powder.

Welling-M. (1983) described carbon disulfide fumigation as the most reliable way to rid a collection of psocids. However, his more detailed explanation of the procedure (pers. comm.) indicated that this was a major undertaking, potentially very hazardous for the operator, and it will not be described here. In addition he discovered that polyethylene foam pinning bottoms shrink massively on exposure to carbon disulfide, bunching all the specimens together in a hopelessly damaged mass.

Because of the expanding Occupational Safety and Health Administration (OSHA) regulations regarding exposure to chemicals in the workplace, and the justifiable concern of curators regarding chronic exposures, many institutional collection managers have largely abandoned chemicals in favor of a "spot-check and freeze" regime

(D.G. Furth, pers. comm.). All incoming material, and any drawers showing signs of active infestation are frozen at –20 to –25°C (–4 to –13°F) for two or three days. This is adequate to kill all stages of development and is a technique usable at home (advance consultation with local management is advised!). The freezer being used should be checked with a maximum-minimum thermometer to determine its temperature range, and more time used for temperatures higher than ideal. If the drawers or boxes are wrapped in plastic bags before freezing and left enclosed until they return to room temperature, there is no problem of relaxation from condensing moisture.

On finding a single infested specimen in a box or drawer, some collectors treat that specimen with a drop or two of alcohol. While this is effective, it does not address the possibility that other specimens may be infested, with damage not yet apparent. Freeze-treating the entire drawer is preferable.

For a detailed account of overall protection of your premises, consult Alpert and Alpert (1988) for an approach designed for museums but with many details pertinent at home. For example, they note that mouse-bait not eaten by mice is a breeding site for museum pests, and that mice killed by mouse-bait are breeding sites for museum pests!

Problems with molds relate directly to humidity and temperature, the first being most important. Molds cannot grow on material with a water content below 7%. Lepidopterists are most active in the summer, and this may make it tempting to have your workroom in a cool cellar. This can be a bad choice, because the relative humidity will be higher there and growth of mildew is often rampant. Spraying walls, floor and ceiling with Captan will reduce but not eliminate the problem (active agent phthalimide—a fungicide available in numerous preparations from hardware stores; see Hazards, Chapter 10). "No More Mildew" is a similarly useful product (active agent chlorothalonil—National Allergy Supply, see Appendix L). Working in as dry an area as possible, leaving airspace behind any banks of storage cabinets, opening and leaving boxes open as little as possible during warm humid weather, and protecting unspread material in storage (as described in Section 4 of this chapter), are effective ways to avoid problems from molds.

If a drawer or box does become affected by molds, treatment with

PDB for several weeks can stop the spread. Freezing is not effective for killing or eliminating molds. A specimen invaded by mold is usually beyond salvage, but superficial growth can allegedly be destroyed by painting with liquid phenol (Cribb 1988). This substance is crystalline at room temperature, and it must be liquefied by placing its container in warm water prior to use. *Caution: phenol can be absorbed through human skin and is toxic.* Exposure to fumes from a cloth pad soaked in formaldehyde has been advocated to kill molds, but any specimen so exposed becomes permanently hardened and is no longer able to be relaxed and respread. In addition, the fumes are highly irritating and often debilitating to humans.

Silica gel desiccant, available in packets or bulk from suppliers, helps to reduce humidity within a drawer. This agent must be periodically reactivated by drying in a hot oven. A change in color from blue to pink signals a need to reactivate it. Because of its limited capacity to absorb water, however, it is not very useful in drawers or boxes that are not completely sealed or are opened frequently.

Light is an insidious enemy of stored specimens. Spread specimens left exposed for public admiration gradually become less admirable as pigment colors fade, and restoration is not possible except by replacing the specimens every few years. Diffraction colors, as in morphos or coppers, are an exception, but pigment colors present on such species fade nevertheless, and the beauty of the specimen is degraded. Prophylaxis is simple—store everything in the dark, with closed cabinets for all glass-topped drawers and cases.

The risk of physical damage is ever present, as mentioned so often above. Dried specimens are brittle and have no flexibility—any touch or sharp jarring can cause damage. Bumping a leg, wing, abdomen or antenna of one specimen against another can cause loss of a part. This risk is enhanced when you are working with shingled specimens. A dangling bracelet or sleeve cuff can plow up a whole row of beauties. A misdirected sneeze is a small tornado.

Unanticipated extraneous violence can be eye-opening—if you are having your house reshingled, specimens boxed on shelves against an outside wall can lose abdomens from the shock of the carpenter's hammer! If you live in an earthquake-prone area and store boxes booklike on a shelf, a restraining cord or wire across the front of the shelf will keep the boxes from being jiggled to the floor by a temblor.

If you stack them one upon another, more complete enclosure by a door or a net is advisable.

Physical protection of specimens being shipped by mail is covered in Chapter 11.

8. Remedies

Certain problems that develop with spread specimens can be corrected, to a degree, but in most instances anticipation and avoidance is a better strategy.

Sagging or folding up of wings long after spreading can result from storage in chronic high humidity and is best managed as outlined above. However, it readily occurs when specimens are removed from the board before drying is complete. In addition, spreading an incompletely relaxed specimen commonly results in recidivism, in the same way that a warped door, forced to close with pressure springs back out of shape when the restraining latch is released. Repeat relaxation and spreading is sometimes successful, but prevention is better.

Occasionally spot relaxing is useful while a specimen is still on the spreading board, to reposition a wing that has slipped down, or an antenna that has popped up out of the plane of the wings. Using a small hypodermic syringe and a short small-bore needle, place a drop of 95% alcohol at the base of the antenna, taking care not to bump and break it. The alcohol soaks in and acts as a wetting agent. Then similarly place a drop of water in the same place; it will quickly soak into the tissues. Let the specimen "marinate" for several minutes, then reposition the antenna by gently pushing it with a setting pin at the base of the antenna (the portion of the shaft that has not been moistened will still be hard and brittle). Once the antenna is in position, let the specimen dry for several hours to ensure evaporation of the alcohol-water mixture. This same method may be applied at the base of a wing to soften the tissues. For large specimens, several drops of alcohol and water may be necessary.

Greasing is a problem with many heavy-bodied species, especially females and species whose larvae are borers. A taxidermic approach is to slit the underside of the abdomen lengthwise, prior to spreading, scrape out the insides, and stuff it with cotton. This is tedious and leaves a damaged specimen, usually without internal genitalia. Some

think that it is the only approach for giant skippers and cossids.

Organic solvents can be used effectively to remove grease. The agent of choice should ideally be not flammable and not toxic (see Chapter 10 for a discussion of these possibilities). Submerge the specimen to be degreased in a few ounces of the solvent in a wide-mouthed jar with a tight cover, and leave it for several days, varying the time as experience teaches with regard to various degrees of greasing. Then remove it, drain excess solvent by tipping the specimen and touching wing tips and abdomen to the inside of the neck of the jar, or to a piece of blotting paper, and allow to dry by evaporation in a well ventilated place, pinning it into a cork in a wood block. Another method of draining is to place the specimen so that the entire under surface is in contact with dry plaster of Paris or fuller's earth. After an hour of contact, dust off any adhering powder with a camel hair brush (Covell 1976a). Matted body hairs can be carefully fluffed up with the brush (Heitzman & Heitzman 1991). Specimens so treated may come out as good as new, with the exception of iridescent (diffraction) colors, which may remain darkened. Some specimens regrease after many weeks or months, indicating the need for longer treatment. Degreasing is best done in warm weather, when evaporation is faster and good ventilation is easy to maintain. Because many solvents affect plastics adversely, use only a glass jar with metal lid, and avoid plastic pinning surfaces. Some collectors remove the abdomen from species likely to become greasy, soak it in solvent for an extended period, then glue it back on (see below) after complete evaporation of the solvent.

Pins may corrode after immersion in trichloroethylene for degreasing, presumably from dissolution of the protective finish, exposing the bare steel. Use of stainless steel pins can avoid this problem

Some labels do not tolerate some degreasing solvents. This should be managed by removing the pin label, or by specifically testing the label-solvent combination in advance, using a spare label. For example, a solvent may cause laser-printed pigment to separate from the paper, or make writing ink bleed into it. On the other hand, letterpress printing and some xerographic printing may remain stable. With so many possible combinations of process, paper, and solvent, a preliminary trial is the safest course (Winter 1994). This is

particularly true if printing methods of two sorts are used on the same label.

With a steady hand and proper adhesives, broken parts can be reattached. Because of past experience with use of fraudulent "spare parts" to upgrade a specimen or even to fake a gyandromorph, some believe that repairs should never be made—that any detached small parts (antennae, legs, abdomens) should be placed in a small gelatin capsule pierced by the specimen pin, and wings in an envelope similarly pierced.

If it is your choice to restore a detached part, first have a reliable adhesive available. Ferris (1973) advised against the use of household glues, such as Elmer's Glue-All, since it can form a visible white blob and also injure delicate structures. Clear shellac can be used undiluted to restore detached legs, wings and antennae. If you wish to repair a wing tear, a 1:1 dilution with wood alcohol (methanol) is preferable. The shellac should be applied to both the surfaces to be joined (he cautioned that shellac may stain). He recommended that when legs or antennae were being replaced, the specimen should be placed on a spreading board and the loose part stabilized with fine pins until the adhesive dries. He found the "slow" varieties of cyanoacrylate cements (akin to Crazy Glue) were very useful. On the other hand, J. M. Johnson (pers. comm.) stated that one part Elmer's Glue-All plus six to eight parts isopropyl rubbing alcohol made a fine cement leaving no shine or visible residue. He applied it with a pigeon wing feather from which all but the last one or two vanes had been removed from the shaft. The specimen to be repaired was supported on a spreading board on which a strip of mylar tracing paper had been laid, shiny side up, to prevent gluing the specimen to the board. Drying was rapid.

Clear fingernail polish is sometimes used for repairs. It sets enough to hold the parts in place after a few seconds, and dries hard in a few minutes (J. Taylor, pers. comm.). The bonding can be dissolved with acetone or ethyl acetate.

Supply houses carry a cement especially selected for repairing Lepidoptera.

Kemp (1991) noted a safe and delicate way to handle a detached butterfly antenna during repair. Using a water-filled syringe and hypodermic needle with a droplet of water poised at its tip, he touched

the water to the antenna knob and thereby picked it up. After applying a minute drop of cement to the attached side, he held the broken end of the antenna against the cement until it had set, then gently moved the needle away. The cement was stronger than the surface tension of the water, and the antenna remained in place.

9. Special Displays

The methods of pinning and storing specimens in a collection being built with scientific value in mind have already been outlined. A goal of some collectors, however, is to have specimens available primarily for admiration and enjoyment of their colors and patterns, or for convenient demonstration for educational purposes. Several display methods meet these needs. When visible displays are your choice, consider using display case covers fitted with special plastic or glass that screens out UV light.

Riker mounts are shallow glass-topped frames filled with cotton padding or similar substances, and usually made of cardboard with a black embossed paper finish. Spread specimens removed from their pins are arranged on the cotton; the glassed cover is replaced and held in place with the pins provided. Advantages of these mounts are ease of viewing, ease of passing around from person to person for close examination, and the overall attractiveness of the display. Drawbacks are numerous, however. Rearranging specimens often leaves depressions or staining where bodies were previously placed. Legs become snagged in the padding. Data and identification labels, if not done meticulously, can look tacky. Undersides can be viewed only by having a second specimen, spread upside down. Greasing soils the glass and padding as well as the specimen. Dermestids can be excluded only by tightly taping the edges (for certain protection, the mounts should be taped and freeze-treated as soon as they are set up). And removal of pins from the specimens can be both tricky and damaging.

There are several approaches to depinning. When previously dried, relaxed specimens are spread, the bodies usually do not stick to the pins and removal is simple. The drying body fluids of specimens spread while fresh tend to cement the thorax to the pin. If the specimen is large, it is often possible to grasp the top end of the pin with forceps or small pliers and rotate it, using your thumb and forefinger

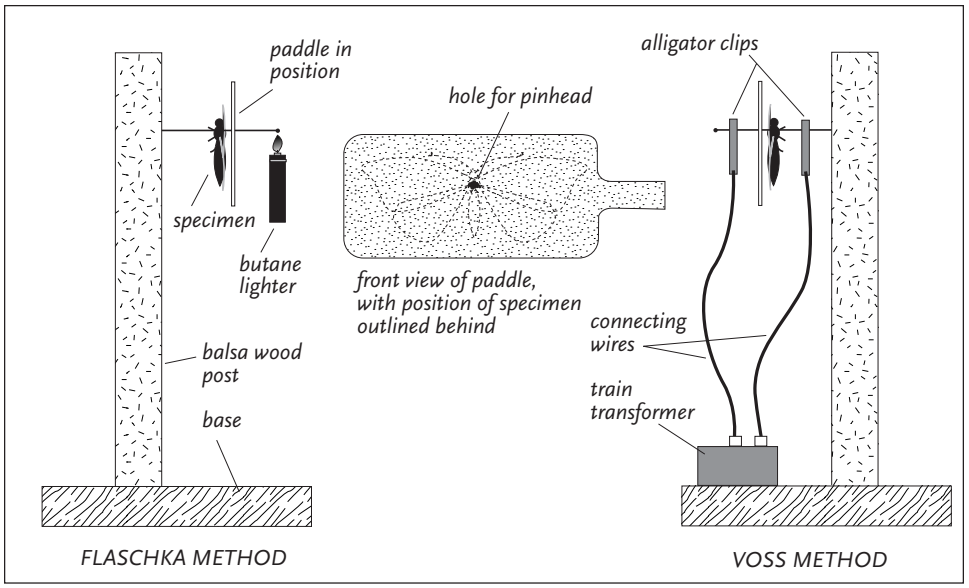


Figure 8–19. Depinning.

against the underside of the body to keep that from turning. Once the pin turns freely, pushing with forceps on the top of the thorax will slide the specimen down off the pin. It can then be placed as desired in the Riker mount. Using this same approach on a specimen with a small slender body easily results in removal of the abdomen, a wing, or even a break across the thorax between the fore and hind wings (now see section on repairs!).

Sticking to the pin can be averted by lubricating it lightly with oil or silicone grease, and if sticking has occurred, a depinning remedy was offered by Flaschka (1974). This calls for a vertical block of balsa wood supported by a firm base (Figure 8–19). The specimen, with pin horizontal, is pinned securely into the balsa. A small paddle made of cigar-box wood and bearing a central hole slightly larger than the pin head is placed so that it hangs on the pin, plane of paddle parallel to plane of wings. A flame from a butane lighter is applied carefully to the head end of the pin. As the pin heats up and the surrounding tissues loosen or char slightly, the paddle is used to push the specimen along the pin towards its point.

A refinement was offered by Voss (1975). Using a low-voltage current from an electric train transformer with wires connected by alligator clips to the head and base of the pin, he heated the pin to a

dull glow. It could then be removed with ease, and the use of an open flame was avoided. Pins so heated lose their temper and should be discarded.

A probably obsolete but historically interesting device for displaying single specimens was the Denton mount. This was a cast plaster of Paris block with a smoothly contoured depression in the upper surface, on which the specimen was placed. A precisely cut glass rectangle was then placed on top and the edges sealed permanently with tape. The data were written on a label on the back of the block. The reverse of the specimen was not visible, but the blocks could be passed about safely from hand to hand for inspection and admiration. Denton-mounted specimens have remained in excellent condition for over one hundred years.

Another type of permanent individual mount, the glass mount, can be made readily by anyone handy with a glass cutter or with Plexiglas. This mount makes both surfaces of the specimen visible and is particularly useful for passing around in classrooms (Figure

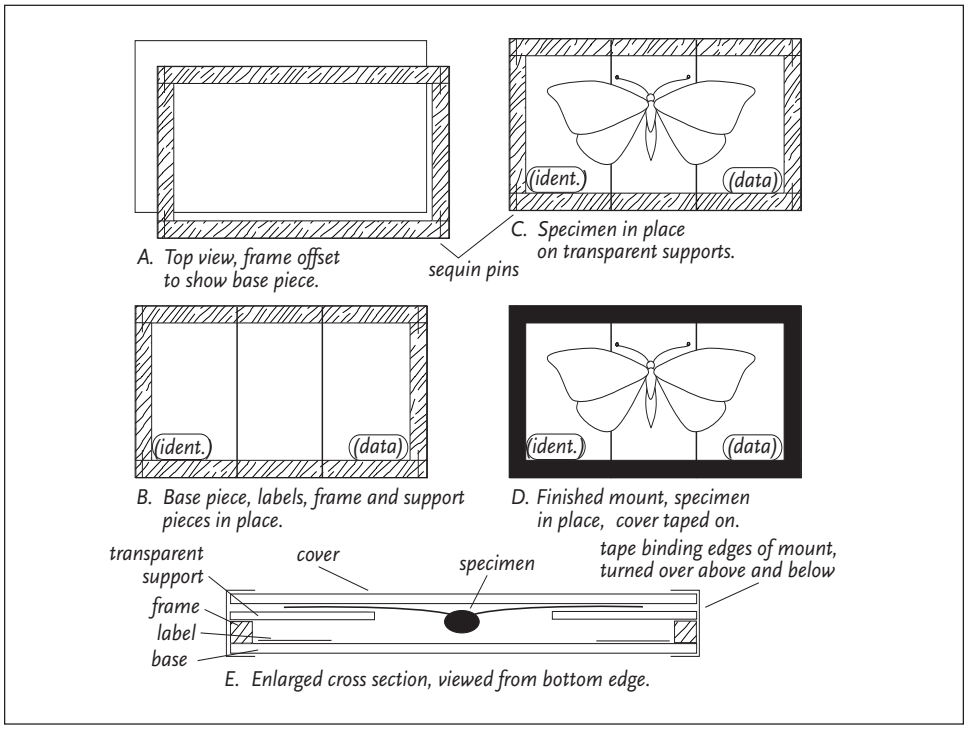


Figure 8–20. Glass mount.

8–20, A–E).

1. From window glass ("single thickness") or 3 mm ($1/8$") thick Plexiglas make a base rectangle about $1\,1/2$ times the spread dimensions of the butterfly or moth (make two of these, the second to serve as a cover).
2. Cut four 6 mm ($1/4$") square sticks of balsa wood to form a frame lying upon and flush with the perimeter of the base (A). Push a sequin pin into each corner to hold the shape of the frame. A higher frame may be necessary to accommodate a very thick-bodied specimen.
3. Stick a data and an identity label to the lower inside corners of the base before assembling.
4. Make two equal transparent rectangles the same height as the base but less than half the width, and lay these on the frame so as to leave a gap down the center (B, E).
5. Place a thoroughly dry, spread, depinned specimen on these supports, with the body in the center of the gap, and cover with a top piece of glass or Plexiglas the same size as the bottom (C, E).
6. When the edges are all perfectly aligned, seal the edges with tape, lapped over a bit onto the top and the bottom (D). Make vertical cuts at the corners before folding down.

If the specimens were free of infestation when sealed, they remain permanently protected. Routinely freezing each completed preparation is a good safety measure. Store the mounts in the dark when not in use.

10. Preserving Immature Stages

The foregoing material has been directed at the preparation and preservation of adult Lepidoptera. It is often desirable to preserve examples of immature stages, both for your own interest and for possible taxonomic evaluation. The goal is to maintain size, shape (with all surface details), and color, the last being frequently impossible.

One of the oldest techniques, now seldom used, involves inflating and drying larvae. *The Butterfly Book* (Holland 1898) describes the procedure in detail and it will not be repeated here. It produces specimens of limited scientific value, since important features of the

ninth and tenth abdominal segments are usually destroyed.

A more recently developed technique, freeze-drying and vacuum dehydration of caterpillars, is excellent but requires rather sophisticated and expensive equipment. It is clearly described in an article by Dominick (1972).

Pickling is the commonly used method of preservation and is well suited for use by amateurs as well as professionals (Godfrey 1982, Stehr 1987). Ethyl or isopropyl (rubbing) alcohol, 70–80%, is used as a preservative. Acetic acid stops enzyme action, avoids darkening, and helps to retain flexibility. These agents are mixed in the ratio of nine parts alcohol to one part glacial acetic acid. Distension can be maintained by killing the larva in water just off the boil (1/2 to 1 1/2 minutes, depending on size of larva) or by injecting anally with the acid-alcohol mixture (larger larvae). *Be sure to wear protective goggles when injecting,* for protection from accidentally scattered droplets. Select larvae that are not newly molted (these do not distend well) and not in the premolt stage (the old skin may separate during the preservation process). Larvae of different relative sizes should be chosen from different instars when you are working with known ova. Using the largest larvae at each stage may take all females, leaving you with only males to rear through. Injected larvae should be left 24 hours in the acid-alcohol solution. Boiled larvae should be placed temporarily in 25% alcohol, then 50% alcohol, for two hours at each level. Thereafter larvae treated in either fashion should be transferred to 70–80% alcohol for permanent storage in individual glass vials with screw-on Poly-Seal caps (they are leak and evaporation proof).

Glass vials sealed with neoprene stoppers are still in use. For these, to bleed off excess pressure while inserting a stopper, place a No. 1 insect pin in the neck of the bottle until the stopper is well seated, then remove the pin. Vials should be inspected at regular intervals, so that declining fluid levels can be "topped up" to prevent drying out. Record the killing and preservation techniques on the vial label, for replenishing the preservative and for assessing the efficacy of the techniques for long-term storage.

If you are rearing a single larva, especially an unknown, you will not want to sacrifice it. Instead, take good color photographs (discussed in Chapter 2) at each stage, make descriptive notes, and save cast skins and head capsules in alcohol.

Unhatched, or hatched, uneaten egg shells can be preserved by cementing them to a rectangle of card stock and mounting on an insect pin with a data label. Dead pupae or shed pupal shells can be dried, pinned and labeled in the same manner as adults. Pupal shells may need a dab of glue to keep them from sliding down the pin. Small pupal shells can be placed on the same pin as the spread adult, beneath it and above the data labels. Larger specimens can be glued to a small strip of index card and similarly pinned. Live pupae are dealt with in the same manner as larvae; wait two days after pupa formation before processing. A pupa that has produced parasitoids can be pin-mounted. Adult parasitoids from any stage should be preserved.

Because of the impossibility of preserving many of the colors during pickling, the value of taking color-accurate photographs of all larval stages cannot be overemphasized.

Careful labeling is essential to assure the correlation of the photographs, notes, and exuviae with each other. Every vial and pinned specimen should be individually labeled and should include the source locality of the original material (ovipositing female, wild-collected eggs or larvae); substrate on which wild ova or larvae were found; whether it was found feeding on a particular foodplant, or, instead, fed upon a proffered foodplant; instar at which larva was preserved, if known; age since oviposition or hatching, if known; date of preservation; name of rearer; rearing batch number.

Collections of immatures are not necessarily things of beauty, but if properly prepared and well cared for, the specimens will be of permanent scientific value.

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Chapter 9.

PREPARING GENITALIA

Genitalia of Lepidoptera are almost as important to lepidopterists as they are to the Lepidoptera themselves, as in many groups they give the clear and sometimes only sure distinction between one species and another.

The following procedure for making permanent slide-mounted genitalia preparations has been adapted from a series of articles by L. Paul Grey (1991–92) written for the amateur, as he put it, "as I wish such guidance had been offered to me in the distant past." This is the procedure arrived at by one skilled amateur. There are many parallels and variations to his approach, and further refinements for making genital preparations may be found in Clarke (1941), Clench and Miller (1976), and Hardwick (1950).

Minimum needs

The chapter is about as minimum as can be —no shortcuts.

The goal in making genital preparations is to expose and remove from the abdomen the distinctive sclerotized and related membranous genital structures and to prepare them for visual examination and photographic documentation.

The approach described is effective for making preparations of medium and large moths and butterflies, and useful for practicing and learning to deal with genitalia. Before working with smaller species, especially with "micro" moths, where the need for dissection is greatest, you will need more delicate instruments and techniques, best learned from the references above. Dissections for the purpose of identification should always be done on good quality specimens, but specimens in poor wing condition are satisfactory for practice work.

1. Preliminaries and Equipment

Acquiring and assembling specific equipment and record materials should precede your first efforts at preparing genitalia. The following are the essentials:

- Catalog notebook: this is a preparer's personal lifetime record, with each page numbered consecutively from number one

onward, thereby providing a catalog number for the specimen being dissected, and containing all the information entered on the specimen's data label, the date of dissection, the genus, species, author, and date (if known), and whether the preparation is preserved on a slide, in a vial, or in a capsule (also note whether it is stored on the same pin as the specimen it came from). If a genital preparation is discarded, transferred elsewhere, or otherwise disposed of, those facts should be recorded in this catalog. A checklist number may be included as a temporary convenience, but remember that checklist numbers tend to be replaced or changed every few decades, while the catalog and the labels generated in conjunction with it bear permanent records.

- Before starting a dissection, three labels should be prepared: a pin label for the specimen whose abdomen has been purloined, a slide label (or labels) for the finished preparation, and a small, numbered label to accompany the preparation from step to step. Labels are made in advance on a copier or a laser printer, in 4-point type, with spaces left for penning in data pertinent to the specimen being dissected.

Labels

1. The pin label is about 10 x 20 mm ($7/16$ x $13/16$"), on paper of archival quality (explained in Chapter 8), and includes space to write in the preparer's personal catalog number and the dissection date. The sex symbol of the specimen should also be penned in. An example follows (A, in Figure 9–1).
2. The slide label is on white gummed paper available in print shops, 25 x 25 mm (about 1" square) and preprinted to simplify penning in the preparer's catalog number, dissection date, collection data and name of collector, and genus, species, author, and year, when known (B, in Figure 9–1). This label is applied to the left-hand end of the slide. If your labels seem too crowded with all this information, use a second label at the right end of the slide for genus, species, author and year.
3. The small processing label, completed with a dark lead pencil, contains the preparer's catalog number and must accompany the preparation until its permanent labels are in place (C, in

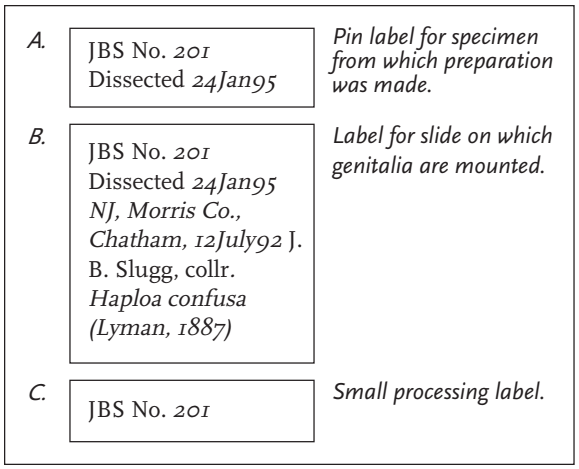

Figure 9-1. Labels for genitalia preparations. Plain type is preprinted. Information in italics is penned in by hand.

Figure 9–1).

• Magnification is necessary, ideally from a stereoscopic dissecting microscope, but it can sometimes be accomplished with an ordinary monocular scope (in this scope the motions you see are the reverse of what you are doing!), or with a 4x or stronger jeweler's loupe.

• "Syracuse watch glasses," flat bottomed round glass dishes with a shallow cylindrical well are needed for dissections (Carolina Biological Supply, Appendix L).

- Straight and L-shaped dissecting needles can readily be homemade, but long-nosed very thin jeweler's forceps are a necessity, straight for removing preparations from KOH, and curved or 45° angled for manipulation (or finer, higher quality forceps: Fine Science Tools, Appendix L).
- For scissors, "iris scissors" are superior to small embroidery scissors. Or try to get an unused No. 11 surgical scalpel blade from your physician. A home made "micro-scalpel" can be even better:

1. With a hammer, strike the tip of a minuten lying on a heavy piece of iron (another hammer, wrecking bar, or vise anvil) to flatten it into a tiny cutting edge. Several tries, with different needles, may be necessary to produce an effective blade.
2. The needle is then cemented into the end of a tapered wooden matchstick for a handle. Or use a bamboo shish kebab skewer—even the tapered end can be neatly split with a razor blade to make a cleft for the scalpel. A fragment broken from the edge of a razor blade can be similarly mounted.

- Small brushes are essential (hobby or artist supply shops); synthetics in sizes 00 to 000000 are adequate for starters. For more delicate work with smaller preparations the more expen-

sive artist's sable brushes are ideal. If rinsed often they will last a long time; sables should not be used until the preparation has been washed free of KOH. Brushes can be trimmed to various lengths and angles for different operations.

- Potassium hydroxide (KOH) pellets. This chemical is obtainable from some supply houses, or from friendly science teachers, lab instructors, or researchers. *Caution: this is a corrosive chemical, damaging to eyes and skin.*
- Chlorazole black E, for staining (Sigma Chemical, Appendix L).

2. Genital Description and Terminology

The structure of the genitalia of Lepidoptera, in most groups, shows striking individuality on a species-by-species basis, such that these structures are a major part of the description and definition of the species. In some groups, however, such as buck moths *(Hemileuca)* and fritillaries *(Speyeria)*, the interspecific differences can be so minor as to confuse rather than enlighten. Study of mitochondrial DNA may be the ultimate means of distinction. In addition, there may be intraspecific variation, exemplified by asymmetry between the two male claspers, or seasonal variation, such that summer brood genital structure differs from that of the spring brood (some *Plagodis* geometrids). Here, rearing successive broods often can provide clarification.

Figure 4–4 depicts the principle structures of Lepidoptera genitalia. Further features of the genitalia and their modifications and functions are detailed by Klots (1956).

3. Procedure

The following describes the sequential steps for preparing genitalia of Lepidoptera:

1. Remove the entire abdomen from the specimen. Rough handling may remove the hind wings along with the abdomen. Use the fine, sharp-pointed scalpel. Apply pressure downward with the point of the scalpel at the junction of thorax and abdomen, against upward pressure from beneath the abdomen with a spoon-shaped dissecting tool. The best way to break is up. Forceps alone also work well.
2. First bath is in 10% (by weight) potassium hydroxide (KOH) in

distilled water. The purpose is to saponify the fats in the abdomen so that they can be washed out. *This solution is caustic and can eat holes in clothing and skin, creating painful, slow-healing ulcers.* Any such caustic burns should immediately be flushed copiously with running water, and may need medical attention. Eye burns require immediate flushing with water, and are a medical emergency. A KOH bath can be used many times before it is necessary to replace it, but it breaks down in light, and over time. Make up only what you will use in a few weeks, and store in a dark place.

3. As abdomens are processed they must be accompanied through the KOH bath and all other baths by the processing label described previously, so that identity of the specimen is never in doubt. As an alternative, the processing label can be stuck to the underside of a piece of plastic tape and transferred to the outside of each successive piece of glassware used for the specimen (as described in Chapter 6, Figure 6–12).
   As you make your first tries, work a single preparation through from beginning to end before trying to manage several at once.
4. Dip the separated abdomen in rubbing alcohol or a weak solution of detergent to speed wetting, place in a plain-necked glass vial, and fill $^{1}/_{2}$–$^{2}/_{3}$ to top with KOH solution, using a dropper. Stand each vial in a wood block with multiple holes (similar to a block for holding insect pins) to prevent spilling and to simplify grasping a selected vial. Soaking overnight at room temperature is usually adequate, but heavily scaled noctuids may require a longer time or use of stronger KOH solution: 12% for males and 14–15% for females. Curiously, many gelechioids require ±15 hours in 20% KOH. *Do not heat a vial containing KOH over a flame or other heat source.* It boils quickly, at low temperatures, with explosive splattering and risk of serious eye injury.
5. Cleaning is carried out in successive changes of water, usually two or three. The goal is to express through the thoracic end of the abdomen the fat and other material saponified and softened by the KOH, working from behind forward with external pressure from an L-shaped dissecting needle. Also, all the abdominal vestiture must be brushed off, so that internal

structures can be viewed through the now transparent abdominal wall. Start the brushing after the washing, or the KOH residue will dissolve the hairs of your brush. Female genitalia and those of very small moths may be damaged by cleaning efforts using a rigid needle tool. Instead use two brushes, one applied flat to stabilize the pelt, and the other, with the bristles cut to short stubs, to tamp through the first. In butterflies, where the abdominal segments are often heavily sclerotized, bleaching is necessary to attain transparency. Place the cleaned butterfly abdomen in a dilute solution of chlorine bleach (not over 20%). When sufficiently cleared, stop the reaction with a drop of vinegar and rinse immediately in water. Too prolonged bleaching will destroy the specimen.

6. Staining is necessary at some point, and doing it now, in water, helps visualize internal structures for dissection. Grey recommended chlorazole black E in aqueous solution: first put a small amount of stain into rubbing alcohol (70% isopropyl) and let stand for several days, with repeated shaking. Then run through filter paper and make a "massive" [his word] dilution with water. Optimum measurements and dilution are determined by trial and error, and the final solution is very stable. To apply the stain use a 1 ml hypodermic syringe with a No. 30 needle. The point should be ground down and the end beveled smooth. The end of the needle is worked into the thoracic end of the abdomen, which is then held in place on the end of the needle with fine curved forceps; the abdomen (held vertically with thoracic end up) is gently filled with stain until a small amount emerges from the anus. At this point the abdomen is immediately immersed in 20–25% isopropyl alcohol, to "fix" the stain. Dissection is best done after several washes at this alcohol concentration, as higher concentrations lead to brittleness and easy damage. The abdominal wall should now be clear and the internal structures lightly stained and visible. It is in the foregoing procedures that opinions differ most as to optimal methods.
7. Dissection. For the male, using fine scissors and forceps, slip the genital capsule (including the aedeagus) free of the abdomen and release any confining tissues, so that the valvae can be

spread laterally and the juxta visualized. Now remove the aedeagus, gently teasing it out in the direction of the thorax. Omit this step when working with an unfamiliar group, until you can learn whether removal will destroy definitive characters (as in Blastobasidae and Elachistidae). [Grey also chose to omit eversion of the vesica, as being too complex for a beginner. For this technique, see Lafontaine 1987 or McCabe 1980.] Transfer the aedeagus to 99% alcohol for dehydration, rolling it onto its side and flattening it slightly for later imbedding on the slide. [Again, know the group with which you are working; rolling the aedeagus on its side may obscure some characters.] "Valvae should be worked open in each bath, to facilitate final positioning. At the same time the tegumen may be rolled somewhat, to permit a good lateral view of the uncus. The latter all too often is squashed down between the valvae, obscuring other characters as well as its own shape. Another sin to be avoided is allowing the aedeagal sheath to droop down and obscure the juxta; it should be pushed up into the tegumen area." When all stray debris and air bubbles have been worked out, the organs are transferred to a 99% isopropyl alcohol dehydration bath. "After all the desired shaping, I recommend (unorthodox!) applying as much pressure as a restraining cover glass will permit without breaking, and then dropping on a nickel for good measure, to increase the distortion."

But here a word of caution. This last bit of preparation for slide mounting, with its emphasis on distortion, may indeed give a preparation sufficient for you to compare one of your unknowns with a published illustration for identification. But if it turns out that you are dealing with a true unknown (new species) your preparation will limit the opportunity of an expert to make a detailed description. With a true unknown, exhaust all other means of identification first, and if "new species" becomes a serious consideration, leave the dissection to an expert. Because genital preparations mounted permanently on slides can be viewed from only one angle, Burns (1964) advised strongly that all preparations be stored in vials (in glycerin), rather than slide-mounted. Vials should be of such size that the genitalia are in no way crowded or folded (the very small 5 x 12 mm ($3/16$ x $7/16$") vials for pin-mounting are not always roomy

enough). This allows subsequent removal and examination from any angle that taxonomic comparisons might require. Vials should have modern synthetic caps or stoppers that prevent dissipation of the glycerin (these were described in Chapter 8, Section 10). Some workers store the genitalia dry in No. 5 gelatin capsules. Because the capsule is easily pierced by a specimen pin, the genitalia may be mounted on the same pin with the specimen from which they were taken.

Grey recommended cleaning the male abdomen thoroughly inside and out, dehydrating it and mounting it on the same slide with the genitalia. In some groups it contains diagnostic features, as for subfamily recognition in Gelechiidae.

The chlorazole black staining process makes possible "see-through" preparations of female abdomens, with the internal genitalia visible in natural positions. Internal cleaning and removal of air bubbles requires meticulous work. The same dehydration process is used as for males, and they are similarly slide mounted. To make conventional female preparations, snip the abdominal segments away from the genitalia, taking care not to damage bursae lying close to the abdominal wall. For transferring the "floppy" female parts from one bath to the next, use an angled spatula made from half a spade forceps with the spade end heated and bent. Curved tipped forceps also work well. The dehydration process is the same as for males.

While Grey referred to the use of Euparal (from supply houses) as a mounting medium compatible with isopropyl alcohol and doubling as a clearing agent, he characterized the slide-making process as a "separate art" and left it for others to describe. Cribb (1973) advised as follows:

1. In the center of a clean 25 x 75 mm (1 x 3") microscope slide (continuing to use the temporary processing label for the specimen in hand) place two drops of Euparal, transferred with the end of a matchstick rather than a dropper pipette (to avoid air bubbles).
2. Lift the parts to be mounted from the dehydrating bath with the point of a pin, then submerge them in the drop of Euparal on the slide. For a male, be sure to include the aedeagus, and if the abdomen is mounted, it should be belly up. Using a dissecting needle, orient the parts so that the posterior points

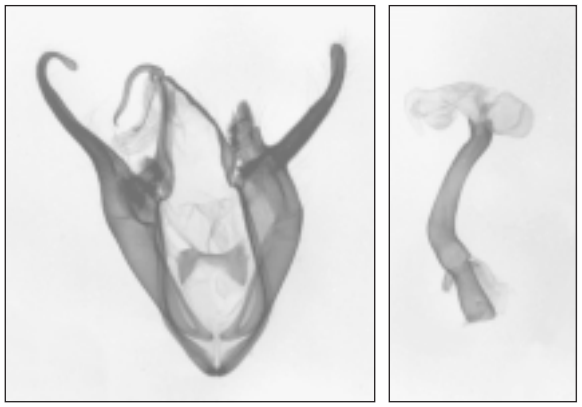

Figure 9-2. A male genitalia preparation with aedeagus separated (a catocaline noctuid: Zale buchholzi).

towards you.

3. Place a cover slip with one edge not quite touching the drop and then, supporting the opposite edge with the point of a pin, lower the slip hingelike onto the drop, very slowly so as to allow any air bubbles to drift to the edge and escape. If you first use a soft brush to wet the under surface of the cover slip with 95% ethyl alcohol, the glass makes better contact with the Euparal and air bubbles are minimized.
4. After the slide has dried a bit, clean up any mess in the label areas and apply the permanent label(s).
5. Because Euparal takes several months to dry thoroughly, the slides should be stored flat, and protected from dust, for at least a week. In cool surroundings, oven-drying of the slides at about 50°C (125°F) for several days is mandatory. In either event, they will then be firm enough to transfer to a slotted slide box stored book-like, so that the slides remain always horizontal. Figures 9–2 and 9–3 illustrate finished male and female genital slides.

Clear fingernail polish has been used as a mounting medium for genitalia and has held up well for as long as ten years (J. Taylor, pers. comm.). Its ultimate durability is unknown. It could certainly be useful as you are learning the process; it can be dissolved with acetone or ethyl acetate.

Steinhauser (1971) described a means of examining skipper genitalia without dismemberment or dissection, starting when the specimen is still fresh and spreadable.

1. "Holding the fresh insect by the thorax in normal pinching position, the genital armature can be extruded by gently pulling...the clasps [outward] with fine forceps.
2. Once the genitalia are fully exposed, grasp the abdomen with

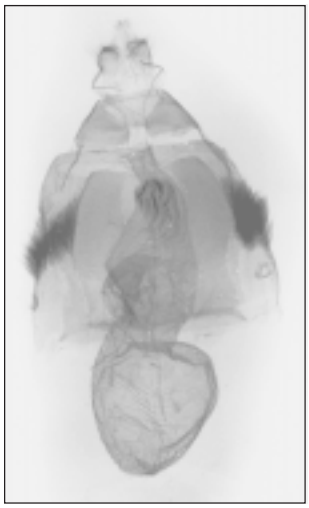

Figure 9-3. A female genitalia preparation (an acronictine noctuid: Acronicta hasta).

fine curved forceps immediately forward of the vinculum. [This] further spreads the clasps.

3. A small quantity of [Duco] cement is spread over the area of the junction of the clasps, which are held spread wide with a second pair of forceps until the cement dries, five minutes or less."
4. When the specimen, either spread or papered, was thoroughly dried, he found that the cement was easily peeled off, or failing that, was readily washed off with a drop or two of acetone, leaving the genitalia well exposed for study.

This technique, or a variation thereof, can be used for *Euphydryas, Erebia,* etc. and other species in which the clasps are the principle diagnostic components of the genitalia. It can also be used for noctuids on the boards as soon as they have been spread, and may not require cement to retain position. Caution: the protruding parts are very brittle and easily damaged.

You can photograph slides quite simply using a reversed 24 mm wide-angle lens on a camera mounted on a focusing rail so that the optical axis is vertical. A copying stand makes a solid camera support, and a homemade stage supports the genitalia slide (Figure 9–4).

The base board is about 7.5 x 30 x 2 cm (3 x 12 x $^3/_4$") and the two uprights about ($4^1/_2$ x $4^1/_2$ x $^3/_4$")—"about" indicates that you will adjust the dimensions to accommodate the mirror, flash, etc. that you will be using. The deck is 0.3 cm ($^1/_8$") thick and covers the uprights exactly (A, C, in Figure 9–4). In the center of this make a hole 3.5 cm (1 $^3/_8$") in diameter.

The shuttle is about 22 cm ($8^1/_2$") long and of a width that will just allow it to slide on the base board, back and forth between the uprights. A pair of dowel pins in the base board, as shown, delimits the path of the shuttle.

Mounted on the shuttle is a supporting block with a 45° slope, and laid against this is a mirror about 7.5 cm (3") square. A 0.6-cm ($^1/_4$") wooden curb glued crosswise on the shuttle keeps the mirror from sliding down (the mirror needs to be removable for cleaning, and for

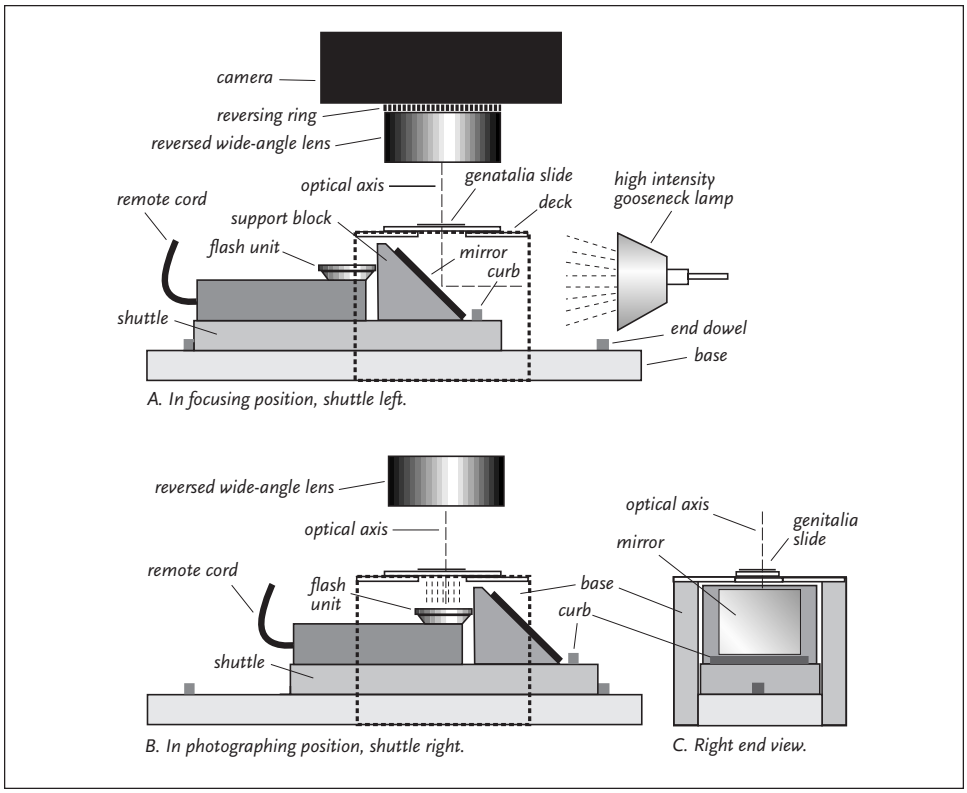

Figure 9–4. Stage for photographing genitalia slides.

wrapping when not in use).

Using the stage:

1. Set up the camera and rail on the copying stand.
2. Place the stage on the copying stand base, lay an opened paper clip across the hole in the deck, and focus roughly to ascertain the best height for the camera. Try to arrange everything so that it is high enough for you to focus comfortably, without contortions. An eyepiece magnifier helps.
3. Focusing: place a high-intensity desk lamp to the right of the stage and push the shuttle all the way to the left (A, in Figure 9–4). Lay the genitalia slide over the hole in the deck and focus carefully.
4. Shooting: place your flash unit (with remote wiring) on its back on the shuttle, touching the support block, with the output directed upwards. Push the shuttle all the way to the right

(B, in Figure 9–4). Caution: remove any "diffuser" accessory from the front of the flash, or its facets will show in the picture. Cover the front of the flash with plain white copy paper to insure a totally blank background.

5. Exposure: follow the instructions for your flash unit as to the degree of sophistication it offers. Do a test roll initially to learn the best combinations, including one or two stops each side with the over/under control.

Making genitalia preparations is time consuming and calls for meticulous attention to detail, but a well prepared slide is unsurpassable for identification, can be a thing of beauty, and makes an intriguing subject for the photographer.

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Chapter 10.

HAZARDS

Everyone can benefit from reading this entire chapter, regardless of your area of interest in moths and butterflies. It contains material of importance to anyone who enjoys outdoor activities in the wild. Its purpose is to describe some of the hazards to which the lepidopterist (or anyone pursuing any field of natural history, for that matter) may be exposed in the field or in the lab —in one's work and storage area at home. The study of moths and butterflies is not inherently dangerous, but knowledge of conditions present in the wild can make the difference between a pleasurable trip and a degree of disaster. While this book is indeed a "how-to" or a "do-it-yourself" manual, this chapter is not a "do-it-yourself" medical handbook. Its goal is to help you include "what-if" considerations in your plans and activities. The traditional "ounce of prevention" has not lost its value. An effort has been made to cover North America north of Mexico quite generally, with a few added examples of risks present in tropical countries, particularly those for which a number of weeks of lead-time may be necessary to develop protection.

Minimum need

A "look before you leap" attitude.

Defensive strategies suggested against some of the biological hazards will be followed, in the section on chemical hazards, by a description of potential risks posed by these defensive measures!

1. Noxious Arthropods

Insects contribute more annoyance, inconvenience, and outright risk than any other elements in field activity. Biting insects lead the onslaught.

In many areas, mosquitoes top the list. Their painful bites, the subsequent itchy wheals, and the buzzing in the tent at night are all too familiar. The possibility that the mosquito is carrying an infectious disease is not so obvious. Light colored clothing gives questionable protection from mosquitoes. Repellants, such as N,N-diethyl-m-toluamide (DEET, used in "Off" and "Muskol"), applied on clothing and exposed skin can protect for 8–10 hours (Schreck &

McGovern 1989). Problems with DEET arise from using the agent too often and in too high a concentration. In combination with permethrin (discussed under ticks) it is close to 100% effective in protecting from bites. Old-fashioned oil of citronella is briefly effective and significantly odorous. Avon "Skin So Soft" lotion praised by some has been a disappointment to most, and has been found to give protection for only about 30 minutes (Abramowicz 1989). The purported value of vitamin B_1 (thiamine) as an insect repellent is a myth.

There is a curious phenomenon that some people experience with mosquitoes. If the biting insect is allowed to gorge itself and fly off satisfied and undisturbed, no skin reaction develops at the site of the bite. Possibly removal of the blood meal also removes the salivary irritants initially injected. Dealing charitably with a small number of mosquitoes trapped within a tent will quickly quiet them down for the entire night (personal observations of the author). Unfortunately, nothing is known about whether this favorable treatment alters their efficiency in transmitting disease organisms.

Flies of many sorts can darken the day. Black flies (Simuliidae) are vicious biters and are active in spring or early summer; at higher latitudes they can be so numerous as to be frequently inhaled. The bites, that may not appear until many hours later, itch for a week and in young people give rise to harmless swollen glands about the back of the head and neck. Some people get enough allergic swelling of the eyelids to limit vision. Protection involves use of tight-cuffed clothing, long sleeves, head nets, and application of a repellent to exposed skin. DEET is effective and lasts for many hours.

Other biting flies, including deer flies, horse flies, and green-heads (Tabanidae), give sharp pain at the time of biting. Since they anchor their mouthparts while feeding, they are vulnerable to a quick swat at that moment! They can be kept at bay by the same approach as for black flies.

Gnats and midges can also be controlled by repellants, but they can fly or walk through ordinary netting. Newer tents have netting impervious to these beasts.

Stinging insects—ants, wasps, hornets, and bees—occur in almost all the ecosystems a lepidopterist is likely to visit, and they are hazardous to the extent that your activities interfere with theirs. If you

blunder into a hornets' nest hidden waist high among the weeds, their reaction will be defensive. If you brush off the yellow jacket that is trying to clean the remnants of lunch off your face, she will likely take issue. If attacked, it is prudent to leave the area as sedately, and with as little flailing of the limbs, as possible. This is seldom possible. Crushing the attackers releases alarm pheromones that attract further defenders. In short, the best policy is to try to anticipate and avoid—to learn to recognize signs of Hymenoptera in residence and to respect their territories.

The venom of fire ants is quite different from that of other Hymenoptera, and there are no cross-reactions in allergic people. A sting produces pain, a local blister, and a red swollen area that may take as long as ten days to resolve. If you stumble over a fire ant nest, the residents will retaliate, and multiple stings are the rule. Avoidance is the best strategy. This is the species that least qualifies as being welcome at a picnic.

If you are stung by any hymenopteran, the pain can be relieved quite quickly with a drop of ammonia (that you might have with you as a killing agent), or a paste made of baking soda or meat tenderizer, applied to the bite. Do not use ammonia on the face. A retained honey bee stinger should be scraped out with the point of a knife or a finger nail. Grasping the stinger with forceps compresses the poison sac and injects more venom. If you are a person who has demonstrated past allergic hypersensitivity to Hymenoptera stings and are at risk of an anaphylactic reaction (potentially fatal in 10–15 minutes), be sure always to carry with you an "EpiPen" or "AnaKit" (prescriptions required) so you or a colleague can administer a lifesaving injection of epinephrine. This is only a first aid measure and should be followed by prompt and definitive medical treatment (Hodge & Tecklenburg 1993).

Urticating larvae of Lepidoptera should be recognized and respected as hazardous, but not feared. Many caterpillars bear large, vicious-looking spines that are absolutely harmless—those of the mourning cloak, *Nymphalis antiopa*, or the royal walnut moth, *Citheronia regalis*, for example. Their spines are blunt and without venom. Other larvae have branching spines tipped with venom-loaded needles, such as the buck moths and io moths, *Hemileuca* and *Automeris* spp. (Saturniidae), or have fine, hollow hairlike spines

with a venom bulb at the base as in the grape leaf skeletonizer, *Harrisina americana* (Zygaenidae). Such spines may be hidden among soft, unarmored hairs. Others with venomous hairs include the browntail moth, *Euproctis chrysorrhoea* (Lymantriidae), various flannel moths (Megalopygidae), and the saddleback caterpillar, *Sibine stimulea* (Limacodidae) and other slug caterpillars. While most of these urticating larvae give merely a severe stinging and burning sensation lasting several hours, that of the saddleback can be severe enough to require hospital treatment of more generalized symptoms. Mild local reactions can be handled with cellophane tape, to try to remove the hairs, and soap and water washing. Eye irritation from urticating hairs should be treated by flushing with water, and may require medical treatment.

In times of outbreaks of gypsy moth larvae, *Lymantria dispar,* large numbers of airborne nonvenomous larval hairs can accumulate about collars and cuffs and give rise, in susceptible people, to an irritating dermatitis similar to that caused by fiberglass fragments. Application of cellophane tape to lift hairs from skin, thorough washing of clothing and victim, and use of steroid ointments can help to reduce the irritation.

Ticks and mites are very subtle as they attack, and you may not notice them for many hours.

Ticks are present from middle latitudes through the tropics. They stay attached for several days until they are fully engorged with blood drawn up through a beak (haustellum) plunged deep into your skin. They then drop off. They can be removed with fine curved forceps by grasping the haustellum right at the skin surface and yanking it out. Any feces on the skin should be flushed off, since rickettsiae (see below) can enter the body through minor abrasions.

A good defense against ticks is to tuck your pants cuffs into your socks, then spray your clothing thoroughly with permethrin (a synthetic pyrethrin available in several preparations from hardware stores and veterinarians). One application is good through as many as five launderings (Schreck & McGovern 1989). Treatment does not repel ticks, but in one experiment all ticks that explored treated clothing for as little as 15 seconds were dying within one hour (Lane 1989). Dark colored clothing attracts ticks less than white or light colors, but light colors certainly make crawling ticks more visible.

Chiggers are common in warm temperate climates and through the tropics. The tiny red larval mites attach to and digest the surface of the skin, and in many people give rise to an itching, swelling, even blistering sensitivity reaction lasting a week to ten days. Prophylaxis is the same as for ticks, and the traditional "flowers of sulphur", dusted on the outer clothing, is also useful. Chiggers transmit no diseases in the western hemisphere, but are the vector of scrub typhus in Asia.

Spiders tend to be feared more than they are understood. In contrast to the situation with all the arthropods already discussed, large warmblooded animals are not on the spider's normal menu, and spider bites, while painful, usually result when the spider is cornered and feels threatened. The bites are usually benign and will subside without special attention. There are exceptions:

Tarantulas are very large, very hairy, but very measured and deliberate in their movements. They are most commonly found on open ground in dry areas. A bite is indeed painful, but not life-threatening. It should merely be washed well with soap and water. Tarantulas also have readily-detached fine hairs that are irritating and urticating. They can be removed from skin with strips of cellophane tape, and from eyes by copious flushing with cold water. Avoid rubbing the affected skin or eyes (Smith 1982).

The black widow *(Latrodectus mactans)* is another matter. It hides in dark, dank areas not often frequented by the lepidopterist. The classical and most ignominious encounter is the bite from beneath the seat of an outdoor privy. These bites can be life threatening and require prompt medical attention. There are no effective first aid recommendations.

The brown recluse *(Loxosceles reclusa)* and closely related species in the Southwest are infrequently encountered by lepidopterists unless the creatures happen to hide in a sleeping bag or clothing, but their bites are extraordinary. There may be no pain at the site of the bite for a number of hours; a red bump, then a blister develops, progressing to a black scab after several days. When the scab separates, an ulcer is revealed; this may enlarge slowly for several months. Treatment may require surgical excision of the lesion to avoid a disfiguring scar.

Scorpions are commonly encountered in warmer climates. They

are active at night, and they sometimes retire to the shelter of an empty boot before the sun comes up. If you discover this with your toe, your day will be ruined. Inverting your shoe and banging it on the floor will usually dislodge any vagrants. Sleeping bags should be carefully checked before entry. Scorpions exposed to the ultraviolet from a blacklight in the dark fluoresce strikingly. The only treatment of a bite is application of ice (if available) and a good washing. Exception: the sting of the bark scorpion *(Centruroides sculpturatus)* of the Southwest can give severe nervous system symptoms and requires prompt hospital management. This is particularly a hazard for children under six; few adults have problems.

Centipedes, even the very large ones of the southwestern U.S., deliver a bite that is acutely painful but not dangerous. Management is as for stinging insects.

Millipedes do not bite or sting, but some species exude a noxious, odorous defensive secretion that can cause skin blistering, and in the eye can cause painful inflammation. Washing the skin, and flushing out the eye, are the best treatments. If vision seems impaired, see a physician. Children should be advised not to handle these animals.

Occasionally a small insect will have the misfortune to fly or fall into your nostril, ear canal, or eye.

For the nostril (be careful to breathe in through your mouth and out through your nose) a well-timed sneeze will usually remove the interloper.

In the ear canal a small moth is a roaring hurricane, and a plodding beetle pushing against your eardrum is a screaming horror. Drowning by dripping mineral or olive oil into the canal works quite rapidly; water is slower. The quality of the water is not important. You will eventually want to wash the bug out, using an ear syringe bulb.

If a tick, attached inside the ear canal, does not eventually release after drowning with oil, medical attention is advisable.

Lamb's wool ear plugs will protect canals during moth collecting.

In the eye, tears tend to flush beneath the lid towards the nose, and they usually drown the insect quickly. Turn your gaze downward if the creature is beneath the upper lid, upward for the lower lid. Gently massage the upper lid towards the nose with the fingertip. A companion can hold a lid down or up to look for the foreign object

and fish it out with the folded edge of a clean, tear-moistened tissue. Flushing can be tried, but the water should be clean and not laced with the chemicals often added to drinking water.

2. Diseases Acquired in the Field

The diseases acquired from infectious arthropods and vertebrates have incubation periods, from exposure to the onset of symptoms, of less than one day to as long as a year, but most become apparent in a few days to a few weeks. They are often accompanied by fever. One of these diseases may ruin a field trip almost instantly, or it may wait until after your return home to rear its ugly head. Since physicians always should, but frequently do not, inquire about a patient's recent travel when considering a febrile illness, it is of utmost importance that you volunteer and emphasize this travel information yourself. Your physician may not be thinking about diseases not regularly seen in your home area, yet treatment for some of these imported infections must be started within one or two days of onset of symptoms to be successful.

"Incubation period" (here abbreviated "Inc") is the length of time between exposure to a disease and the development of its symptoms. In the disease descriptions to follow, the most common period is stated, but longer or shorter intervals can occur. The most obvious symptoms will be preceded by "Sx."

Lyme disease, a spirochetal disease transmitted to humans from infected mice or deer, is carried by various species of the tick genus *Ixodes*, most effectively by the tiny nymphal stage, mostly in early and mid-summer. While its chief concentration is in the northeastern U.S., it is being found in more and more areas throughout the country. Antibiotic treatment is often effective. One line of defense is early search for and removal of the ticks from your skin; *I. dammini* (alternatively and controversially called *I. scapularis),* the primary vector in the Northeast, begins to transmit the infection only after about 24 hours attachment, but the safe interval seems to be shorter with other *Ixodes* species operating elsewhere. Inc: 7–12 days, but many weeks or months for neurologic and cardiac symptoms. Sx: fever, headache, rash; later, neurologic and cardiac symptoms, arthritis, especially of the knee.

Babesiosis, an infection produced by a protozoan, is mentioned

only because it is transmitted in exactly the same manner as Lyme disease, and often simultaneously therewith. Symptoms are rather like malaria and can confuse the diagnosis of Lyme disease. It occurs in the northeastern U.S. Inc: 7–14 days. Sx: fever, chills, muscle pains.

Malaria, another protozoan disease, is carried from infected humans by anopheline mosquitoes in which part of the parasite's life cycle occurs. Early treatment of infected persons is generally effective. The disease is present in virtually all tropical and subtropical areas, but it is rarely transmitted within the U.S. Travellers can be protected by prophylactic medication started one week before reaching an endemic area. Because the distribution of prophylaxis-resistant strains of malaria throughout the tropics varies continually, your physician will need to seek up-to-date advice on the area you plan to visit. This is available via the Centers for Disease Control malaria hotline, 404-332-4555. Inc: 10–30 days, varying with species of malaria parasite. Sx: fever, shaking chills, headache, intestinal symptoms.

Encephalomyelitis of several sorts is transmitted from wild birds to humans by mosquitoes. Despite the inclusion of the term "equine" in the names of some of these diseases, horses have no role in perpetuation or transmission of the disease. They, like humans, are merely (very visible) indicators that the disease is present in the area. A dead horse is hard to overlook.

Eastern equine encephalomyelitis (EEE) is seen in some years in late summer in the eastern U.S. and also in the Caribbean and in Central and South America. Only about 4% of people infected develop illness, but the mortality rate of those becoming ill is about 60%. The very young and the elderly are at greatest risk, and there is a high incidence of permanent severe neurologic damage in survivors. There is no effective treatment. Prophylaxis involves use of netting and repellants, and avoidance of evening and nighttime collecting in those times and places where current risk has been publicized (commonly in the *Papaipema* season and in attractive swampy habitats!). The natural host is birds. Inc: 7–10 days. Sx: fever, severe headache, seizures, coma.

St. Louis encephalitis, "the most important mosquito-borne disease in the United States" (Wilson 1991), occurs from Canada to

Argentina, with periodic large outbreaks in July and August in the central part of the U.S. Age incidence is similar to that for EEE, mortality is much lower, treatment is ineffective, and prophylaxis involves protection from mosquitoes. The natural host is birds. Inc: 4–21 days. Sx: fever, headache, meningitis symptoms.

Western equine encephalitis occurs from Canada to Argentina, with periodic outbreaks in the central and western U.S., mainly in mid- and late summer. Incidence is highest in infants, but higher in males than females at all ages, and in those whose occupational and recreational activities keep them out of doors. Treatment is ineffective, and mortality is about 5%. Prophylaxis again involves protection from mosquitoes. Natural hosts are birds and other vertebrates. Inc: 5–10 days. Sx: fever, headache, backache, vomiting.

Plague (bubonic plague, black plague, black death) is not just a feature of world history, but an enzootic disease of rats and other rodents occurring in pockets on every continent but Australia. Transmission is by the bite of an infective flea, by inhaled droplets from an infected human, or by a bite or scratch from an infected animal. The presence of ±20 cases per year in the U.S., primarily in the Southwest, with a 33% mortality rate, points up its significance. Avoidance of direct contact with small rodents is the first line of defense. To avoid a bite or transfer of fleas, children should be advised never to handle a wild animal that is approachable. The above-mentioned advice about reporting your travels to your physician is particularly important with this disease. Presence or absence of effective communication can be the key to cure or failure. Inc: 2–7 days. Sx: fever, headache, painful swollen gland in armpit or groin.

Rickettsial diseases of several sorts are transmitted by *Dermacentor* ticks, both by the bite and by contamination of broken skin by tick feces. Rocky Mountain spotted fever is the most publicized, and its name is misleading. In many years there have been more cases in the eastern U.S. than in the western mountains. Most cases occur from April through October. Prompt antibiotic treatment can reduce the mortality rate from about 25% to 5%. Prophylaxis is protection from ticks, as described above. Inc: 5–7 days. Sx: high fever, headache, vomiting, rash.

Tick paralysis is not an infection, but rather a nervous system disorder arising from a toxin produced by some *Dermacentor* indi-

vidual female ticks, particularly in North America. Why some ticks produce this toxin is unknown. Weakness and paralysis occur after an engorging tick has been feeding for 4–5 days, and progression to respiratory paralysis can be fatal. Removal of the tick is followed by recovery in 1–2 days.

Yellow fever occurs in tropical America and Africa, roughly from 12° north latitude to 15° south, below 2000 m (6500'), but it has occurred in isolated summertime outbreaks within the twentieth century as far north as Boston. Mosquitoes transmit the virus from infected monkeys or humans, and *Aedes albopictus,* recently introduced into the U.S., has the potential for becoming an effective vector. Mortality ranges from 5% to 50%. There is no specific treatment, but prior immunization with specific live virus vaccine is safe and highly effective. Consult your physician at least a month before traveling to an endemic area, especially if use of cholera vaccine is also being considered (Peter 1997). Protection from mosquitoes is, of course, recommended. Inc: 1–6 days. Sx: fever, chills, headache; later, jaundice and bleeding.

Biting mammals will sometimes transmit rabies. The wild animals most commonly infected in the U.S. are raccoons, skunks, foxes, and bats (but not rodents). In the West, bears have recently become involved. If any of these animals bites without provocation, it may be rabid and the bite should be dealt with as such. The rabies virus in saliva from an infected animal is infectious through mucous membranes or broken skin, without the need for a bite. Early medical treatment is the only defense against an otherwise fatal disease. It is most effective if given within the first 24 hours, but it is often effective later. Individuals who have received rabies vaccine prior to exposure are not sufficiently protected, and must have additional treatment if bitten.

The role of rodents as reservoirs of disease affecting humans, especially diseases mediated by arthropod vectors, has been indicated above. In addition, a hantavirus infection (a hemorrhagic fever) has recently been identified in the southwestern U.S. There is no arthropod vector. It is acquired through broken skin, or inhalation of aerosols of secretions and excretions of small rodents. Camping out of doors, rather than in vacant cabins, may be a means of avoidance.

Contaminated water is another widely available source of infec-

tious disease, most commonly by ingestion, but also, in some instances, by wading or swimming in it.

Enteric bacteria, that is, bacteria from the intestinal tracts of other humans, are responsible for much of the "traveler's diarrhea" infamous for marring so many exotic vacations. It results from drinking contaminated water that has been neither boiled nor chemically treated, from eating salads, or fruit that you have not peeled yourself, and most important, using ice of uncertain pedigree. Tooth-brushing with unsafe water is another common source of infection. Beverages made with boiling water are safe, as are bottled carbonated drinks that you open yourself. Water treated with iodine tablets (such as "Potable Aqua" from sporting goods stores) is safe but tastes bad. Taking antibiotics to prevent these infections will sometimes work, but it is not recommended. Widespread prophylactic use of antibiotics eventually induces large numbers of drug-resistant strains of bacteria, and treatment becomes increasingly difficult for all patients. Inc: 1–3 days. Sx: abdominal cramps, nonbloody, often explosive, diarrhea.

Bacillary dysentery, with onset as early as 7–8 hours, bloody diarrhea, and severe straining, is caused by different bacteria and requires different treatment.

Cholera is a bacterial infection endemic in southern and southeast Asia, and on the Gulf coast of the U.S. Spread to other areas is frequent, including a recent epidemic in South America. It is acquired from food or water bearing human fecal contamination. The organisms can survive in ice, salt water, boiled mineral water, and in foods such as partly cooked fish, crabs and shellfish. Treatment of the severe diarrhea with fluid replacement and antibiotics can avert death from dehydration. The available vaccine offers only a few months of protection and is half the time ineffective; it is seldom recommended and is not currently required anywhere in the world. Inc.: 1–3 days Sx: painless voluminous watery diarrhea.

Amebiasis is caused by a protozoan commonly found in Mexico, India, west and south Africa, and parts of South America. It spreads from person to person largely through contaminated food and water. The organisms are killed by boiling and by iodine treatment of water, but not by normal chlorination procedures. Treatment of the painful bloody diarrhea and the internal complications is possible with various medications. This can be a major disease. There are no

prophylactic medications. Inc: 1–3 weeks. Sx: abdominal pain, fever, diarrhea, often bloody, weight loss; may become chronic.

Giardiasis "is the most common cause of chronic diarrhea after travel" (Wilson 1991). The lepidopterist at greatest risk is the camper who utilizes surface water for drinking. The purest looking stream may have been contaminated farther up by a resident bear, beaver, or wolf. *Giardia* cysts remain infective for months in cool water. Untreated, the disease can give cramps and diarrhea for months. Several effective medications are available. Inc: 1-3 weeks. Sx: watery, frothy, foul-smelling diarrhea; abdominal discomfort and flatulence, fever.

Shistosomiasis is caused by several species of flukes that pass part of their life cycles in fresh water snails. Free-swimming cercariae, released from the snails, penetrate intact skin in as little time as 30 seconds, during wading or swimming, or they are ingested in drinking water. The disease is prolonged and debilitating, and can affect the skin and almost any other organ in the body. It is treatable, to a degree, with one medication. Inc and Sx: skin rash, after a few hours; fever, after 2-10 weeks (if it occurs at all); liver and kidney disease, many weeks to several years later.

In certain situations inhaled dust can introduce infectious disease. These illnesses are not special hazards for lepidopterists, but simply something to consider in the event of obscure illness developing after return from an endemic area.

Coccidiomycosis (valley fever) results from inhalation of dust containing spores of the infecting fungus, and it is widespread in the southwestern U.S. Infection can occur from working with dry soil, being out in a dust storm, or even driving through a dusty area with windows open. Most infected individuals never develop disease, but a few will develop chronic disease in the lungs or elsewhere in the body. Inc: 10–14 days. Sx: fever, cough, chest pain.

Histoplasmosis is caused by another fungus living in the soil. In North America it is commonest in the Ohio and Mississippi river drainages. The disease is often acquired from stirring up dust in caves or chicken houses, gathering dead wood, or simply from inhaling dusty air. Incubation and symptoms are as for coccidiomycosis.

The list of diseases that the field lepidopterist might encounter is far longer than the limited selection presented here. Anyone interested in a full list of the specific risks for a particular country can

benefit from consulting *A World Guide to Infections* (Wilson 1991). The information is organized by countries and by specific diseases.

3. Hazardous Vertebrates

Amphibians and reptiles frequent many places enjoyed by lepidopterists, and some encounters can be dangerous.

While toads secrete a poisonous mucus from the skin, this is ordinarily not a problem unless it is inadvertently transferred from your hands to your eyes or to the lining of your mouth. The irritation produced can be relieved by flushing with copious amounts of water.

The Gila monster, found in the Southwest and in Mexico, can deliver a poisonous, bulldoglike bite and may hang on for a quarter of an hour. While no deaths have been documented, the bite gives pain and local swelling, and general symptoms of weakness and dizziness. Medical attention should be sought. The animals should be watched for and avoided, and should never be handled. They are a protected species in Arizona.

Coral snakes are small but have exceedingly toxic venom. Bites require prompt medical attention. Coral snakes have red and yellow bands in contact with each other. In harmless mimics these colors are separated by black. First aid: transport immediately to medical help; if this will take more than 30 minutes, apply a loose constricting band between the heart and the bite, as near to the bite as possible.

Rattlesnakes are found throughout the U.S. but they are most numerous—and therefore produce most bites—in the southeastern and southwestern areas of the country. Venom from a bite can be life-threatening and can cause local tissue damage. The following recommendations on precautions, control, and treatment are quoted verbatim from Smith (1982, p. 112–113), by permission of the College of Agriculture, University of Arizona.

Precautions and Control: When involved in outdoor recreations such as hiking, camping, hunting, or fishing, follow these precautions:

1. Do not handle, harass, or attempt to kill or capture snakes.
2. Do not handle "freshly killed" snakes.
3. Do not approach within striking range while attempting to identify a snake.
4. Do not put your hands or feet in places you cannot see. Look

before you move.
5. Look before you sit.
6. Move objects only after assuring that hands will be out of striking range of any snake that might be underneath.
7. Do not gather firewood after dark.
8. Hike on trails and avoid tall grass and heavy underbrush.
9. Make camp on open ground.
10. Do not hike or hunt or fish alone.
11. If you hear a rattlesnake, stand still until you are sure of its location; avoid running or jumping blindly.
12. If you are bitten, get away from the snake as quickly as possible to avoid multiple bites.

Treatment: Epidemiologic studies conducted by David Hardy have revealed that over 95% of recent Arizona rattlesnake bites occurred in a location where the victim was within 30 minutes of professional medical assistance. For this overwhelming majority of potential snakebite victims, the following first aid procedure is recommended:

1. Keep the patient calm, warm, and reassured.
2. Apply a constricting band to the bitten limb between the bite and the heart, close to the bite. **Important:** The band should be loose enough to permit insertion of a finger between it and the constricted limb.
3. Immobilize the limb with a splint and/or sling.
4. Do not use ice, cold packs, or freon sprays to treat the bite. Do not apply a tourniquet (ligature that stops all blood flow). Do not apply a cut and suck procedure.

In the extremely unlikely event that several hours or longer will be required to obtain professional medical attention, an incision-suction procedure as outlined below may be of benefit to the bite victim. **Important:** Improper incision procedure may cause unnecessary injury to the victim and accomplish no benefit. The instructions contained in most commercial "snake bite kits" are dangerous and should not be followed.

1. Calm the victim, apply a constricting band, and immobilize the bitten limb as outlined above.
2. If a very sharp instrument is available (such as a scalpel blade, hobby knife blade, razor blade, or very sharp penknife), it may be used to make a short linear incision through the skin at

each fang puncture. The incision should be 3 mm deep and 5 mm long (1/8" deep by 1/4" long) and should be accomplished by a single controlled stroke of the blade or by inserting the sharp point into the fang puncture and lifting the blade to cut away from the body. Do not press the blade into the flesh and do not attempt to incise with a dull instrument.

3. After incisions have been made or a decision made not to incise, suction is applied to the bite (suction applied to the uncut fang punctures is useful). Oral suction is effective, or a mechanical system may be used.
4. After begun, suction should be continued for at least 45 minutes, but should not delay transportation of the patient to a medical facility.
5. Do not use ice, cold packs, or freon sprays to treat the bite. Do not apply a tourniquet (ligature that stops the flow of blood). Do not incise a bite on a finger or thumb. Do not make more incisions than the one through each fang puncture.

Cottonmouths are aquatic snakes found mainly in the southeastern U.S., although the western cottonmouth is sometimes found in uplands in south central areas. They may vibrate their tails like rattlesnakes when disturbed. Because these snakes can so closely resemble the harmless nonvenomous water snakes, any aquatic snake within the range of the cottonmouth should be considered dangerous (Conant 1958). The methods for avoiding rattlesnakes should be equally applicable here, as should the management of bites.

Copperheads are in the same genus as cottonmouths but prefer drier habitats, hence the alternate name "highland moccasin," as opposed to "water moccasin" for the cottonmouth. They are retiring and rather nonaggressive unless pressed. The same precautions and management apply. However, because their bites rarely inject more than a small amount of venom, antivenin treatment is generally unnecessary.

A few species of "hindfang" snakes, with venomous fangs near the back of the upper jaw, occur along the Mexican border. They are not often encountered, and their venom is only minimally toxic to humans. They should not be handled.

Large carnivores should be kept in mind in mountainous, forested and northern regions.

Grizzly bears are the greatest concern and, within their range, should never be forgotten and never taken for granted. They do not tolerate close approach, especially if accompanied by cubs, and can initiate a potentially fatal charge that no human can outrun. Since the bear would rather avoid you, carrying a rattly noisemaker, such as a tin can containing a few pebbles, tied to your wrist or ankle, can alert the bear to your presence and allow it to retire. They occur in the Rocky Mountains of U.S. and Canada, British Columbia, Yukon Territory, and most of Alaska (Burt & Grossenheider 1952).

Black bears are found in the Appalachian Mountains and adjacent forests, Florida and the Gulf Coast, the northern Midwest, the Rockies and mountainous regions to the West Coast, and all but the most northern parts of Canada and Alaska. This species is generally less aggressive than the grizzly bear, but should be accorded equal respect.

The mountain lion is sufficiently secretive as to be very successful in avoiding people, but again, a female with cubs should be admired only from a distance.

Alligators should not be overlooked in the swamps and along watercourses in the Southeast. They grab a limb, then make a rolling retreat into the water, aiming to break the limb and drown the victim.

Various miscellaneous vertebrates will sometimes bring you grief.

A skunk may do his nighttime foraging along the same route that you have selected for sugaring for moths. He should obviously be treated with great deference. In the event of a catastrophe the best move is to travel in the open back of a pickup truck to the nearest shower, undress completely (out of doors) and shower several times, head to toenails, with a good shampoo or dish detergent (not the dishwasher type). Any lingering aromas can be diminished by applying and flushing off the traditional tomato juice. Contaminated clothing can be confined to a plastic bag for subsequent laundering or dry-cleaning, or it can be buried.

Different chemistry is employed by Nigro (1996), who states: "To remove the smell of skunk from a pet, mix 1 quart of 3% hydrogen peroxide (from drugstore) with a quarter cup of baking soda and a teaspoon of liquid soap. Bathe the animal with this and rinse with tap water." No mention is made of its efficacy on people, other than a related advisory to "stay out of restaurants for three days."

Landowners, especially those bearing arms, are a potential hazard

as they investigate lights wandering through their woodlots. It is advisable to obtain advance permission for sugaring or blacklighting on private land. It is sometimes also worthwhile to make an early evening visit to the local gendarmerie to explain that the bluish light that will be glowing in the boondocks carries no sinister implications. Trying to explain after the fact has been known to prove difficult.

Raptors are only rarely a problem, but I have had the experience of setting up a light trap in close proximity to the nest of a broad-winged hawk in my yard. Each morning as I checked the trap, the hawk would stoop to within an inch of my pate in a very threatening manner. Wearing a heavily padded leather cap gave considerable psychological comfort.

4. Noxious Vegetation

It pays to be familiar with plants that can cause grief for the lepidopterist. The damage that can occur to a net entangled in brambles, thorns, or saw-edged vegetation can equally befall clothing or skin. The list of armed plants is long and their designs varied, and some of them create impenetrable barriers or traplike entanglements.

Sandburs are a particularly vicious example, waiting as they do to pierce the knee of the photographer maneuvering into position for a shot. They tangle in socks and shoelaces, to come to your attention just at bedtime. While they are most safely removed with forceps, very wet fingers can sometimes soften the tips of the spines enough so that they can be comfortably removed by hand.

Urticating plants give red, raised hivelike lesions upon light contact with the skin. They produce more pain than harm, and the lesions usually subside within half an hour. Some tropical varieties are far more potent than the temperate species. Getting to know nettles is very worthwhile, not only to avoid brushing against them, but also because they are such a useful larval foodplant for a number of nymphalid butterflies.

Poison ivy, poison oak, and poison sumac *(Rhus radicans, R. toxicodendron,* and *R. vernix),* produce a severe itching, blistering, oozing skin eruption in susceptible individuals. The eruption may begin in a few hours to several days, depending on the intensity of skin contact. After known contact, prompt, copious washing with soap and water can reduce the severity of the rash. The species

present different growth forms in different parts of the country. Learning to recognize the toxic and the nontoxic species of the genus *Rhus* is an important part of the education of any outdoors person. Avoidance is the key word. Good sources of botanical information are tree and shrub field guides, such as Petrides (1958, east, and 1992, west), or botany manuals, such as Fernald (1950).

5. Allergies

Some people with a propensity for developing allergies become allergic to elements of Lepidoptera, be it to scales, cuticular debris, or fecal particles. Reaction occurs either on contact with the skin or eyes, or on inhalation. Symptoms are a raised itchy rash, inflamed itchy eyes, a "hay fever" sort of itchy runny nose, or outright wheezing and asthma. This occurs most commonly in people employed in insect rearing facilities, but also in research workers and occasionally in amateur lepidopterists (Mass Med Soc 1984). Your physician can advise on management of acute symptoms, and this is another situation where sharing with the physician the possibility of a Lepidoptera-related illness is prudent. Long-term management may involve switching to a different group of insects or working in an airflow directed away from your body and out the window. In extreme cases, reducing your level of activity with Lepidoptera to outdoor photography might be necessary.

6. Physical Hazards

A number of hazards, physical in origin, oppose the naturalist in the field. Many are general, a few are specific to lepidopterists.

Routine scrapes, cuts and puncture wounds await anyone enjoying the out-of-doors. They can usually be handled with a good soap and water washing, application of "triple antibiotic" ointment, and a bandage. To protect against tetanus, a ubiquitous and potentially lethal risk from any penetrating injury, including stings and bites, get a tetanus toxoid booster every ten years to maintain your immunity.

Sprains, dislocations, and fractures, from simple falls or from stepping into an animal burrow, can leave you unable to reach safety and medical attention.

The cartoon of the lepidopterist, net in hand, eyes on the prize, stepping off the edge of a cliff, is drawn from life. This happens! The

best precaution is to keep aware at all times of what is (or is not) beneath your next footfall, and never to wander alone in sparsely populated areas.

Alterations of temperature and hydration: the human body is designed to operate most efficiently within a narrow range of internal temperature and level of hydration, and it has sophisticated built-in mechanisms to maintain this balance. If heat is gained from the surroundings faster than the body can dissipate it through sweating and radiation, or if it is being lost to the environment faster than it can be regenerated through shivering or other muscular and metabolic activity, then serious debilitation—hyperthermia or hypothermia—develops, accompanied by altered mental functioning, especially impairment of judgement and decision making.

Dehydration—from water loss (perspiration) in excess of water intake—accelerates development of hyperthermia. In hot, arid climates perspiration evaporates so rapidly as to be hardly noticed. Once any of these conditions has developed it may be very difficult for the victim to seek help, if alone.

Prophylaxis involves forethought and planning before entering a hot environment. Drink extra fluid before starting out, and carry enough with you to meet the anticipated needs of the day, if there is no certainty of being able to find refills. Wear loose light-colored clothing that will reflect rather than absorb the sun's heat. Shade your head with a broad-brimmed hat. Seek shade and reduce your activity during the hottest part of the day—most butterflies are smart enough to do this! The observation that "mad dogs and Englishmen go out in the noonday sun" is not an idle patter song.

Hypothermia is a less common threat for the lepidopterist, but in high altitudes and high latitudes, where clement weather can degenerate in just a few minutes to clouds, rain, and falling temperature, the threat is real. It is most important to carry rain gear to stay dry. Wet clothing cannot retain body heat. Extra warm clothing should be available, and snack food, as an extra source of energy for body heat, can be helpful. Dehydration is not a concomitant problem.

The admonition not to explore alone applies equally to these potentially lethal problems.

In anticipation of becoming accidentally separated from a group, or if you do choose to explore alone, keep in mind that "there is just

one method to keep from getting lost, and that is to stay found" (Angier 1956). "Staying found" calls for using a compass and for certain specific strategies as you trek away from your vehicle or camp, and others for the return trip. Staying alive calls for awareness of wilderness survival tactics. Off-season reading to develop an understanding of the necessary skills could someday be lifesaving. Angier's pocket-sized book serves this purpose admirably and was still in print in 1996. Books by McDougall (1992), Mears (1992) and Olsen (1990) are likewise worth examining.

7. Radiant Energy

Ultraviolet light (UV) deserves particular attention, both as we encounter it in the natural environment, and as we use it to attract moths at night. It can injure the skin and the eyes.

Ambient UV is a normal component of sunlight (Figure 10-1). It is partially screened out by the atmosphere to a degree directly proportional to the amount of air traversed. Hence UV insolation is greater in the tropics than at the poles, greater at high altitudes than at sea level, and greatest between 11 a.m. and 2 p.m. The ozone layer in the stratosphere is a very effective ultraviolet filter, especially for the shorter UVB and UVC wavelengths. As the concentration of atmospheric ozone declines, the penetration of UVA to the earth's surface increases exponentially. In addition, the shorter the wavelength, the more light is scattered as it passes through the atmosphere, so that the UV waves reach the earth's surface from many angles in addition to the axis of the sun's rays. Scatter and reflection of ultraviolet is increased 10% by sand, 20% by water, and 80% by fresh snow (the last of infrequent significance for lepidopterists). Cloud cover, unless very heavy, is not an effective UV shield (Javitt & Taylor 1994).

Light waves are measured in nanometers, (nm, one billionth of a meter). The shorter x-rays are measured in Ångstrom units (Å, one ten-billionth of a meter). Figure 10–1 outlines the pertinent portions of the electromagnetic spectrum and their roles in causing injury.

Sunlight affects human skin in several ways (Fitzpatrick et al. 1987). Earliest and most obvious is the inflammation, swelling, pain and peeling of sunburn, which is most pronounced in fair-skinned Celtic people (who may have little or no ability to tan), less in other light skinned groups with increasing ability to develop protective

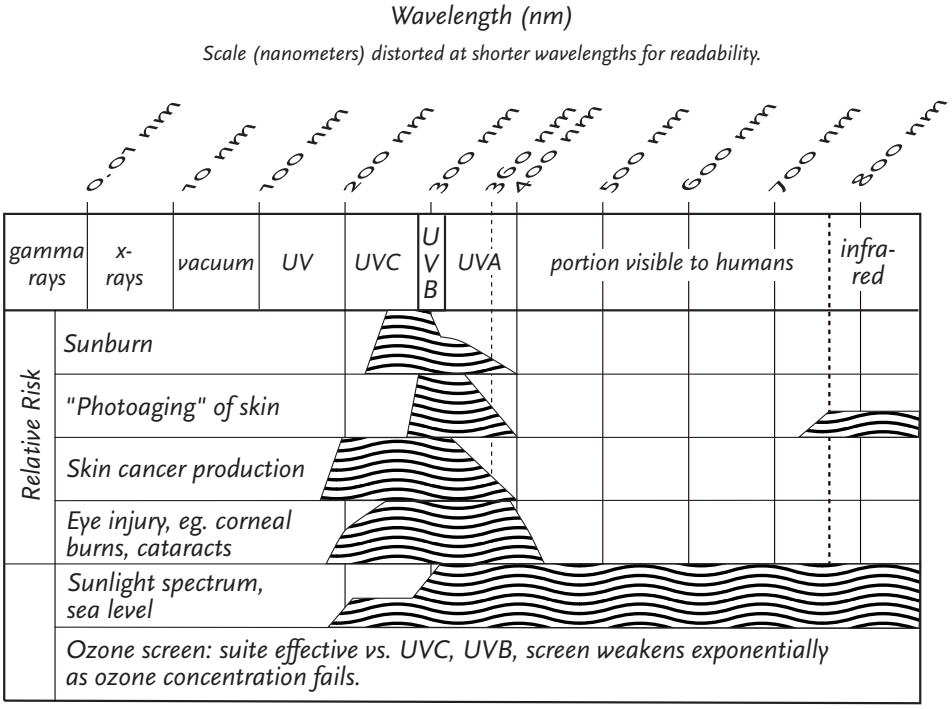

Figure 10–1. Injurious radiation.

tanning in response to UV exposure, and minimal in blacks, who are highly protected by nature.

Another effect, from repeated and prolonged UV exposure, with a lag period of several decades, is "photoaging." This results in multitudinous wrinkles, with yellowed, redundant, dry, leathery, knobby skin. It is not simply accelerated aging but a group of pathologic changes: abnormal masses of degraded elastic tissue develop in the skin. Repair and regression begins as soon as the skin is protected from further radiation, so it is never too late to institute precautions.

A third group of effects are multiple spots, blotches and excrescences that can gradually progress to become actual skin malignancies. These become common in middle age and beyond. Some are low-grade and tend to spread only locally (basal cell cancers), others are more invasive and dangerous (squamous cell cancers). Most aggressive is the malignant melanoma, rapidly growing, widely disseminating, and often appearing in the third decade of life or even sooner. There is concern that the rising frequency of this

cancer in the last few decades relates to weakening of the ozone screen in the upper atmosphere.

Skin can be protected from UV radiation in several ways:

Wise clothing selection is one way to reduce skin damage from UV radiation. Synthetic fabrics screen less well then cotton, and wet fabric of any composition screens less well than dry. Dark colors are more protective, but may contribute to overheating. Light colors seem to spook butterflies more easily, but attract fewer mosquitoes. This dilemma has indeed more than two horns!

Sunscreens provide useful protection. The sunscreen protection number is the factor by which the sun exposure can be increased before minimal reddening of the skin occurs. A factor of less than 15 is considered inadequate. A good sunscreen must remain on the skin and continue to be effective through sweating and swimming. It should be applied one-half to one hour before exposure and reapplied after swimming or sweating. Because product names and ratings keep changing, current information should be obtained from a physician or pharmacist.

The effects of sunlight on the eye relate to the same wavelengths that affect skin. UV radiation reaching a layer of the eye is either absorbed (with the potential for causing tissue damage to that layer) or transmitted without causing damage. The human cornea absorbs nearly 100% of UVC (<290 nm), but most of these wavelengths have already been filtered out by stratospheric ozone. A healthy young human lens absorbs most UV below 370 nm, and with aging it absorbs increasing amounts of UVA. Only 2% of 360 nm UV reaches the retina (Javitt & Taylor 1994).

Exposure of the cornea to excessive UV radiation gives a painful irritation, as experienced in "snow blindness" or "welder's flash." Because of a delay in onset of up to six hours, there is no early warning of overexposure. The condition subsides spontaneously in about 12 hours.

Studies of development and incidence of cataract (clouding of the lens of the eye) show significant association with excessive exposure to UV radiation. Since cataracts develop slowly over many years and show no symptoms until loss of vision ensues, this risk should not be taken lightly.

Because of the efficient absorption by the lens, UV damage to the

retina is rare and unlikely, with the exception of the retinal burns of "eclipse blindness." However, people who have had a lens removed or who already have a diagnosis of macular degeneration should take special efforts to limit possible UV exposure.

Eye protection against UV radiation is not difficult.

Wearing a broad-brimmed hat is very helpful but may interfere with easy handling of a camera.

Ordinary prescription lenses give varying degrees of protection. Glass lenses allow passage of 80% of radiation above 340 nm and so are of little benefit. Plastic lenses give excellent protection to 360 nm, then transmit increasingly to reach 80% at 400 nm (visible threshold); these are of borderline benefit. "UV treatment" of these lenses reduces transmission at 400 nm to ±1%. Contact lenses pass only 20–40% up to 400 nm, and are somewhat more beneficial than untreated eyeglasses.

Sunglasses, according to the absorptive standards set by the American National Standards Institute (ANSI), are required to screen out 60% of UVA to qualify as "general purpose" glasses. However, a *Consumer Reports* study indicated that every lens in the 200 brands they tested, from cheap to very expensive, glass or plastic, screened out at least 90% of UVA, and in most instances reached 98–99%! "Photochromic lenses," that darken in response to UV light exposure, barely met the ANSI standards when faded, but exceeded 90% absorption when totally darkened. Mirror finishes and polarizing lenses by themselves gave little reduction in UV transmission. Glasses that slide a little way down onto your nose allow a great deal of UV to pass by over the top (Javitt & Taylor 1994).

Lepidopterists in the field usually base a choice about wearing sunglasses on eye comfort, to reduce the need to squint, vs. concern that wearing sunglasses can reduce the ability to spot larvae and small insects. The degree of hazard from going without glasses relates mainly to the degree of reflectance of the substrate you are examining.

Artificial UV (BL and MV): the emissions of the UV sources used for moth collecting have been discussed in detail in Chapter 7. Risk of skin damage from exposure to these sources is very low, especially if you are wearing long sleeves to ward off mosquitoes. An exception would be collecting with an MV lamp whose outer glass bulb has

been ruptured, leaving only the inner quartz jacket. This allows passage of large amounts of UVB and UVC radiation that is dangerous to skin and eyes. *A broken bulb should be turned off immediately and removed after it cools down.*

It should be clear from the preceding discussion that collecting around artificial UV sources carries the potential for acute (but delayed) and cumulative chronic injury to the eyes. This is particularly true with MV lamps, since the risk rises with the intensity of, and your proximity to, the light source. If you collect around sunlamps, eye protection is absolutely essential, since these sources put out even shorter-wave UV. It is equally important to have front and side protection from flying glass, should a bulb explode spontaneously while you are working around it.

Protection of eyes while collecting around BL or UV is easily accomplished with properly selected goggles. Because the intensity of the MV lamp is such as to make you direct your gaze at an angle to the light source, it is important that the goggles have side pieces to give greater than 180° protection. They should also be roomy enough to accommodate eyeglasses comfortably, if you wear them, and be secure against sliding down out of place (Winter 1995). Prescription lenses and contact lenses do not begin to meet these requirements. A source of excellent and inexpensive protective goggles is NoIR Medical (Appendix L).

8. Chemicals

Many activities of lepidopterists involve use of chemicals. It is important to understand the usefulness, the shortcomings, and in particular the potential human hazard presented by each of them. Many useful agents, popular in the past, have been largely abandoned because of increasing appreciation of human toxicity. Some of this has been voluntary, and some because of rulings of the U.S. Occupational Safety and Health Administration (OSHA).

Ideally, one would like to be able to give hard and fast recommendations for the use of various chemical agents for various purposes: "Use this one, avoid that." It is not too difficult to find well documented cases where heavy acute or prolonged exposure to a particular chemical did *not* produce an adverse outcome, or at the other extreme, to find anecdotal accounts of dire results from apparently

trivial exposure. Further investigation of some of the latter uncovers evidence of a preexisting abnormality for which the chemical exposure was the "last straw." (For example, naphthalene exposure in a person born with glucose-6-phospaphate dehydrogenase deficiency can produce hemolytic anemia.) In between there is a great body of work with experimental animals demonstrating toxicity, carcinogenesis, or reproductive abnormality—but it is then difficult to know whether or to what degree these findings are pertinent for humans. Museums and other institutions must necessarily make policy decisions about informing workers of risks, setting up a protocol for protection of collections, training workers in safety precautions, providing proper ventilation, monitoring exposure times, and so on. Naphthalene, for example, is still allowed for use in museums and is still freely available in hardware and department stores for use in homes, but many museum curators have chosen to limit or discontinue its use. The avocational collector, as a committee of one or as a family undertaking, should go through a similar educational and decision-making process, even though OSHA does not require it for individuals.

A task-oriented arrangement of the subject of chemicals seems most practical, since that simplifies focusing on those agents that are pertinent to your chosen activities. *All the chemicals covered here should be studied to appreciate the breadth of the problem.*

Personal protection

Protection from noxious or disease carrying arthropods, and from sunlight, is important to all lepidopterists, with the exception of those who limit their activities to book-reading and catalog-browsing.

- Toxicity of DEET is minimal, if you use it at a concentration of 50% or less, in conjunction with adequate protective clothing, and apply it to as small an area of skin as possible. Avoid use on very thin skin, such as armpits and eyelids. The maximum safe concentration for young children is 20–30%, and it must not be used at all on infants. At most, it need be reapplied only every 4–6 hours, usually longer, or after swimming. Ingestion or heavy absorption can give nervous system symptoms and convulsions.
- Permethrins give long-term protection from ticks when applied

to outer clothing. If absorbed or ingested, they are rapidly eliminated. No specific symptoms of toxicity are described, other than allergic reactions in some sensitive individuals.
- Sunscreens are generally nontoxic. Choice depends on efficacy, availability, and absence of personal allergy to any ingredient.

Cleaning agents

Anyone rearing immature stages employs cleaning agents, since equipment must be cleaned and disinfected between usages.

- Chlorine bleach is a familiar household agent, and precautions for its use are clearly printed on the label. **Bleach must not be mixed with detergents, particularly those used for hand-washing dishes—free chlorine gas or phosgene can be produced. It also must not be mixed with ammonia (caution when cleaning old ammonium carbonate killing jars), since the mixture produces chloramine gas.** Without excellent ventilation, any of these gases quickly produces severe lung damage.
- Alconox detergent (from supply houses) is an excellent cleaning agent, produces no poisonous gases, and can be used safely with bleach.

Disinfecting immatures

Rearing occasionally calls for the use of chemicals to disinfect eggs or larvae.

- The properties of chlorine bleach have already been covered under “cleaning”.
- Potassium pyrosulfite is used as a generator of sulfur dioxide. This latter chemical is a highly irritating gas that produces sulfuric acid on contact with the moist lining of the respiratory tract. It should be used only where excellent ventilation is assured.

Preserving immature stages

This uses a few relatively familiar chemicals. They are not dangerous if you use reasonable ventilation.

- Ethyl alcohol is very useful as a pickling fluid, but it is difficult to obtain without proper permits. Its toxic properties are familiar to millions around the globe.
- Isopropyl alcohol is a satisfactory substitute and is sold in

drugstores as rubbing alcohol. Toxicity is shown mainly in the central nervous system—inebriation, headache, stupor, and coma—and often a bleeding stomach inflammation.

- Glacial acetic acid, another ingredient of pickling fluid, is obtainable from drugstores, usually by special order. This substance has irritating vapors, is locally injurious if splashed in the eye or ingested, but as a dilute solution it is a substance with which the body can deal (cider vinegar contains acetic acid).

Killing agents

A killing agent is necessary for the collector, since "pinching" has its limitations.

- Carbon tetrachloride has been abandoned as a killing agent because of its marked toxicity to the liver, even by inhalation of fumes. In addition, it stiffened insect thoracic muscles severely, making spreading difficult, and "sweated" the inside of the jar, leading to loss of wing scales upon contact.
- Ethyl acetate has been the preferred replacement. It is obtainable from some chemical and biological suppliers, and in drugstores as nail polish remover (do not use acetone-based polish remover). It is a nervous system depressant and is flammable but can generally be used safely. Its vapors are mildly irritating to the eyes and nose. Because of its volatility, a jar must be recharged frequently, and sweating can occur. After short exposure insects can recover and produce eggs successfully.
- Cyanide (potassium, sodium, or calcium) is a classical and reliable killing agent, **but it is rapidly lethal to humans on ingestion or introduction through broken skin.** Inhalation is a somewhat lower risk. Moths "knocked out" by cyanide will recover in 1–15 minutes after removal from the jar, and are unharmed for egg laying. Killed specimens have moderate muscle stiffening. **Because of its extreme toxicity, use of cyanide cannot be recommended.** Except for people with institutional connections, it is virtually unobtainable.
- Aqueous ammonia solution is a rapid killing agent that produces little stiffening but does alter green colors with exposure

beyond a few hours. It is very volatile, and on hot days pressure builds up in the jar, so that there is an outburst of fumes on opening it. These fumes are very irritating to eyes, nose, and lungs, but cause no long-term harm. Frequent recharging of the jar is necessary, and sweating can occur.

- Ammonium carbonate, a solid releasing ammonia fumes, has the same advantages and drawbacks as aqueous ammonia solution, but a jar remains effective for months without recharging. Both these agents have been used in Europe for many years and are increasingly favored in the U.S.
- Vapona, used in pheromone traps, is covered in detail below, under Fumigation.
- Ethylene glycol, used as a killing agent in some kinds of moth traps, is nonvolatile and is also nontoxic unless ingested. If spilled, it presents no environmental hazard.

Mold inhibition

Inhibition of mold growth is necessary for protection of freshly captured specimens that will not be spread or frozen immediately, and for specimens being relaxed prior to spreading.

- Captan, applied by spraying, can be used for mold control in large areas but not within collections. It has a potential for causing cancer and for disrupting endocrine gland function.
- Chlorocresol will prevent mold growth for months in papered material, as well as in the relaxing jar. It is of minimal toxicity, even if ingested. It looks like granulated sugar, and like all chemicals, it must be kept locked up and out of reach of children.
- Chlorothalonil (for same purpose as Captan), is mildly irritating to eyes and respiratory tract but nontoxic; use protective goggles when spraying.
- Formaldehyde fumes, inappropriately recommended to suppress mold growth, are extremely irritating to the eyes and respiratory tract and have been suspected of causing some of the symptoms of "sick building syndrome."
- Naphthalene has proven toxic to many people, causing liver and kidney failure, hemolytic anemia, and cataracts from prolonged exposure. It has no insecticidal properties, and its

effectiveness as a repellant is controversial. Although it does inhibit molds, more effective and less toxic agents are available.

- Phenol (carbolic acid), a solid with a melting point slightly above room temperature, is an effective and durable mold inhibitor for relaxing jars. Ingestion or absorption through the skin can give severe kidney damage, but with careful handling this problem can be avoided.
- Thymol is an effective mold inhibitor for the relaxing jar, either used directly as crystals or as a solution in alcohol. Toxicity is low.

Fumigation

Fumigating collected material can be looked upon either as a continuing necessity or as an occasional activity in response to demonstrated need. The latter approach has been adopted by many museum curators in the interests of personal safety, and it has been found to be effective. Because chronic toxicity from the fumigants commonly used was the stimulus for Environmental Protection Agency (EPA) and OSHA regulations, avocational lepidopterists are well advised to follow the same principles in caring for collections. The gist of the regulations can be found in Zycherman and Schrock (1988).

- Naphthalene has already been discussed above. Its classic and massive use in the past does not justify continuing its use.
- Paradichlorobenzene (PDB) is an effective insecticide when used as a fumigant. Acute effects of heavy inhalation are headache, dizziness, and drowsiness; chronic effects include skin inflammation and kidney or liver failure. Vapors in concentrations well below the danger level are notably irritating to the nose and eyes. If this symptom is respected, toxicity problems can be minimized. The use of PDB requires good ventilation. Because its vapors are heavier than air, the material should be applied within the top level of an enclosed cabinet that must be airtight with a good seal along the door edges. In many collections its use is limited to heavy two- to three-week treatment of single infested drawers or boxes. Toxicity is relatively low, and the material is available in hardware and department stores for home use.

- Vapona (DDVP, dichlorvos) strips are approved for use in storage cases, and are available from hardware stores for home use. Absorption through skin or by inhalation or ingestion can give headaches, dizziness, nausea, difficult breathing, excessive salivation, and achiness. Children seem more susceptible. It has not been proven to be carcinogenic, but it causes reproductive defects in animals. The material should be handled only with rubber gloves. It is insecticidal and effective in protecting stored material (Stone & Edwards 1988). A 1.3 cm ($^1/_2$") square will theoretically protect a Cornell drawer, or equivalent, for a year. However, since the rate of release of the DDVP drops to about one-tenth after a month or two, and since cutting the strip accelerates the rate of release, this figure is unrealistically long if the drawer is opened frequently. If aluminum foil is used to keep the product from staining the pinning bottom of a drawer, the aluminum is often reduced to a powder.

Degreasing

Removal of exuded body fats can be done effectively with a number of agents, and all of them have their drawbacks. However, this procedure is an infrequent activity, and with proper precautions, particularly with regard to flammability and to ventilation, it can be carried out safely.

A simple way to arrange good ventilation is to work near an open window in front of which is an electric fan directed outward. This draws air and fumes from your work area directly out of the room and reduces the chance of dissemination throughout the building.

- Benzene has been commonly used and recommended, but its high flammability and high degree of toxicity to the human blood-forming system makes it unacceptable.
- Carbon tetrachloride, used for years for "dry cleaning" in many situations, is nonflammable but also unacceptable by reason of the toxicity outlined above under killing agents.
- White gasoline has its following, and while its toxicity is probably low (it is a mixture of hydrocarbons, not all clearly defined), its high flammability makes it an unacceptable risk.
- Trichloroethane, available in hardware stores, is nonflammable and effective but carries some risk of central nervous system

depression and acute cardiac abnormalities. With good ventilation, its use does not carry a high risk. Note its ability to damage some pin labels (Chapter 8).

- Trichloroethylene has properties similar to trichloroethane, but it is significantly more toxic and has been banned as a dry cleaning and degreasing agent.
- Xylene toxicity is akin to but lesser than that of benzene; it can produce drowsiness, dizziness, inebriation, shivering and vomiting. Eye-splashes can damage the cornea.

Genitalia preparation

Preparation of genitalia (Chapter 9) requires a small number of chemicals that can be safely managed with a few precautions.

- Ethyl and isopropyl alcohol have been commented upon above, under preservation of immatures.
- Xylene (discussed above for degreasing) is an important transitional solvent in the preparation of permanent slide mounts of genitalia in balsam. It is needed in limited quantities and can be safely handled if good ventilation is maintained. It is not needed for Euparal preparations.
- Potassium hydroxide is corrosive and must be kept away from skin, eyes, and mucous membranes. The small quantities and controlled situation in which it is used for preparing genitalia makes its use possible without hazard, but it is prudent to protect eyes from splashes.
- The mounting medium Euparal is not volatile or toxic.
- Stains, such as chlorazole black E, are used in minute amounts and do not present a threat if both the dry chemicals and their solutions are kept off the skin and out of the mouth and eyes.

9. Commercial Carriers

Federal Aviation Administration (FAA) regulations restrict what can legally be carried on commercial aircraft. Variations in temperature and air pressure during flight can cause hazardous leakage from apparently tight containers. The following are some of the items so regulated (listed in FAA pamphlets from 1993 and 1994, available from your regional FAA office), that may be of interest to some lepidopterists. These items are prohibited in checked or carry-on

baggage:

- aerosols containing flammable materials
- gasoline, flammables
- propane, butane cylinders, lighter refills
- wet-type batteries, such as used in cars
- any equipment containing fuel
- safety and "strike anywhere" matches
- flammable paint and paint related material
- corrosive material
- poisonous material

There are certain exceptions for personal care and medical needs, for example:

- toiletry and medicinal articles containing hazardous material (such as flammable perfume) totaling no more than 2.2 liters (75 oz) may be carried on board. Contents of each container may not exceed 475 ml (16 fl oz) or 475 g (1 lb).
- dry ice for packing perishables, in quantities not to exceed 1.9 kg (4 lb), may be carried on board an aircraft provided the package permits release of carbon dioxide.

"This leaflet explains important regulatory requirements but is not all-inclusive." "Many other hazardous materials are also prohibited. When in doubt, check with your airline." (These quotes are from the FAA pamphlets.)

Most examples of toxic reactions to the agents considered here occur at home, and it is not the intent of this book to outline medical management. If someone, particularly a child, has ingested one of these substances, immediately call your local Poison Information Center for advice.

Symptoms of chronic toxicity may be hard to distinguish from those of other diseases. Whenever an obscure set of symptoms is being investigated, be sure to tell your physician about any chemical to which you may have been exposed.

10. Hazards to Equipment

It is a sad commentary on today's society that any unfamiliar object left unattended out of doors is at risk of vandalism. It is almost

as though the watchword were: "If you don't understand it, destroy it!" Even if you have set something out on private property, with full permission, a passerby may get his "high" for the day by trashing it.

Rearing sleeves and bags, left unattended for days at a time, are a common target. A tag bearing some advice such as "Caution: bag contains stinging caterpillars. Emergency medical care available at (phone number of local hospital)" may convey a certain amount of protection, if the miscreant can read. While a sleeve is not a major investment, loss of the caterpillars can leave you feeling bereaved.

Bait traps are a considerably larger investment and their loss to raccoons or bipeds is very frustrating.

The most vulnerable and expensive piece of equipment is the light trap. The light advertises its presence, the battery has obvious cash value, and the light source and electric eye seem worth stealing or destroying. Generators are even more attractive to thieves.

There is no foolproof defense against vandalism, but obscurity helps. Make rearing sleeves of dark green or gray material, and whenever possible set them out where they will be screened by vegetation, and far from the beaten path. Bait traps should be similarly colored and located, with extra attention to limit access by raccoons, or whatever your local varmint may be. Dark green pheromone traps work best for sesiids and are least obvious. Most difficult is finding a site where a photo-cell-controlled light trap, left unattended for several days, can function well at night without being obvious and inviting by day.

To reiterate, the purpose of this chapter is not to suggest that the study of Lepidoptera carries unacceptable risks. We constantly accept and balance risks in all aspects of our lives. The purpose is to describe many of the risks, so that you may be able to make informed choices and decisions, to adopt effective preventive or protective strategies, and when necessary, to seek reliable remedies.

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Chapter 11.

INTERACTIONS, REGULATIONS, & TRANSACTIONS

Enjoyment and study of Lepidoptera is greatly enhanced by interaction with other lepidopterists. Information that is "old hat" for one may open a new door for another. Most lepidopterists are ready to share information on biology and behavior. They will also share information on local occurrence of various species, as long as they perceive that your interests fit in with their concepts of what is good for the species and its habitat.

Minimum needs

- *Curiosity*
- *An open mind*
- *Sooner better than later, a field guide covering your area*

1. Organizations for Lepidopterists

There are many groups, scattered around the world, organized for the interest and benefit of lepidopterists. Some are broadly focused, some limited to a small geographical area. Many include professional as well as amateur lepidopterists. Some are entomological organizations that include many lepidopterists among their members.

Because so many lepidopteral organizations are "nonprofits" with volunteer officers, no paid staff and no permanent office, they may seem difficult to locate from one decade to the next, if you happen to pick up the address or phone number from an older publication. It is for this reason that The Lepidopterists' Society has adopted a "default address": Natural History Museum of Los Angeles County, 900 Exposition Boulevard, Los Angeles, CA 90007-4057.

An annotated list of organizations for lepidopterists, both domestic and foreign, is found in Appendix K, but a detailed description of The Lepidopterists' Society and its activities is presented here. Much of what is said in the next several pages is couched with this Society as a framework.

Organized in 1947, the Society is "... a nonprofit educational and scientific organization. It shall be the purpose of the Society to promote internationally the science of lepidopterology in all its branches; to further the scientifically sound and progressive study of Lepi-

doptera; to publish periodicals and other publications on Lepidoptera; to facilitate the exchange of specimens and ideas by both the professional worker and the amateur in the field; to compile and distribute information to other organizations and individuals for purposes of education and conservation and appreciation of Lepidoptera; and to secure cooperation in all measures tending to that end" (from the Society Constitution). It is worth noting the emphasis on "the amateur in the field." Amateurs have been and continue to be a major source of field observations.

The *News of The Lepidopterists' Society* is of particular value to avocational lepidopterists, since it carries notices (placement is free to members) indicating species of "livestock" (eggs, larvae, pupae) they have available for trade or sale or wish to obtain. This publication, therefore, gives very timely information as to what is available, and where. It also carries an active and early book review section giving a helpful overview of new publications, sources, and costs. Informal articles, contributed by members, cover all manner of subjects pertinent to the lepidopterist.

The *Journal of The Lepidopterists' Society* is a more formal peer-reviewed scientific publication, with articles, profiles, general notes, detailed book reviews, obituaries, and technical notes. Many of the authors are avocational lepidopterists, who need not be intimidated by rubbing shoulders with PhDs. Validity of information is more important than the formal entomological education of the author.

2. Individuals

Interaction with lepidopterists with similar interests can be as enjoyable as interaction with Lepidoptera, and the helpfulness and hospitality of lepidopterists toward one another has been legendary—a majority of lepidopterists are genuinely interested in sharing information with and learning from their peers.

The Society's biennial membership directory is indexed by personal interests, geography, and name of member; it is an introduction to others with like interests throughout the country and throughout the world. This can be a source of information by contacting individuals, prior to traveling, as to where to go to see what, sometimes eliciting from a local expert an invitation to a joint outing. Thus have begun many lifelong friendships.

In any organization correspondence is a rewarding aspect of membership for many members, yet others may not share our enthusiasm or particular interests. They may be too busy or simply not interested in responding to unsolicited correspondence. Your queries to other members may generate more responses if you include a self-addressed stamped envelope (and your fax and e-mail addresses, if you have them) with your letters. Careful consideration of how you list your own interests (that serve as an invitation to others to contact you) can protect you from queries you do not wish to consider—and you may even request omission of your name from the Directory.

The Society has its own internet website, and a regular column in the *News* strives to keep readers current regarding new websites of interest to lepidopterists.

3. Regulations and Wildlife Laws

Lepidopterists have long been aware of the importance of regulating the transport of live insects from one part of the globe to another. The problems caused by the importation and release of the gypsy moth, and the presumably accidental introduction of the cabbage white, are adequate examples. It is obviously reasonable for permits to be required for importation of live material for rearing or for exhibition. But the existence of similar regulations controlling the importation of dead insect specimens has come as a surprise to many. The regulations were there, but were often unrecognized or unenforced. Enforcement has become active, and recognition is imperative.

After considering the regulatory layers, routes will be offered at the beginning of Section 4 for planning domestic and international shipments.

Regulations are here discussed in general terms only. Because they change from time to time, the specifics and their application to your activities should be obtained from the agencies responsible for enforcement, in most instances the U.S. Fish and Wildlife Service (USFWS), accessible via your phone book. In some situations the state counterpart is more appropriate.

There are several layers of restraints. The Lacey Act was passed in 1900. It established regulations prohibiting importation into the U.S.

of species considered to be injurious to human beings or to agriculture, forestry, wildlife resources, etc. In 1981 it was amended to strengthen federal laws and allow improved assistance to states and foreign governments in enforcing fish and wildlife laws. It includes arthropods, whether or not bred, hatched, or born in captivity, as well as any part, product, egg, or offspring thereof, so Lepidoptera are not overlooked. It provides for major fines and imprisonment. Mere possession of an illegally obtained species, even if taken by someone else, is a violation. Agents acting under properly issued search and seizure warrants can confiscate illegally obtained specimens (Wade 1993).

Another layer is CITES, (rhymes with "nighties"), the Convention on International Trade in Endangered Species of Wild Fauna and Flora. The CITES list of protected Lepidoptera includes various species in the genera *Ornithoptera, Papilio, Parnassius, Teinopalpus, Trogonoptera,* and *Troides* (USFWS 1994). CITES regulations are passed by international treaty and are revised from time to time. Information is best acquired from the Office of Management Authority, USFWS, or any Wildlife Inspection Office.

In addition, each country by its own authority has the right to list its own protected, threatened, or endangered species. Countries the world over are increasingly requiring the possession of a specific permit for any and all collecting (whether of "protected" species or not). The laws of the country of origin are paramount: if a species is exported from country X (where possession and export are illegal) to country Y (where possession of that species is legal), it remains illegal for you to import it from country Y (in the vernacular, "butterfly laundering doesn't wash"). Be sure that all certificates document the country of *origin*. It is wise to contact the management authority in a country directly to ascertain its laws before collecting, buying, trading or exchanging, to avoid importing material illegally taken by yourself or exported by a supplier. Uncertified protected material brought into the U.S. is subject to confiscation and the bearer is at risk for fines and imprisonment, no matter how the material was acquired.

The Endangered Species Act was passed in December 1973 "...to provide a means whereby the ecosystems upon which endangered species and threatened species depend may be conserved, to provide a program for the conservation of such endangered species and threat-

ened species, and to take such steps as may be appropriate to achieve the purposes of the treaties and conventions set forth in subsection (a) of this section." The act provides authority for federal agencies to act in various ways to conserve listed species. It issues prohibitions against harassing (taking), possessing illegally acquired specimens of these species, and engaging in import, export, and interstate commerce (including trades); provides for fines and imprisonment under civil and criminal codes. For the photographically inclined, even close-up photography, if it results in trampling of habitat, may be interpretable as "harassing" (McKown 1992). Table 11-1 is the list of U.S. endangered species of Lepidoptera as of July 1998 (USFWS 1998).

It behooves the collector and also the watcher or photographer to know the list of endangered species and their ranges, habitats and flight periods, to avoid indiscretions. Learning to distinguish them in the field from similar-appearing species or subspecies is both valuable and difficult. Not all the endangered butterflies are referenced in a single book, but each can be found in one or another of the following books listed in Appendix J: Howe (1975), Scott (1986), Tilden and Smith (1986), or Opler and Malikul (1992); *Euproserpinus euterpe* is covered in the sphingid fascicle of MONA (Hodges 1971). The possibility of a partial extra brood outside the usual flight period should always be considered. California is host to 15 of the 21 federally listed species. Thirteen have ranges limited to that state, so an ability to recognize them is less important elsewhere. The range of one extends into Baja California. Two species occur only in Colorado. The species endangered in Florida has a broader range south of the U.S. Eleven states share either one or two endangered species. In 37 states there are no federal listings to "endanger" the lepidopterist. And there are no species of Lepidoptera listed as "Endangered (similar appearance)," in contrast to the situation existing with a few of the endangered vertebrates.

Up-to-date USFWS information is available through their website: http://www.fws.gov/.

In addition to the federal listings, many states have their own lists of endangered and threatened species, and these lists are enforceable by the USFWS under the Lacey Act. Content of these lists can be learned from state wildlife agencies and from Natural Heritage

Common Name	*First listed*	*Scientific name, and range*
Lange's metalmark	1 Jun 1976	*Apodemia mormo langei:* nr. Antioch, CA
Uncompaghre fritillary	24 Jun 1991	*Boloria acrocnema:* San Juan Mts., CO
El Segundo blue	1 Jun 1976	*Euphilotes battoides allyni:* Coastal southern CA
Smith's blue	1 Jun 1976	*Euphilotes enoptes smithi:* Coastal Monterey Co., CA
Bay checkerspot	18 Sep 1987	*Euphydryas editha bayensis:* San Mateo Co., CA
Quino checkerspot	16 Jan 1997	*Euphydryas editha quino (=E.e. wrighti):* Orange, w. Riverside and San Diego Cos., CA
Kern primrose sphinx	8 Apr 1980	*Euproserpinus euterpe:* s. CA, ? extinct.
Palos Verdes blue	2 Jul 1980	*Glaucopsyche lygdamus palosverdesensis:* Palos Verdes Peninsula, Los Angeles Co., CA
Pawnee montane skipper, also "Montana skipper," but not "Pawnee skipper"	25 Sep 1987	*Hesperia leonardus montana:* S. Platte R. Canyon, CO
Mission blue	1 Jun 1976	*Icaricia icarioides missionensis:* Twin Peaks, San Francisco Peninsula, CA
San Bruno elfin	1 Jun 1976	*Incisalia mossii bayensis:* San Bruno Mts., San Mateo Co., CA
Lotis blue	1 Jun 1976	*Lycaeides idas lotis:* Mendocino Co. and Warner Mts., CA
Karner blue	14 Dec 1992	*Lycaedes melissa samuelis:* WI, MI, northern IN, northwestern OH, NY, NH
Saint Francis' satyr	18 Apr 1994 26 Jan 1995	*Neonympha mitchellii francisci:* south central NC
Mitchell's satyr	25 Jun 1991	*Neonympha m. mitchellii:* northern IN, southern MI, northern NJ, northeastern and northwestern OH
Schaus' swallowtail	28 Apr 1976 31 Aug 1984	*Papilio aristodemus ponceanus:* Florida Keys
Laguna Mountains skipper	16 Jan 1997	*Pyrgus ruralis lagunae:* San Diego Co., CA
Callippe silverspot	5 Dec 1997	*Speyeria callippee callippee:* San Mateo and Solano Cos., CA
Behren's silverspot	5 Dec 1997	*Speyeria zerene behrensii:* Mendocino Co., CA
Oregon silverspot	2 Jul 1980	*Speyeria zerene hippolyta:* OR Coast Ranges; Diamond L., Douglas Co., OR; along Columbia R.; adjacent WA and CA
Myrtle's silverspot	22 Jun 1992	*Speyeria zerene myrtleae:* Marin and San Mateo Cos., CA

Table 11-1. U.S. endangered species of Lepidoptera.

programs. Updates of the list of endangered and threatened species are also available from the internet.

Material collected in the recent and more distant past by amateur naturalists and individual scientists has generally been soundly documented as to time and place of collection and is highly valuable to science. Many of these specimens are not now well documented in terms of the permitting that may unknowingly have been necessary at the time of collecting. The donation of these specimens to permanent repository collections may be clouded by liability for possession of inadequately documented material.

Recognizing this dilemma, the Association of Systematic Collections (ASC) and the USFWS entered into and signed a Memorandum of Agreement (MOA) 21 May 1996 (ASC 1996), that "... applies only to specimens collected at least five years prior to the date of this agreement, and neither imported into or exported from the United States within the five years prior to the date of this agreement."— a "grandfathering" of pre-May 1991 material if it is being donated to a qualified institution.

The agreement states, in effect, that:

- The donor selects a repository institution that will agree to accept the specimens, bearing at least minimal data.
- A specific form ("Appendix 1"—ASC/MOA Donation Form) will be filled out by the transferring party, USFWS, ASC, and the repository institution.
- Within 6 months of return of the approved form from USFWS to the donor, the specimens must be delivered to the accepting institution as a direct transfer not involving the ASC or USFWS. "Bulk back loan" to the transferring party is not allowed, but small groups of these specimens may be borrowed for research purposes, as allowed by the institution's routine loan policy.
- While there is emphasis on "trophy specimens" in the MOA, that emphasis is on big-game vertebrates. Insects and shells purchased from catalogs are allegedly not considered to be "trophies" (E. Hoagland, pers. comm.) if accompanied by basic data.

4. Transactions Involving Lepidoptera

For those who choose to purchase, sell or exchange specimens or livestock, there are numerous logistical details to be considered in advance. Insufficient attention to packing can result in damage to specimens. Inadequate understanding between parties to a transaction can lead to ill-will or loss of money. But most important, *failure to understand and heed the laws and regulations discussed in Section 11–3 can lead to serious breaches of law and criminal penalties.* You must determine, in detail, the *current* status of pertinent laws and regulations, of both the exporting and the importing country, prior to *any* contemplated purchase, sale or exchange of Lepidoptera. Sources of such information have already been noted.

Before you embark on a transaction be sure to find out how (by what carrier) your package can be transported and what your specific responsibilities will be in the process.

- Will the carrier handle the shipment you propose?
- Do you need merely to drop it at a retail pickup (such as a post office) and expect it to be delivered to the addressee's mailbox or home?
- Are there any required labels you can't provide yourself?
- Are any advance permits necessary (as for livestock), that you must provide or must be provided to you by a foreign recipient?
- Will you or someone designated by you need to be at a specific place and time to send your package or take delivery of it?
- Will there be any extra expenses involved, beyond expected shipping costs?

Start by contacting one or more convenient shippers (such as DHL, FedEx, United Parcel Service, U.S. Postal Service), and explain the contents and destination of your proposed shipment. If they express any uncertainties, and also if you are dealing with livestock, make a similar inquiry of the USFWS (address below).

Practical help with the U.S. aspects of importation and exportation can be obtained from the U.S. Fish and Wildlife Service, Office of Management Authority, 4401 N. Fairfax Drive, Room 432, Arlington, Virginia 22203, telephone 1-800-358-2104 (Wade 1993). They are very accommodating and return calls promptly.

Dead insects do not require a permit from the U.S. Department of

Agriculture, but all parcels should be clearly labelled as to content by the shipper by means of a complete inventory in a plastic outside envelope.

The foregoing reemphasizes the importance of permits, already discussed in detail at the beginning of Chapter 7, on collecting. Before you collect, obtain permits. Then save them forever as an integral part of your collection.

When dealing with an individual (buying or bartering), insist on a signed receipt from the seller that lists every item and certifies that all material was obtained in compliance with international laws. When purchasing from dealers it is advisable to purchase from a licensed commercial dealer subject to inspections, with proper documentation of imports and exports. All buyers should insist on an invoice listing all specimens, with origin of each, and signed by the company. Keep permanently all correspondence, invoices, receipts, etc., to document acquisition of material.

5. Sources of Lepidoptera

While a few of the suppliers' catalogs list a limited number of species of butterflies available as rearing kits, a much broader list of offerings is to be found in the "Market Place" section of the *News of The Lepidopterists' Society,* and in similar notices in the newsletters of other organizations. A provider with limited offerings of pupae listed now may next season be able to provide ova, for example, or other species, in response to your advance request.

The egg and pupal stages are the ones most commonly amenable to shipment. If a shipment of perishable material is being arranged, it is good to discuss with the shipper the circumstances at your end, and request specific advance notice of shipping date. Ova sitting all day in a sun-blazed mailbox become simply expensive baked eggs.

Papered specimens also are available through notices in the *News,* and from price lists offered by dealers.

If you are purchasing or exchanging specimens, the definition of quality looms large. What looks great to the seller may be classified as shabby by the buyer. It would be ideal if there were a universally accepted grading system, clearly interpretable and understandable. One system, rather like a traditional school report card uses "A, B, C" with or without suffixes "+" or "–" (Rahn 1982) for wing-membrane

damage and scale loss. Another system (Pavulaan 1985) uses combinations of membrane condition (A, B, C) and scale condition (1, 2, 3). Mather (1985) pointed out that one value of a specimen lies in its proof of the existence of that particular species at a specific time and place, and that a worn specimen gives more information as to how it had spent its life than did a fresh one. The utopia of a uniform grading system has not been reached, and the best advice seems to be that the participants in a transaction arrive at a mutual understanding regarding quality before proceeding further.

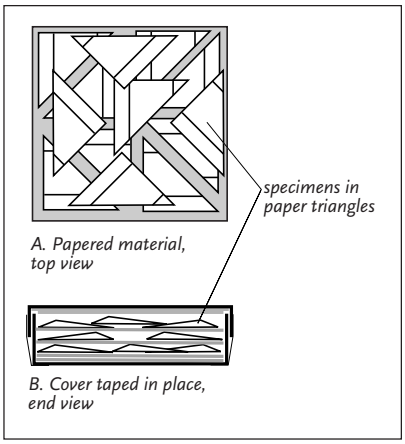

Figure 11–1. Boxing papered specimens.

In transactions involving countries other than your own, it is desirable to have an advance understanding as to acceptable form of payment, determination of exchange rate, whether payment is to be in advance or on receipt of specimens, whether unacceptable or damaged material will be replaced or refunded with cash (as opposed to credit vouchers), whether you will accept substitutions for out-of-stock material, etc.

In the event of a fraudulent domestic transaction not resolved by the parties themselves, the postal inspector can sometimes assist in resolution. Individuals victimized in international transactions can seek assistance from foreign ambassadors, who can intervene and sometimes resolve disputes.

6. Packing and Shipping Specimens

Safe shipping of dead specimens by mail or via parcel services is absolutely dependent on safe packing, but this is not difficult. It does, however, require more than a simple envelope. A well-spread, brittle-dry butterfly, placed in a letter envelope and run through the rollers of the postal service quickly fulfills the "dust to dust" prophecy. To assure safe travel, you must use "double boxing," to be described later.

Pack papered specimens with two points in mind: not so loose as

to allow jostling to occur, and not so tight as to result in crushing. This is easy to do (Figure 11-1):

1. Select a strong cardboard or plastic box with a capacity a little greater than you think you will need.
2. As you pack, make an inventory list of the contents.
3. Arrange the triangles or envelopes in layers, with the abdomens not stacked directly on top of one another. Initially and periodically put in a layer of paper towel to reduce the likelihood of the contents slipping about (A).
4. Fill any remaining space at the top of the box with crumpled tissues, but not so much as to produce crushing when the box is closed and the cover taped in place (B). Tape a copy of the inventory to the outside of the box.

Pinned specimens are shipped in insect boxes, with several special precautions (Peigler 1992). The box should be lightweight but strong. Unit trays with properly fitting covers are one satisfactory container type, as are heavy cardboard boxes made especially for shipping pinned specimens (Figure 11-2).

1. Select a box with a sound pinning bottom that will hold pins firmly. This should be 1 cm (3/8") thick and glued or stapled securely to the bottom of the box. Plastizote, cork and ethafoam are good; styrofoam and corrugated cardboard are unsatisfactory.
2. Check each specimen to make sure it is not able to rotate on its pin. A swiveling specimen can be stabilized by gluing: raise the specimen to the head of the pin, apply a small drop of glue (white glue or clear fingernail polish) below the thorax, then lower the specimen to the proper level on the pin.
3. Seat each specimen so that the pin goes through the entire thickness of the pinning bottom. This is where plier-type pinning forceps are particularly useful. A specimen with a large or long abdomen should be supported by a bare vertical insect pin placed closely against each side of the abdomen and seated firmly in the pinning material. As with papered material, make a complete inventory of each container as you pack it. Particularly valuable individuals, type specimens, or extinct species should be boxed individually in covered unit-trays.

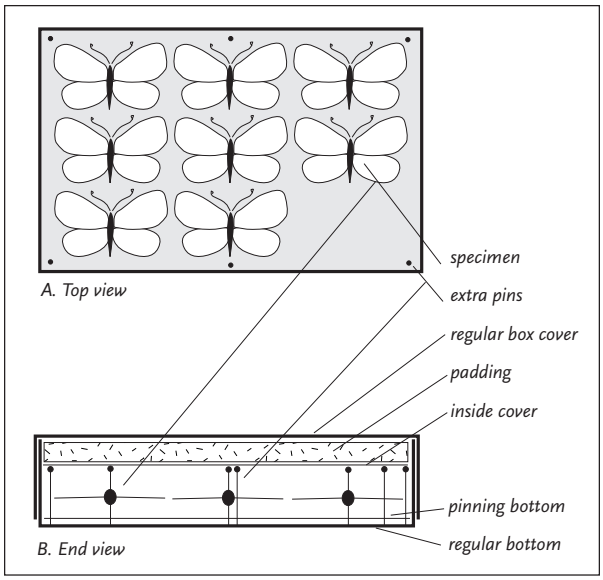

Figure 11–2. Boxing pinned specimens.

4. Cut a rectangle of cardboard as an "inside cover" so that it just fits the inner dimensions of the box, and lay it in the box so that it is supported by the heads of the specimen pins. If there are not enough specimens to fill the box, insert a few empty pins at random in the vacant areas for additional support. Place crumpled tissues, "bubble pack" or cotton on top of the inside cover, so that it is held snugly in place when the box is closed. This cover gives added security against pins working their way up and out of the pinning material during transit. A couple of tabs of tape on the top surface of this cover make it easy to remove for unpacking. The box itself should be taped or tied shut, and a copy of the inventory taped to the outside. The inner box should be clearly marked "fragile" for the benefit of a customs inspector who may choose to open it.
5. Do not include in a box containing pinned specimens any cocoons, vials or ampoules, fumigant crystals, microscope slides, or other dense objects that might work loose in transit and cause major damage.

The completed specimen containers must now be further protected, by double boxing (Figure 11-3).

1. Center the specimen container within a rugged corrugated cardboard carton that is large enough to accommodate a layer of padding no less than 10 cm (4") thick on all sides, using crumpled newspaper or packing popcorn. It should be packed tightly enough to prevent shifting, but not so tightly that it no longer cushions against shocks.
2. If you need to send several containers in one carton, tape

them firmly together and pad them as a unit, or seat them separately with ample padding to keep them from touching one another.

3. The outer box should be securely taped. Wrapping paper and twine are no longer acceptable, since the twine may snag on automated machinery for parcel handling.

Customs requirements for shipping dead insects into a foreign country vary with the country of destination, over time, and according to the purpose for which the insects are being transferred. The local embassy office of the destination country can ordinarily provide the phone number or address of their country's customs service, so that you can seek up-to-date information before shipping.

Upon import or export, all people are required to complete an import/export declaration form (USFWS Form 3-177). All people are required to notify their nearest port of entry upon expectation of an import, and all parcels are subject to inspection and clearance by USFWS. To reduce the need to enter the box to view specimens, include, in an accessible plastic pocket on the outside of the box, Form 3-177 and copies of your inventories of the specimens being shipped. Shippers of nonscientific material must complete this information and obtain clearance prior to import/export. A past practice of a private importer seeking an informal "curatorial assistant" appointment at a museum to gain "institutional status" can be interpreted as a ruse. When completing Form 3-177 note that exchange specimens have domestic value.

The extra cost of overnight or express or air shipment may be justifiable for specimens of great value. However, surface or sea mail, and third or fourth class are successful if your packaging has been done as described and time is not a factor. A large shipment should be split into several smaller packages. Registration will enable you to trace an undelivered shipment.

The first consideration in shipping livestock is its legality with regard to crossing state and national boundaries. This applies both to the insect species and to any accompanying foodplant. No shipment, or hand carriage, of live material should be undertaken without thorough prior understanding of pertinent regulations. It may be illegal to carry a monarch caterpillar from Canada into the U.S., even though, left to its own devices, the same caterpillar would fly across a

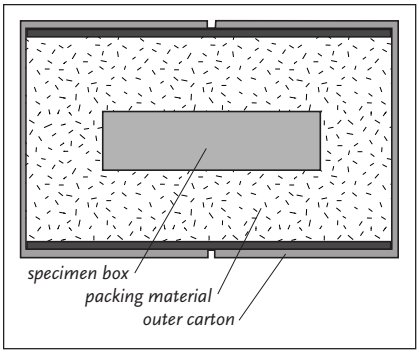

Figure 11–3. Double boxing.

few weeks later under its own power! The U.S. Department of Agriculture regulates imports and exports to and from the U.S. Responsibility for compliance with regulations for material exported from the U.S. lies with the shipper, and for imports, with the recipient. Advice and cooperation of the foreign party is likewise important. Information and necessary forms can be obtained from USDA, APHIS, PPQ, BATS, Plant Pest Permit Section, 4700 River Road, Unit 133, Riverdale, MD 20737-2036. Fax 301-734-8700. The foregoing acronyms stand for U.S. Dept. of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine, Biological Assessment and Taxonomic Support.

Responsibility for compliance with regulations for live material imported into the U.S. lies first with the recipient. Working with at least 60 days lead time, you must obtain from and submit to the USDA PPQ Form 526 indicating the name(s) of the "pest species" you wish to import, your proposed method of preventing their escape, and other pertinent information. Any plant material to accompany the livestock must have a similar application. In USDA terminology, any insect is classified as a "pest," as is any accompanying plant material. If approved, the application will be returned to you as a permit (perhaps with additional conditions attached), accompanied by green and yellow mailing labels, with full instructions, to be sent to your shipper. You must inform the shipper of these instructions. Some states require similar permitting to move live material into or out of the state (list available from APHIS). In that case you send PPQ Form 526 first to the state's Department of Agriculture, whence it will be forwarded to APHIS. Different labels are provided for state permits.

You should request a copy of "Permit Procedures for Butterflies and Moths" from USDA, APHIS, PPQ, Biologic Assessment and Taxonomic Support, 4700 River Road, Riverdale, MD 20737-1228, phone 301-734-8896 (the slight variation from the previously noted USDA, APHIS address is significant). The application/permit form

calls for the scientific name of the "pest," and that of any accompanying host plant; life stages; origin and destination, port of arrival; intended use; methods to be used to prevent escape, and other information. The permit may impose specific requirements pertinent to the species. In addition, USFWS Form 3-177, while not an application for permit, is used to notify the USFWS that you are importing or exporting. There are exceptions for exported material valued at less than $250 and not for sale (Metzler 1994). Current details, again, should be sought from the source noted above.

Some states have restrictive laws comparable to U.S. laws. When you request a BATS form, FWS will tell you whether to route the form first through a particular state agency.

The practicalities of shipping livestock vary with the developmental stage and the normal life cycle of the insect. You will need to have an estimate of the length of time the trip will take, and some concept of the temperatures to which the shipment is likely to be exposed. You will then have to judge whether the livestock will remain in its present stage (usually egg or pupa) long enough to arrive unhatched. Overwintering ova (expecting to hatch at time of budburst, as with *Catocala*) are usually fully developed unhatched larvae, ready to chew their way out in response to the first rise in temperature (such as while languishing in the post office). If they can be shipped before having had their wintertime cold exposure, unhatched arrival is more likely. Ova that will hatch without undergoing diapause may take as little as five days to as much as two weeks to hatch, depending on species and ambient temperature. Knowing what to expect can help you determine whether shipping makes sense. Good communication with the other party, so that you can know if he or she is ready to deal with the ova on arrival, is vital. Federal Express or another expeditious carrier can shorten travel time. Ova that have been deposited in patches on paper need only be layered loosely in a small shipping box, with crumpled tissue to prevent jostling. Double boxing is not necessary. Loose ova, or ova removed from their substrate, travel well in short lengths of plastic tubing with the ends plugged with cotton, or in a bottle cap covered with paper and sealed with tape. Either of these devices can be shipped in a small box or in a bubble-pack envelope.

For larvae to be shipped successfully it is necessary that the travel

time be short, that the foodplant can be kept fresh without becoming moldy, and that the subjects not be cannibalistic. The inner container should be unbreakable, caterpillar-proof, and provided with a bit of crumpled paper toweling to absorb moisture and reduce jostling. Moderately mature leaves, if the larvae will eat them, travel better than very young fresh growth. This container is then shipped in a somewhat larger box, padded with shipping popcorn or crumpled paper. Some states prohibit importation of noxious weeds (that your larvae may have chosen as their foodplant). You can obtain specific information from your state Department of Agriculture.

The pupal stage is usually good for shipping, but you still must know, by species, what time frame is available to you. While hatched larvae may be able to be rescued, adults that eclose in transit are a total loss, except perhaps as breeding stock. Pupae should be shipped in a small rigid box and protected by crumpled tissues from crushing or rattling. Double boxing is not necessary unless the primary box seems flimsy.

Shipping adult Lepidoptera should be considered primarily in terms of purpose. Breeders of butterflies in the tropics, and their customers in the butterfly houses, have discovered that the loss of life in transit is unacceptably high, in terms both economic and humane, as compared with transit in the pupal stage. The practice, for such purposes, has been largely abandoned.

On the other hand, researchers investigating genetic issues often need breedable adult butterflies for their work, or they may wish to have virgin female buckmoths, for example, for field experiments on cross-attraction by pheromones. Such live specimens, if of species that can be securely placed in paper or glassine envelopes with the wings folded over the back, can be boxed and shipped successfully by air express. Species that feed as adults (butterflies, as opposed to buckmoths), should be fed and handled, before shipping, as described for short-term live storage in Chapter 6. Amateurs commonly participate in such undertakings in response to published research requests.

7. Reporting Findings and Observations

Many of the observations that you make regarding Lepidoptera will have been made and even published before, but there is also a great likelihood that you will come up with new information. To test this,

read all you can about the species and its biology and geography (see Appendix J for book lists). If your observations seem to be new, or at odds with published information, it is time to come forward.

Vehicles for your notes and observations are several. Informal articles are accepted by various newsletters (see Appendix K) for a list of organizations and their publications). Just contact the editor of the newsletter of your choice, including a double-spaced typed or word processor draft of your article. The professional journals (listed in Appendix K) accept formal articles, technical notes, general notes, etc. These are reviewed by individuals knowledgeable in the particular group being reported upon. Each journal indicates in an "instructions to authors" page what the ground rules are for that publication. Aiello's article on preparing a report on a life history (Appendix A) gives a model for recording this very important information, and Drummond's article (Appendix B) on demystifying journal-level writing is likewise very helpful. The important thing if you have information to share is to move ahead with the expectation of success. If it is indeed new and worthy of publication, you will receive advice on putting it into optimal form.

8. Places to Visit

Whether or not you personally embrace the concept of collecting, your interest in and recognition of Lepidoptera can be broadened and strengthened by visiting collections accessible to you in local museums and universities. Many have displays open to the public, while others have scientific collections accessible only by special arrangement. You can start by calling the museum or the university entomology or zoology department to find out what is available. If you have unidentified specimens that you wish to try to identify by comparison with named material, you should indicate this interest. Museum policies in this regard vary with the institution, but many are very accommodating and may even offer professional assistance with your task. This assistance may be offered gratis, or for a fee, or perhaps in exchange for a few of your duplicate specimens. It is worthwhile to ascertain in advance which of these conditions may apply.

Another route to museum access can be the membership directory of the Lepidopterists' Society. Local members may already know, or

even be, museum personnel and may be willing to introduce you. They can also help you to learn whether there are local entomological groups whose meetings you could attend.

Gardens, enhanced for the benefit of butterflies and open to the public, are becoming more and more common in the U.S. They provide an opportunity to see what species are prevalent in the area, and will commonly provide ideas for improving the attractiveness of your own garden for butterflies. The Lepidopterists' Society has published a booklet listing butterfly gardens (Ruffin 1993).

Butterfly houses, specially designed greenhouse-like structures wherein visitors can wander on paths among flowers and free-flying butterflies, are very common in Great Britain but are only recently beginning to flourish in the U.S. They are attractive places to watch butterfly behavior: feeding, courtship and mating, and oviposition and complete metamorphosis (except in those facilities where they choose to exclude larval foodplants from the visitor area because of considerations of "neatness"). They are also excellent places to fine-tune your photographic techniques, since the subjects are numerous and tolerant. *Caution:* the intermittent use of mist generators to maintain humidity in some butterfly houses may call for quick action—popping a hat over your camera to protect it from moisture! Locations of North American butterfly houses are indicated in Appendix K.

Insect zoos are similar in concept but exhibit a broader spectrum of entomological groups. They are listed with the butterfly houses, in Appendix K.

9. Sources of Books

Descriptions and brief commentary on new books can be found in the book section of the *News of the Lepidopterists' Society,* and more detailed reviews of major works appear in the *Journal.* The *News* listings usually include a source for ordering the book, and its price.

Books published less recently are included here in a subdivided and annotated list in Appendix J.

Libraries are a good place to consult before investing in some of the less specialized books targeted for the general public. A bit of browsing can help you decide whether to borrow or buy. Don't overlook the possibility of borrowing a book through your local interlibrary loan system.

If you want to buy it, most bookstores can get you any book in print. If you have a local discount book dealer you may save significantly.

Out-of-print titles are sometimes available from specialty dealers (listed in Appendix L). Some deal solely in remainder books; others purchase used books for resale. Another avenue is to watch the "for sale" notices in the *News,* or to insert your own "wanted" notice for the title you seek. In addition, some used book dealers ("Yellow Pages" in your phone book) will look for specific titles for you (inquire about search fees).

10. Sources of Equipment and Materials

An effort has been made throughout this text to indicate a "generic" source for objects or materials whose source is not immediately obvious.

To supplement this, Appendix L lists suppliers carrying items of interest to lepidopterists. Companies in this list can provide many of the things you are looking for.

Reviewed and augmented by
Susan S. Borkin, Julian P. Donahue,
Charles Ianni, Constance Ianni, and Eric H. Metzler

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Chapter 12.

GUIDELINES FOR COLLECTING

Here and there in this manual there is mention of appropriate behavior on the part of individuals observing or studying moths and butterflies—toward the habitats being traversed, the insects under study, and toward other lepidopterists and the general public.

Guideline:

A statement or other indication of policy or procedure by which to determine a course of action.

Collecting Lepidoptera, when done with forethought, can provide positive contributions to science. It tells us what species are where, and the collected specimens enable taxonomists to clarify interrelationships among species. This in turn helps define rational routes to preservation of lepidopteran habitat and species, with potential benefits throughout the affected ecosystem.

Considerate behavior on the part of individuals observing or studying Lepidoptera has already been emphasized. This relates especially to habitat protection, understanding biology, respect for laws and for the sensibilities of others, and to use and care of collected material.

There has also been an effort here to present multiple ways to meet needs, without any "thou shalt" or "thou shalt not" imprimatur—except when it has been necessary to point out hazards.

Guidelines, such as those below, are composed to enlighten their users as to choices that need to be made as they pursue their chosen activity, and where matters of choice are superseded by matters of law.

In a poll of Lepidopterists' Society members in 1978, 88% of those responding favored definition of a policy on collecting and preservation of collections. Such a document was adopted in 1982. An update was approved in 1996 by the Executive Council of the Society. Its text follows:

The Lepidopterists' Society Statement on Collecting Lepidoptera

The Lepidopterists' Society affirms that collecting Lepidoptera is one of many legitimate activities enabling professional and avocational lepidopterists to further the scientifically sound and

progressive study of Lepidoptera and education about Lepidoptera as well as encouraging interaction between professional and avocational lepidopterists.

The foregoing statement is accompanied by the following collecting guidelines. The guidelines elucidate the manner in which collecting should be conducted. Practitioners are encouraged to adopt these guidelines and to use them for the instruction of others. They may be reproduced without permission.

Collecting Guidelines

Preamble

Our responsibility to assess and preserve natural resources, for the increase of knowledge and the maintenance of biological diversity in perpetuity, requires that lepidopterists examine the practices of collecting Lepidoptera for the purpose of governing their own activities. To this end, the guidelines are based on these premises:

0.1 Lepidoptera is one of the largest orders of insects. Lepidopterans are an important component of biological diversity.

0.2 Lepidoptera are conspicuous and scientifically well known, thus they are frequently used as indicator groups for conservation programs.

0.3 The collection of Lepidoptera

- 0.31 is a means of introducing children and adults to awareness and study of their natural environment;
- 0.32 has an essential role in the elucidation of scientific information, both for its own sake and as a basis from which to develop rational means for protecting the environment, its resources, human health, and the world food supply;
- 0.33 is an educational activity which generally can be pursued in a manner not detrimental to the resource involved.

1. Purposes of Collecting

1.1 To create a reference collection for study and appreciation.

1.2 To document regional diversity, frequency, and variability of species, and to provide voucher material for published records.

1.3 To document faunal representation in environments undergoing or threatened with alterations by humans or natural forces.

1.4 To participate in development of regional checklists and institu-

tional reference collections.

1.5 To complement a planned research endeavor.

1.6 To aid in dissemination of educational information.

1.7 To augment understanding of taxonomic and ecological relationships for medical and economic purposes.

2. Collecting Methods

2.1 Collecting adults or immature stages should be limited to sampling, not depleting, the population concerned. Numbers collected should be consistent with the purposes outlined.

2.2 Where the extent and/or the fragility of the population is unknown, caution and restraint should be exercised.

3. Data Sharing

3.1 All data should be recorded, and the data should be made available to all interested parties.

4. Live Material

4.1 Rearing to elucidate life histories and to obtain series of immature stages and adults is to be encouraged, provided that the collecting of the rearing stock is in keeping with these guidelines.

4.2 Reared material in excess of need should be released only in the region where it originated, and in suitable habitat.

5. Environmental

5.1 Protection of the supporting habitat must be recognized as the sine qua non of protection of a species.

5.2 Collecting should be performed in a manner such as to minimize trampling or other damage to the habitat or to specific foodplants.

5.3 Property rights and sensibilities of others must be respected.

5.4 Collectors must comply with regulations relating to publicly controlled areas, to individual species, and to habitats.

6. Responsibility for Collected Material

6.1 All material should be preserved with all known data attached.

6.2 All material should be protected from physical damage and deterioration, such as light, mold, and museum pests.

6.3 Collections should be made available for examination by qualified researchers.

6.4 Collections or specimens, and their associated written and photographic records, should be willed or offered to an appropriate scientific institution, if the collector lacks space or loses interest, or in anticipation of death.

6.5 Type specimens, especially holotypes or allotypes, should be deposited in appropriate scientific institutions.

7. Related Activities of Collectors

7.1 Collecting should include permanently recorded field notes regarding habitat, conditions, and other pertinent information.

7.2 Recording of observations of behavior and of biological interactions should receive as high priority as collecting.

7.3 Photographic records, with full data, are to be encouraged.

7.4 Education of the public regarding collecting and conservation, as reciprocally beneficial activities, should be undertaken whenever possible.

7.5 All known data should be recorded with the specimens, such as date, location, collector, habitat, larval host data, and parentage of immatures, when known.

8. Traffic in Lepidopteran Specimens

8.1 Collection of specimens for exchange or sale should be performed in accordance with these guidelines.

8.2 Rearing of specimens for exchange and sale should be from stock obtained in a manner consistent with these guidelines, and so documented.

8.3 Mass collecting of Lepidoptera for commercial purposes and collection of specimens for creation of saleable artifacts are not included among the purposes of the Society.

9. Legal Considerations

9.1 Collectors should comply with local, state or provincial, federal and national, and international laws and regulations that govern collecting and possession, commerce and exchange, import and export, and protection of species. Collectors should comply with additional local, state or provincial, federal and national, and international laws and regulations governing live material.

The foregoing was adopted by the Executive Council of The Lepidopterists' Society 13 June 1996, in Houston, Texas. The collecting guidelines committee consisted of:

Vitor Becker, Planaltina, BRAZIL
Lincoln P. Brower, Gainesville, Florida, USA
Charles V. Covell Jr., Louisville, Kentucky, USA
Thomas Emmel, Gainesville, Florida, USA
J. Donald Lafontaine, Ottawa, CANADA
Stephanie McKown, Camas, Washington, USA
Eric H. Metzler, Chair, Columbus, Ohio, USA
Kauri Mikkola, Helsinki, FINLAND
Scott Miller, Honolulu, Hawaii, USA
Paul A. Opler, Fort Collins, Colorado, USA
Kenelm W. Phillip, Fairbanks, Alaska, USA
Jerry A. Powell, Berkeley, California, USA
June Preston, Lawrence, Kansas, USA
Floyd Preston, Lawrence, Kansas, USA
Frederick W. Stehr, East Lansing, Michigan, USA
J. Benjamin Ziegler, Summit, New Jersey, USA

Chapter 13.

DISPOSITION OF COLLECTIONS

Even though the eventual transfer of your collection of Lepidoptera to other hands may be decades away, it is worth while to read this chapter now. It could help to fine-tune your activities with Lepidoptera. But keep in mind that some of the specifics will change, especially with regard to appraisal and taxes.

If you are a serious collector of Lepidoptera, you have the obligation to prepare and label your specimens properly and to protect them from damage and deterioration, as discussed in Chapter 8. As you develop your collection you can take specific steps to enhance its value (Stockton 1985, 1986). Furthermore, plans should be made for eventual transfer of your collection to competent hands, in the form of a museum or educational institution, or to a private buyer. This transfer should be considered and, ideally, arranged before waning interest or failing health leads to diminished care and consequent deterioration of your holdings (Merritt 1981).

When your collection changes hands, you may possibly realize considerable return, in the form of a tax deduction related to the value of your donation to an educational or other tax-exempt institution, or an after-tax profit on a sale. The various questions to be considered will be addressed in the sections to follow.

You may well gain more personal satisfaction from donating a collection to an organization where it will be reliably cared for and will be of scientific or educational benefit to inspire and train another generation of lepidopterists, than from selling it for the monetary rewards.

1. Factors Affecting Collection Value

As you collect Lepidoptera you keep in the back of your mind criteria determining what and how many specimens you are seeking. This serves to satisfy your personal goals. It is also worth considering, as you collect and prepare specimens, what you can do now to enhance your collection's value for eventual transfer.

The condition of the moth or butterfly is paramount, yet an identifiable specimen, even with much scale loss and wing membrane damage, can be of just as much value to a researcher interested in the

range and flight period of a species. In general, reared or little-flown specimens with minimal scale loss and no membrane damage are of greatest value.

Preparation should match the standards described in Chapter 8 as to pinning and spreading. Field pinned, unspread specimens are worth less but outrank papered specimens. If the wings of the latter are not folded up over the back, they are of lower value. The value of spread, unpinned specimens in display mounts can vary greatly, depending on the purpose for which the recipient may wish to use them. In any case, specimens should be pest-free and in pest-proof containers; damage from past infestation may reduce value.

Data labels (covered in Chapter 8) are absolutely essential. No label equals no value. The "where-when-by whom" label is basic; a rearing label on reared material adds some value, as does a determination label on the pin (as opposed to a group determination label in the drawer).

A sex ratio of one male to one female is most valuable, but a maximum of three to four males to one female is acceptable.

Large numbers of one species are a reducing factor, unless they come from many different areas, exhibit a range of local or clinal variation, or are of a species rare in collections.

Place of origin is significant; exotic species usually have more value. (In this context Canada and the U.S. are treated as a biological unit.)

Immature stages, whether preserved wet or dry, should be preserved in keeping with the recommendations of the pertinent preceding chapters. Material preserved wet should be in properly stoppered containers so that maintenance needs will be minimal.

The foregoing section is based largely on information provided by F. Sala (pers. comm.) and articles by Stockton (1985, 1986).

2. *Value to the Recipient*

Value, like beauty, is in the eye of the beholder, and depends considerably on the focus of a recipient institution or buyer. A collection should meet the recipient's needs, and not merely duplicate present holdings. Potential donees may turn down proffered collections because of this.

Some material is of unquestioned scientific value, however. This

includes type specimens, historical specimens, aberrants, hybrids, gynandromorphs, etc.

Sometimes a subdivision of your collection will have particular educational value for a school, nature center, etc. This might include such categories as local synoptic collections, mimicry exhibits, or host-parasite displays. Often such subsets can be made up for a particular donee without significantly reducing the overall value of your holdings. Beware, however, the potential buyer or donee who is bent on "cherry-picking"—selecting only your most valuable specimens and leaving the rest for you.

3. Related Items of Value

Maintaining a periodically updated inventory of your collection can serve you in a number of ways:

- It can introduce a potential donee or buyer to the scope and content of your collection.
- It can reduce the labor (and presumably the cost) of an appraisal.
- After transfer of your collection, it will remain a source of data for yourself and others regarding your collecting experience.
- If you become unable to oversee transfer of your collection yourself, your surrogate will have a valuable document to work from.

The inventory is best arranged in taxonomic order, and should contain the various elements covered earlier (see Chapter 3, Section 2, Computerized Records).

The drawers, boxes and other display containers housing your collection may well have appreciated in value since you purchased or built them. If they are of interest to a donee or buyer, be sure they are figured into the transaction. If they are not of interest to the donee, then they may be significant items for separate disposition.

Your collecting, rearing, and spreading equipment may be of interest to other collectors or to science and nature centers.

A segment of your holdings that may have considerable value is your library. A book's value changes over time, depending on wear and care. Out-of-print books may be in demand, or have little value if a more up-to-date publication has come along. The value of a very old book can appreciate considerably.

A description of widely accepted book condition standards will be found in Appendix I.

Single articles from journals ("offprints," "reprints," or "separates") in this age of xerography have less value than in the past but might occasionally find an interested buyer.

It helps to make an orderly inventory of books and separates you wish to offer to potential buyers. This should include author(s), title, publisher, edition (if pertinent), date, and condition. If the title is not self-explanatory, a clarifying note is helpful.

Crisp color slides or negatives of butterflies and moths are often sought by publishers and speakers, but are also becoming more and more available. On the other hand, serial high-quality photographs of life histories and of particular behaviors (resting postures, for example) can sometimes be of interest to instructors and researchers, and their availability is worth publicizing.

4. Personal Records

Collecting permits should be retained permanently so that they can be transferred with the specimens to which they relate (retain copies for yourself); review Chapter 11, Section 3, Regulations and Wildlife Laws. If a permit agreement calls for "eventual deposit" in a particular sort of institution, be sure that your transfer complies with that requirement.

Notes regarding collecting sites, dates, conditions, etc. may be of interest to a recipient, so their availability should be made known. However, for institutions that do not archive such notes, they may have no appraisal value.

If your collection transfer is to be a sale, retained records of expenses involved in its acquisition will provide a basis for reducing the taxability of your proceeds from the sale. Without such records the entire proceeds may be classified as taxable profit. This is particularly important when your collecting involves travel.

5. Inventory for Appraisal Purposes

Regardless of the type of transaction, you will save time (and probably money) in the long run if you prepare an inventory of your collection. Performed in the manner described below, this inventory will give you a broad overview of the composition of your collection

regarding numbers, degree of preparation, origin, unusual specimens, taxonomic range and relative value.

The following relative value scale (Figure 13-1) is taken from H. Weems' unpublished December 1989 version (University of Florida), adapted here to consider only Lepidoptera.

As an example of use of this form for inventory of a collection prior to appraisal, assume your collection totals 350 Lepidoptera specimens of varied species, countries of origin and degree of preparation, and refer to Figure 13-2. Numbers in plain type in the "point value" column are part of the form. Numbers in italics above the double line are those that you fill in as you do your inventory. "Dollar value per point" and "adjustment" percentage (italics) are the work of the appraiser, the former based on the current "market rates" and the latter on his or her overall judgement of the quality and preparation of your material. All the numbers in italics in this example are hypothetical.

For lines a, b, and c, enter in the "No. of specimens" column the number of specimens in each basic category; the sum of your numbers should be the 350 specimens in the example.

For lines 1 through 13, enter the number of specimens having the attributes or degree of preparation listed for each line. A single specimen may "earn" anywhere from zero to several kinds of points in the tally:

- A pinned, labeled, spread domestic butterfly is counted only on line 1 (in addition to its initial appearance on line c).
- A pinned, labeled exotic moth, not spread, but identified to species by genitalic dissection by an authority, will be included in the counts on lines 3, 5, and 7.
- A pair of pinned, spread, minuten-mounted micromoths taken in copula, and accompanied by significant biological data, will be counted on lines 1, 2, 4, and 6.
- You get no points if you identify it correctly yourself, unless you are considered by others to be an authority on that group of moths.

The total of the numbers in lines 1–13 of the "No. of specimens" column can differ widely from the total of column 1, lines a–c (the number of specimens in your collection); the latter is not used in calculating the value of the collection.

Inventory Appraisal

Attributes of specimen*	No. of Specimens	Point Value	Point Subtotal
a. Unmounted (papered, or layered; not bulk trap samples)		0.25	
b. Pinned, unlabeled (such as one-site field-pinned material)		0.7	
c. Pinned, labeled		1.0	
On each line, 1–13, enter total number of specimens having the additional listed attribute:			
1. Spread		1.25	
2. With significant biological, host or habitat data		0.65	
3. Exotic		0.45	
4. Minuten mounted		0.3	
5. Dissected (as with genitalia, etc., in microvial on pin)		2.5	
6. In copula		3.0	
7. Identified to species by an authority		2.5	
8. Identified to genus by an authority		1.25	
9. Bulk sample (such as insect flight trap, UV light trap), domestic		12.5	
More than 20 samples from same locality, domestic		6.25	
10. Bulk sample (as from insect flight trap, UV light trap), exotic		25.0	
11. Extremely rare or valuable**		__.__	
12. Special cases (gynandromorphs, extinct species, etc.)**		__.__	
13. Type specimens***		__.__	
Point total (rounded)			
Dollar value @ $___ .___per point			*$*
Adjustment (0–25% added or subtracted, depending on condition of material): Adjusted dollar value @ (±)_____%			$

* *All specimens are assumed to have basic where-when-who data on pin label, envelope, or in the case of multiple unsorted specimens from one site and date, on the container.*

** *List separately for subjective evaluation by curator.*

*** *Types have a complex list of value factors ranging from 250 down to 1.95.*

Figure 13-1. Appraisal Sheet.

Inventory Appraisal

Attributes of specimen*	No. of Specimens	Point Value	Point Subtotal
a. Unmounted (papered, or layered; not bulk trap samples)	40	0.25	10
b. Pinned, unlabeled (such as one-site field-pinned material)	30	0.7	21
c. Pinned, labeled	280	1.0	280
On each line, 1–13, enter the total number of specimens having the additional listed attribute:			
1. Spread	270	1.25	337
2. With significant biological, host or habitat data	45	0.65	29.25
3. Exotic	50	0.45	22.5
4. Minuten mounted	18	0.3	5.4
5. Dissected (as with genitalia, etc., in microvial, on pin)	7	2.5	17.5
6. In copula		3.0	
7. Identified by an authority to species	7	2.5	17.5
8. Identified by an authority to genus		1.25	
9. Bulk sample (such as insect flight trap, UV light trap), domestic		12.5	
More than 20 samples from same locality, domestic		6.25	
10. Bulk sample (as from insect flight trap, UV light trap), exotic		25.0	
11. Extremely rare or valuable**		__.__	
12. Special cases (gynandromorphs, extinct species, etc.)**		__.__	
13. Type specimens (various: factor range 1.95–250)***		__.__	
Point total (rounded)			740
Dollar value @ $___ .___per point			*$ 590*
Adjustment (0–25% added or subtracted, depending on condition of material): Adjusted dollar value @ (±)__10__%			$ 650

* *All specimens are assumed to have basic where-when-who data on pin label, envelope, or in the case of multiple unsorted specimens from one site and date, on the container thereof.*

** *List separately, for subjective evaluation by curator.*

*** *Types have a complex list of value factors ranging from 250 down to 1.95.*

Figure 13-2. Example of a filled-in appraisal sheet.

If your collection is a large one, you should find it convenient to start with tally sheets such as the example shown in Figure 13-3. Each column tabulates the contents of a single box or drawer. Summing each line (row) separately gives a subtotal at the right which becomes part of the grand total for that line on the appraisal sheet previously described (Figure 13-1).

6. Appraisal

Appraisal involves evaluation by an individual with qualifications acceptable to the Internal Revenue Service, but those qualifications are not spelled out. Appraisers should certainly be knowledgeable in the families in your holdings, and familiar with the paper work involved. The donor cannot make the appraisal, nor may the recipient institution. However, where the issue is scientific value, institutional personnel may qualify, provided that there is demonstrated objectivity

Box or drawer, kind and number								Total
a. Unmounted (papered, or layered)							a	
b. Pinned, but data not on pin labels							b	
c. Pinned, labeled							c	
Number of specimens per container								
1. Spread							1	
2. With significant biological [host or habitat] data							2	
3. Exotic							3	
4. Minuten mounted							4	
5. Dissected (genitalia in microvial)							5	
6. In copula							6	
7. Identified by an authority to species							7	
8. Identified by an authority to genus							8	
11. Extremely rare or valuable							11	
12. Gynandromorphs, aberrants, extinct species, etc.							12	
13. Type specimens							13	

Figure 13-3. Inventory tally sheet. (Lines 9 and 10 omitted as not applicable.)

and no effort to inflate the valuation for the benefit of the donor.

An appraiser should be both accessible and affordable—you would not want to ship a large collection across a continent for appraisal.

The recipient institution should be able to advise you as to an impartial appraiser, but this does not always work out, and the "yellow pages" are no help!

While direct viewing of a collection is the most effective method of appraisal, some appraisals have been successfully carried out by examining photographs of representative portions of a collection for quality of condition, preparation, and labeling of specimens, together with a detailed inventory of the material. Appraisal costs relate to time involved. While past values for individual specimens are available (as in Covell 1970), attempting to state even a rough current figure for the average individual value of domestic specimens would only be misleading. Past quotations and evaluations experienced by donors to institutions have varied from cents to dollars per specimen, and each collection must be evaluated according to content and condition, as well as the current opinion of "the market." But it is safe to say that anyone expecting that the proceeds from a collection transfer will provide for retirement is indeed unrealistic.

7. Potential Recipients or Buyers

After you have decided whether to donate or sell, how do you identify potential recipients? Most lepidopterists who have been collecting for a number of years have developed contacts—be they other amateurs, or professionals at universities or museums—who have helped with advice and identifications. Some of these, or their institutions, may already have expressed an interest in your collection, or can offer suggestions as to whom to sound out.

You may also wish to avail yourself of the "Marketplace" section of the *News of The Lepidopterists' Society*, wherein Society members can give notice of a collection to be transferred. You would include a short statement describing the collection, such as "±8000 personally-collected moths of the Great Basin region, comprising over 900 species pinned, spread and with full data," or "849 purchased specimens of... in the families of..., pinned, spread and with full data," and offering to send a more complete description, perhaps with photographs. This may sometimes find an interested party. The

Entomological Society of America also has advertising opportunities.

Occasionally an auction may be a means of disposing of a collection.

8. Considerations Prior to Transfer

Before agreeing to transfer a collection to an institution, you may wish to find answers to several questions:

- Will the proposed recipient give adequate assurance of appropriate long-term care? It is not a happy outcome to revisit your collection five years later and find it destroyed by dermestids.
- If you want your collection to remain as a unit, rather than being integrated with the institution's current holdings, will the donee honor that request? The answer is usually "no," unless it is a collection of historical significance, a series of hybridization outcomes, or other unique material. Integration increases the usefulness of a collection to students and researchers, because it saves them time. Utilization level of an institutional collection often influences funding by the parent organization!
- Will you, because of your intimate knowledge of your collection, be allowed or requested to participate in integration?
- Does the donee require a contribution of money as a condition of acceptance, to cover the costs of housing your collection? This is becoming a common practice. It is not affected by the Association of Systematic Collections/Fish and Wildlife Service/Memorandum of Agreement (MOA) discussed in Chapter 11, Section 4 (E. Hoagland, pers. comm.).
- Should you have a written contract covering the transfer, or just a "gentlemen's agreement?" This is a matter for personal judgement. But considering the issue of funds for housing, and the physical details and expenses involved in moving a collection (discussed in Section 9), it is a good idea to have a letter of agreement subscribed to by both parties.

Note the difference between a donation and a bequest. A donation transfers your collection with your guidance and during your lifetime. A bequest transfers it via your will, after your death, in keeping with your previously recorded wishes. Willingness of the receiving institution to accept the bequest should be ascertained before the provision is included in your will.

A bequest protects your wishes in the event of unexpected demise. It does not keep you from donating the collection earlier so that you can personally orchestrate the transfer. Nor does it prevent you from transferring it, instead, to some party other than the one designated in your will.

But let us suppose that you have actually promised or pledged that you will donate the collection, rather than merely asking in an informal conversation whether the institution would like to have it. If the institution later acts on that promise, as by turning down an offer of a similar collection, it then may have a legal claim on yours (P. Hurley, pers. comm.).

In the case of sale of a collection, a written agreement becomes particularly important. You should have a signed agreement indicating:

- the scope of the collection and accessory items being transferred;
- the timing and logistics of the transfer, including responsibility for its safety and for the costs of the move and any agreed-upon insurance;
- the sale price and the manner and timing of payment, such as lump sum or installments, and in what currency if international.

Keep in mind that if you are selling a collection it is not ordinarily possible to place restrictions on what the buyer can do with it after acquisition. To attempt to do so would easily drive away a potential buyer.

9. Logistics of a Collection Transfer

Before concluding a transfer agreement, you should confer with the recipient regarding the physical aspects of transfer.

- Determine whether arranging and handling the transfer will be the responsibility of the donor/seller, the recipient, or both.
- Decide whether the transfer should be insured, and at whose expense. This may be more important in a sale than in a donation.
- Ascertain whether the specimens will be transferred by hand in their present drawers and boxes, or repackaged for transfer by hand or by a common carrier. Choice of carrier should be

agreeable to both parties.

- Reach an agreement as to who will bear the costs of the transfer. This may be different in a sale vs. a donation.
- Decide whether the transfer will occur all at once, or in successive lots at later dates.
- Agree on the specific timing of the transfer.

These details should not be looked upon as intimidating. Two parties, working in good faith for the same goal—the safe transfer of a collection of significant interest and value to both of you—should not have difficulty carrying out a safe and smooth transition for your collection.

10. U.S. Tax Considerations

Because federal and state tax laws are complex and constantly changing, no specific advice can be offered here. A certified public accountant will need to advise you about your particular situation. Tax considerations are not affected by the Association of Systematic Collections/Fish and Wildlife Service MOA discussed in Chapter 11, Section 4 (E. Hoagland, pers. comm.).

Some commonly asked questions and discussions follow (this section is based largely on material provided by Robert J. Borth, CPA).

From a tax standpoint, is it more advantageous to sell or to donate a collection?

The nonmonetary benefits of collecting generally outweigh any monetary benefits, but the serious Lepidoptera collector should have some knowledge of the options available for disposing of a collection. A taxable event occurs at the time of disposition. Donating a collection can reduce taxes, while selling a collection may result in increased tax exposure.

If you contribute a collection of Lepidoptera to a qualified organization, a charitable contribution deduction is usually taken for the fair market value (FMV) of the collection at the time of the contribution. The tax savings realized would be the FMV multiplied by the taxpayer's incremental tax rate. Therefore, the deduction is worth more when made during working years when income and tax rates are highest.

The sale of a collection results in a greater tax expense if the proceeds exceed the costs of making the collection. These costs can

be offset against the gain but must be documented. Generally no loss on a sale would be recognized due to the "hobby loss" restrictions.

Who receives a charitable contribution deduction?

You must itemize deductions on Schedule A of your Form 1040 to receive a charitable deduction.

What types of organizations qualify to receive deductible contributions?

Most nonprofit charitable organizations including museums, most nonprofit educational institutions, public parks and recreational facilities, and certain Canadian or Mexican charities qualify for this purpose. Ordinarily the organization you are interested in can tell you its status, and you can often find it in IRS Publication 78 ("Cumulative List of Organizations").

How is fair market value determined?

Fair market value (FMV) is the price at which property would change hands between a willing buyer and a willing seller. An appraiser must sign the taxpayer's Form 8283, Noncash Charitable Contributions, if the donation exceeds $5000. This form must be filed if all noncash contributions for the year total over $500.

If the amount exceeds $5000 a qualified appraisal document is also required. This must not be made earlier than sixty days before the contribution date and not later than the due date of the return. This document includes information different from the appraisal summary and is not attached to the donor's return.

How do I determine my tax basis in the collection?

If you bought the collection the basis would simply be what you paid for it. However, if you collect the specimens yourself, your basis includes relevant costs for collecting equipment, storage, identification, and travel.

When might I need to calculate my basis?

If you donate specimens held for less than one year they are ordinary income property and the deduction is limited to the FMV less the amount that would be ordinary income if you sold the property for its FMV. Generally, this rule limits the deduction to your basis in the property.

If you sell a collection, you deduct your basis from your revenues to determine your taxable gain. Losses are not allowed where the

activity is not engaged in for profit. These "hobby loss" rules do not apply to commercial dealers where the particular circumstances show there is a profit motive history. A bargain sale to a charitable organization for less than the FMV is partly charitable contribution and partly a sale that could result in some taxable gain.

Previously, those taxpayers in an alternative minimum tax situation were required to use their cost basis. Relaxation of these preference rules now apply only if there is a carryover of charitable contributions.

What if I contribute a collection that has decreased in value?

If you contribute a collection with a FMV that is less than your basis in obtaining it, your deduction is limited to FMV. No deduction is available for the difference between cost and fair value.

Are there any limits to the amount of deduction that can be taken in a given year?

The amount of the deduction may be limited to either 20%, 30%, or 50% of your adjusted gross income, depending on the type of organization receiving the donation. Generally, the 30% limitation would apply to capital gain contributions such as a collection.

Can I carry over my contribution that I am not able to deduct in the current year because I exceeded my adjusted gross income limit?

You can deduct the excess in each of the next five years until it is used up. Any amount that is not used after five years is lost.

The donor should enlist the donee's assistance in proving that a donation is related to the donee's exempt functions to avoid reducing the amount of the contribution for tax purposes.

What is the deductibility of an appraisal fee, or of my time spent in making the collection?

Appraisal fees are not deductible as contributions, but can be listed as "miscellaneous deductions" on Schedule A. The value of time and services is not deductible.

What documentation is required for cash donations made to provide cabinetry or for other purposes?

A cancelled check is fine for contributions less than $250. A written acknowledgment is required for contributions of $250 or more. It generally must be received by the date you file the return

and include the amount of cash contributed and whether the organization provided anything to you as a result of the contribution.

When is the contribution deductible?

Usually a donor makes a contribution at the time of its unconditional delivery. The donor must show that he or she has irrevocably transferred title to and dominion and control over the collection. A donor who merely allows an organization a right to use the collection may not take a charitable deduction for the value of this right. There are some partial exceptions to this, too complex to outline here.

Are sales of collections subject to sales taxes?

Sales and use taxes would generally apply to commercial dealers. An individual selling a collection may be able to avoid sales taxes as an occasional sale. Laws vary, state to state.

Reviewed and augmented by
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Patricia Hurley JD, and Frank P. Sala

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Appendix A

How to Prepare Publishable Reports of Lepidopteran Life Histories

A. Aiello

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Introduction

Development is intrinsic to all living things, and, among insects, Lepidoptera undergo perhaps the most remarkable transformations of all. Their larvae (caterpillars) are often large and colorful and, even if small and dull, they exhibit countless interesting behaviors. Lepidopteran pupae may display striking structural or behavioral features as well. Within minutes following its escape from the pupa, the crumpled adult expands its wings to their full size and prepares for its first flight. The splendor of adult Lepidoptera, especially butterflies, has been extolled in art and poetry from earliest recorded times.

In spite of the fascination of metamorphosis, however, the immature stages of Lepidoptera are remarkably less studied than are the adults. In fact, for the majority of species, especially tropical moths, the life histories have not yet been described. That is lamentable, because immature stages and larval foodplants offer a wide array of characters of great value to the systematist attempting to understand evolutionary relationships.

Because habitat destruction is eliminating many species before their immatures can be discovered, one cannot overemphasize the importance of gathering and sharing as much life history information as possible in whatever time remains. A new item of information on immatures, no matter how seemingly trivial, may be useful to a person studying that particular species, or may provide the stepping stone for another person to discover more complete details.

The aim of this paper is, 1) to encourage the amateur to contribute to our knowledge of lepidopteran life histories, and, 2) to provide guidelines for writing useful, publishable reports on the immature stages of butterflies and moths. To make a useful contribution, one need not resort to technical language or highly detailed descriptions. What is important is that the information presented be accurate and that it be communicated in a clear and concise way. It is also important to preserve the adult and, if possible, any discarded immature structures (i.e., larval head capsules, pupal skin, cocoon).

For each life stage there is a paragraph listing the possible components of a description. That is followed by more detailed comments on those components of the description that may require them. You may wish to expand or abridge these lists, depending upon the characteristics of the species you are describing.

Two readily available works that will provide the beginner with an elementary knowledge of butterfly form, structure, biology, and behavior, are Pyle (1984) and Douglas (1986). The Pyle book includes a chapter on moths. Examples of helpful, yet simple and nontechnical descriptions of butterfly immatures can be found in DeVries (1987). If you are so inclined, simple line drawings or photographs can be made of the various stages. Those wishing to develop their artistic skills further will find detailed information on scientific illustration in Hodges (1989). Use of the terms stadium, stage, and instar follow the definitions given by Entomological Society of America (ESA) Publications Council (1985). An older, but excellent introduction to insect natural history is Frost (1959). The most widely used glossary of entomology is Nichols (1980).

Other helpful publications are mentioned under the sections on each stage.

Rearing methods. It is important to report your methods of maintaining and rearing the life stage(s) of your butterfly or moth. For example, was it reared in a petri dish, or a cage of some sort, or did it remain on the original larval foodplant with a mesh bag over it? If it was reared indoors, you might wish to describe something of the conditions, including temperature, and light regime.

1. Eggs

A description of an egg might include collection data; oviposition data; egg size, shape, sculpturing, and color; whether the egg was laid singly or was part of a cluster or string; whether it had any sort of covering; observations on development; egg parasitoids obtained; and what method you used to preserve the egg.

Collection data. Collection data would state where and when the egg was collected, including whether it was obtained in the wild or in captivity.

Oviposition data and larval foodplant. If oviposition was observed, note the time, give a brief description of weather conditions, and report any distinctive behaviors.

If the substrate was not a plant, state what the eggs were laid on, for example: petri dish, or cage screening.

If the substrate was a plant, try to obtain its scientific name, including the author, and if possible give the local common name. It is important to make a voucher specimen of the larval foodplant and to cite that specimen and state where the specimen has been deposited. Labels for plant specimens differ in an important way from insect labels. Plant specimen labels include a collection number. Each plant collector keeps a notebook listing those numbers, together with collection data and identifications. When citing a plant specimen, the collector and number are given in italics, with no punctuation between the name and number; examples are: *Bailey 258, Howard & Proctor 13762*. If the specimen is not numbered, give the collector's name in italics, followed by "in" plus the year in regular type, for example: *Purdie* in 1843.

Explain what part of the plant the egg was laid on. For example, if the egg was laid on a leaf, note whether it was placed on the leaf upper surface or lower surface; near its base, middle or apex; on young or mature foliage.

A warning about leaf terminology: when discussing leaves, it is best to avoid use of the terms "dorsal" and "ventral". The first applications of those terms to leaves were made by plant embryologists, and for reasons related to the orientation of embryonic leaves, dorsal and ventral mean just the opposite of what one might instinctively assume. Even if the writer knows the correct meaning, the reader may not, and vice versa.

Egg size. Egg size (height and width) is given in mm. An acceptable estimate of size can be made using a ruler. If a dissecting microscope is available, more accurate measurements can be made, especially if the scope is equipped with an ocular grid.

Egg shape. To determine the shape of an egg, view it from the side. Most eggs can be described by one of the following terms: spindle-shaped (fusiform), barrel-shaped, turban-shaped (turbinate), conical, spherical, dome-shaped, or flat. If you prefer a more graphic description, that is fine. For example, the spindle-shaped egg of a pierid butterfly could be described as a slender, upright egg, about three times longer than wide, broadest toward the middle and tapering toward the ends.

Egg surface sculpture. If a dissecting microscope or a good hand lens is available, you may be able to include observations concerning the surface texture or sculpturing of the egg. Surface texture and sculpturing can be quite variable. The egg can be shiny or dull, and it can be smooth, pebbled, pitted, ribbed, or grooved. If the egg is pitted, note whether the pits are round

or hexagonal. If there are ribs or grooves, note how many. If there are cross-ribs, try to estimate how many there are. If there are hairs, note their location. Surface sculpturing can present an optical illusion; pits can appear as bumps. If you are able to examine a broken egg, or the partially eaten edge of an egg shell, the nature of the sculpturing will be much easier to interpret.

Egg covering. Eggs, especially when laid in clusters, are sometimes covered by foam or by setae (hairs) from the female.

Egg development. Observations on development would include changes in color and pattern; and time (usually given in days) from oviposition (or collection) to emergence of the larva.

Egg parasitoids. If, instead of a lepidopteran larva, one or more parasitoid wasps emerge from an egg, that fact should be noted. It may be possible to obtain identifications of such wasps, at least to family, from a specialist at a large museum. Most specialists are eager to exchange identifications for host information and specimens. The wasps may be pointed (glued to the tip of a small cardboard triangle, which is then pinned and labelled as if it were a lepidopteran specimen) if you wish, but usually such tiny wasps are preserved in 80% ethyl alcohol (or 70% isopropyl alcohol if ethyl alcohol isn't available).

Egg preservation. Intact eggs may be preserved in 80% ethyl alcohol. Egg shells are best pointed; when placed in alcohol, they usually float because they tend to retain a bubble of air inside. It works well to cut the bit of leaf that the shell is attached to and mount that.

Egg References. Two papers by McFarland (1972a, 1972b) provide an excellent introduction to the diversity, description, and photography of lepidopteran eggs. Supplementary information can be found in Downey (1980).

2. *Larvae*

There are many points worth considering in an account of larvae. At bare minimum, one should report collection data, including the larval foodplant, and should provide a general description of the final instar. Beyond that, the description can be as complex as you decide to make it.

A fairly complete account would include behavior at emergence from the egg; duration (with dates) of each stadium, including whether the larva entered into diapause at any point; a brief description of each instar (including head width); feeding habits for each instar; distinctive behaviors (including shelter construction, defenses, and resting postures); the larval foodplant; and whether parasitoids were obtained.

Larval behavior upon emergence from egg. The larva may escape from the egg by chewing an exit in the top or the side of the egg; the hole may be clean and circular, or irregular, or the larva may chew a long slit (like a latitude line on a globe) to produce a flap; after exiting the egg, the larva may eat the shell partially or completely, or it may not eat it at all.

Duration of larval stadia. Record the number of days that the larva remains in each stadium. It is easy to detect that a larva is nearing a molt by watching for the following two events. 1) The larva will stop eating prior to each molt. If you remove the fecula from the container each day, it will be obvious when feeding has ceased. 2) As the new head develops it will expand and pull out of the old head capsule, gradually filling the prothorax and swelling it to several times its normal size. As well, on light colored larvae, the stemmata (larval eyes) of the new head may show through the sides of the prothorax. During molting, the old head capsule will separate from the body. In most cases, the larva eats its old skin, but usually ignores the old head capsule, which can be pointed for study.

Larval description. Larval descriptions need not be elaborate. For example, to describe an *Ammalo* (Arctiidae) larva as having a smooth red head and a body covered with stiff black setae

(hairs), gives the reader a clear idea of its general appearance. The more detail the better, however.

A description of a larva might include, HEAD: width (in mm), color, pattern, texture, presence of setae (singular: seta); BODY (thorax and abdomen): color, pattern, presence of setae, length and color of setae, abundance of setae (i.e., dense, sparse), presence of other body projections (for example the curved horn of sphinx larvae). Be sure to distinguish between stripes (which run parallel to the body), bands (which go around the body), and oblique lines (such as are found on many sphinx larvae).

When reporting larval size, use head width. Body length increases as the larva feeds, and decreases temporarily following each molt. However, if you wish to report it, body length of a freshly emerged first instar, or of a fully fed final instar can help give the reader a clearer general impression of a larva, especially if it is an extremely large species.

Undoubtedly, the final instar will invite a more complex description than will the earlier instars, because the final instar tends to have a more complex pattern and more setae than do earlier instars.

As mentioned above, examples of helpful, yet simple and nontechnical descriptions of butterfly immatures can be found in DeVries (1987). Eventually, you may wish to advance to more technical larval characters, such as setal arrangement and proleg type. If and when you do, you'll find Peterson (1962) to be especially helpful, not only because it is well illustrated by line drawings, but because it gives comparative information on several different systems of naming setae.

If sufficient numbers of larvae can be collected to allow preservation of a few in addition to those reared, then more detailed descriptions can be made later by a specialist in the group, or by you once you have gained experience. A quick method of preserving larvae is to drop them in tap water, bring the water to a boil, turn off the heat, blot the larvae on a piece of paper towel, and then place them in 80% ethyl alcohol.

Larval feeding habits. Because larval feeding habits often are characteristic of a particular taxonomic group, they can provide information valuable to the specialist. Larvae may be root or stem borers; they may eat leaves, flowers, fruits, seeds, or rotting wood; or they may be scavengers or even predators.

If the larva eats leaves, make note of the type of feeding damage. Larger larvae tend to eat the whole leaf, usually beginning at the margin and proceeding in a pattern characteristic of the species. Less commonly, they eat holes in the leaf, away from the margin, a type of feeding much more frequent in beetles than in Lepidoptera. Smaller species, and early instars of larger species, tend to scrape the leaf surface rather than eating the whole leaf. Their damage can take several forms. Some species scrape the surface and part of the leaf tissue, leaving pit-like marks (pit-makers). Others eat all the way to the other side of the leaf, but leave the window-like epidermis of the far surface intact (window-feeders). Still others eat all the way through the leaf but leave all the veins intact (skeletonizers).

Report whether the larvae are solitary or feed and molt in an aggregation. If they are aggregated, note whether they remain so throughout larval development, or become solitary in later stadia.

Larval behavior. The countless variations in shelter construction, camouflage, and methods of defense employed by larvae make larval behavior the most fascinating part of the rearing process.

Shelters and perches. Larval shelters include mines, tunnels, cases, silk retreats, and leaf shelters. Some larvae make special resting perches.

Leaf mines are made by tiny larvae that eat the inside of the leaf, without damaging either upper or lower epidermis. Leaf mines are of two main types, blotch mines (covering a broad

area) and linear mines (long narrow mines that may meander all over the leaf). Upon completion of feeding, the larvae of some species pupate inside the mine, others drop to the ground to pupate, and still others sever a round or elliptical section of leaf from the mine, and drop to the ground within it to pupate. The damage made by pit-makers and window-feeders (see paragraph on larval feeding habits) may be mistaken for leaf mines until examined more closely. See Hering (1951) for information and further references on leaf miners.

Some larvae make tunnels of fecula and/or frass, held together by silk, on the leaf surface or on bark, and reach out one end to feed. Usually the tunnel diameter increases with larval age.

It is important to distinguish between fecula and frass. According to Frost (1959), the accepted term for insect excrement is "fecula". "Frass" refers to nonexcrement waste particles, such as the wood chips discarded by wood borers.

A number of unrelated larvae make moveable cases from silk, or from leaf material or soil particles held together with silk. Some case makers are case-bearing leaf miners; they remain in their cases on the outside of the leaf, but make a hole in the surface and reach inside to feed.

Many larvae construct silk retreats on the leaf surface. Such retreats may be such flimsy affairs that the larva is clearly visible within, or they may be more elaborate.

A large number of larvae roll, fold, or cut and fold leaves to make shelters. Others merely attach two leaves together, one above the other with silk. Many larvae that make leaf shelters retain their fecula inside the shelter. Why they do that is not known, but it may serve as a barrier against predatory wasps and parasitoids. Early instars of certain nymphalids rest on special perches made by attaching fecal pellets end to end with silk to produce a delicate, thread-like support. Note whether the larva also accumulates fecula or leaf pieces at the base of its resting perch.

Resting positions and postures. A variety of resting postures are found among larvae that remain exposed (i.e., not in a rolled leaf or other shelter) during some part of their life. The larva may remain on the upper surface of the leaf, or hidden beneath the leaf or on some other part of the plant; it may align its body with leaf veins or other plant parts; it may grasp its support with both ends, or it may hold on with one end and extend the other (usually the head end) out at an angle (typical of many geometrids and some noctuids), or it may hold on with the mid section only and let both ends of the body hang down.

Defense. Observations on larval defense are worth reporting. The larva's pattern and/or color may blend with its background, or may stand out in sharp contrast to it; the larva may mimic a twig, a dry leaf, or other inedible or perhaps dangerous object (appropriate movements may enhance the effect); the larva may have false eyes on the thorax or in some other position; it may have eversible odor glands (e.g., dorsal, prothoracic osmeteria, typical of papilionids; ventral, prothoracic glands of some nymphalid and notodontid larvae); or the larva may have urticating (stinging) setae (typical of certain families including Limacodidae, Megalopygidae, some Saturniidae). The larvae of some species engage in twitching or thrashing movements when approached by a parasitoid or predator. These movements may be synchronized among the individuals of an aggregation.

Larval foodplant. If you collected the lepidopteran as a larva rather than as eggs, report the larval foodplant in your discussion of the larva. For information, see the paragraph on larval foodplants under **Eggs.**

Parasitoids of larvae. The larva may be parasitized by wasps (Hymenoptera) or flies (Diptera). Record how many parasitoids were obtained; whether parasitoid pupation took place within the host remains, on the outside of the host remains, or entirely away from it.

If the parasitoid(s) pupated on the outside of, or away from, the host, note whether they pupated separately or in an aggregation, and what sort of cocoon(s) were made. Parasitoid flies don't make cocoons, they pupate within their own final larval skin, which becomes brown or

black. Adult flies should be pointed; the wasps may be pointed or placed in 80% ethyl alcohol. As mentioned under **Eggs**, it may be possible to obtain an identification from a specialist at a large museum.

It is worth noting that some species of flies lay white eggs, visible to the unaided eye, directly on their larval host. Often the eggs don't hatch until the lepidopteran larva is about to pupate, and it may be possible to remove the eggs in time to save the larva.

3. Pupae

Although the majority of moth pupae are brown and difficult to describe without resorting to technical details, there are many simple but useful points worth reporting. Among them are length; color; pattern if any; striking morphological features, if any; presence of a silk girdle; location and orientation; presence and nature of a cocoon; whether solitary or in aggregation; duration of pupation, and whether diapause occurred.

While most moth pupae are brown, there are species with colored and/or patterned pupae. The pupae of butterflies often are strikingly colored, as examples, the shimmering silver pupae of many ithomines, the snail-mimicking pupae of certain *Anaea* species, and the snake-mimicking pupa of *Dynastor darius*.

Many pupae bear projections, for example, the spined pupae of many species of *Heliconius* and pierids, the various head horns of many nymphalid pupae, and dorsal row of stalked star-shaped spines found on pupae of *Historis odius*.

The pupae of papilionids, pierids, hedylids, lycaenids, riodinids, and certain geometrids have silk girdles that encircle the pupae and help hold them in place against the substrate. Many hesperiids pupate supported by a silk sling.

The pupae of most moths are enveloped by a cocoon, made of silk, although some pupae are found naked among leaf litter. The color of the silk varies, as do the other materials (leaves, larval setae, fecula, frass) that may be incorporated into it. The cocoon may be a communal one constructed by an aggregation of larvae.

Report the location of the pupa and its cocoon (if any), especially if it was collected in the wild. Was it in the ground, in the leaf litter, between leaves, or was the pupae on or suspended from some substrate and without a cocoon?

If eventually you would like information on the technical terminology pertaining to pupae, consult Mosher (1916).

Parasitoids of pupae. The same set of considerations outlined in the paragraph on parasitoids of larvae applies here.

4. In Closing

To be able to report information accurately, it is not enough to be a good observer; one must write those observations down and not entrust details to memory. Take this test. After a good look at your subject, whether it be egg, larva, or pupa, look away and write a description of it. Then look again. Very likely you will find it necessary to adjust your description to conform to reality.

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On the next pages are five examples of rearing-data forms for the personal computer (filled in with italics to indicate their use), and one example for recording rearing data by hand. They may be copied without permission.

1. *This is the fully developed blank form, before any data have been inserted. It is best to use a fixed-width font (such as Courier, used here, or Monaco) rather than a proportional-width font, so that the numbers and letters will be aligned in neat vertical columns. "Lot #" and "Locality" are set up with tabs-right, so added text aligns with right margin. As data are entered, delete unused lines in each set of stages.]*

```
Lep. Family:                          species:                    Det. by:                       Lot #
Collected as:                         Date:                       By:                            Locality:
Hostplant Order:                      Family:                     species:                       Det. by:
Parasitoid Order:                     Family:                     species:                       Det. by:

Duration 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35 36 37 38 39 40 41 42 43 44 45
(month)  01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12 13 14

Egg
Instar 1
Instar 2
Instar 3
Instar 4
Instar 5
Instar 6
Pupa
Adult

[Dated Comments, photo frames:]

Duration 46 47 48 49 50 51 52 53 54 55 56 57 58 59 60 61 62 63 64 65 66 67 68 69 70 71 72 73 74 75 76 77 78 79 80 81 82 83 84 85 86 87 88 89 90
(month)  15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28

Egg
Instar 1
Instar 2
Instar 3
Instar 4
Instar 5
Instar 6
Pupa
Adult

[Dated Comments, photo frames:]

Duration 91 92 93 94 95 96 97 98 99 00 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35
(month)  29 30 31 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11

[...overflow onto subsequent page, as necessary to accommodate the notes and observations.]

[A "header" or "footer" such as the following can be helpful for locating species in a paper-copy file.]

Page No.                              Anartia fatima                          Lot: A. Aiello #99-000
```

2. *When rearing several individuals of the same species, in individual containers, this format is useful. The individual numbers go on the left, and the instar numbers are recorded daily. The "month" line is rearranged by cutting and pasting to give the correct starting day and to remove the extra day or days from short months.*

Lep. Family: *Nymphalidae* species: *Anartia fatima* Det. by: *A. Aiello* Lot # *Aiello 99-021*
Collected as: *larvae* Date: *29 Sept 1999* By: *A. Aiello* Locality: *PANAMA, Prov. Panama, Balboa, Ancon*
Hostplant Order: *Scrophulariales* Family: *Acanthaceae* species: *Blechum brownei* Det. by: *A. Aiello*
Parasitoid Order: *Diptera* Family: *Tachinidae* species: *undetermined* Det. by:

```
Duration 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35 36 37 38 39 40 41 42 43 44 45
Sep-Oct  29 30 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12

Indiv 1   a  a  b  b  b  b  b  b  P  P  P  P  P  P  P  Adult
Indiv 2   a  a  a  b  b  b  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  Fly
```

29 September collected 2 larvae at 5 a.m. by flashlight, eating B. brownei, possibly 4th instars, but uncertain. 5 Oct a tachinid larva emerged from one caterpillar.

[Continue dated comments, photo frames.]

3. *When rearing several individuals of the same species, in the same container, this format is useful. The instar number goes on the left, and the number of individuals in each stadium is recorded each day.*

Lep. Family: *Nymphalidae* species: *Anartia fatima* Det. by: *A. Aiello* Lot # *Aiello 99-019*
Collected as: *larvae* Date: *9 Sept 1999* By: *A. Aiello* Locality: *PANAMA, Prov. Panama, Balboa, Ancon*
Hostplant Order: *Scrophulariales* Family: *Acanthaceae* species: *Blechum brownei* Det. by: *A. Aiello*
Parasitoid Order: Family: species: Det. by:

```
Duration 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35 36 37 38 39 40 41 42 43 44 45
Sep-Oct  29 30 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12

Egg       e  e  e  e  e
Instar 1  ..... 1  1  1
Instar 2 . ..... 2  2  2
Instar 3 .. ..... 3  3  3
Instar 4 ... ..... 4  4  4
Instar 5 .... ..... 5  5  5  5
Instar 6 ..... ..... 6  6  6  6  6  6  6
Pupa      ..... ..... P  P  P  P  P  P  P
Adult     ..... ..... Adult male
```

[Dated comments, photo frames.]

4. *When larvae of doubtful stadium are collected it is convenient to use lower case letters to represent stadia, assigning "a" to the earliest instars. Use single capital letters for such terms as "pupa," "tachinid larva" and "wasp larva." The dated comments will serve to clarify the abbreviations.*

Lep. Family: *Nymphalidae* species: *Anartia fatima* Det. by: *A. Aiello* Lot # *Aiello 99-021*
Collected as: *larvae* Date: *29 Sept 1999* By: *A. Aiello* Locality: *PANAMA, Prov. Panama, Balboa, Ancon*
Hostplant Order: *Scrophulariales* Family: *Acanthaceae* species: *Blechum brownei* Det. by: *A. Aiello*
Parasitoid Order: *Diptera* Family: *Tachinidae* species: *undetermined* Det. by:

```
Duration 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35 36 37 38 39 40 41 42 43 44 45
Sep-Oct  29 30 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12

Indiv 1   a  a  b  b  b  b  b  b  P  P  P  P  P  P  P  Adult
Indiv 2   a  a  a  b  b  b  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  T  Fly
```

29 September collected 2 larvae at 5 a.m. by flashlight, eating B. brownei, possibly 4th instars, but uncertain. 5 Oct a tachinid larva emerged from one caterpillar.

[Continue dated comments, photo frames.]

5. *This steplike format is easy to read, but it can be used only when rearing a single individual. The instar numbers go on the left, and are also recorded daily.*

Lep. Family: *Nymphalidae* species: *Anartia fatima* Det. by: *A. Aiello* Lot # *Aiello 99-019*
Collected as: *larvae* Date: *9 Sept 1999* By: *A. Aiello* Locality: *PANAMA, Prov. Panama, Balboa, Ancon*
Hostplant Order: *Scrophulariales* Family: *Acanthaceae* species: *Blechum brownei* Det. by: *A. Aiello*
Parasitoid Order: Family: species: Det. by:

```
Duration 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 32 33 34 35 36 37 38 39 40 41 42 43 44 45
Sep-Oct  29 30 01 02 03 04 05 06 07 08 09 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25 26 27 28 29 30 31 01 02 03 04 05 06 07 08 09 10 11 12

Egg       e  e  e  e  e
Instar 1  ..... 1  1  1
Instar 2 . ..... 2  2  2
Instar 3 .. ..... 3  3  3
Instar 4 ... ..... 4  4  4
Instar 5 .... ..... 5  5  5  5
Instar 6 ..... ..... 6  6  6  6  6  6  6
Pupa     ..... ..... P  P  P  P  P  P  P
Adult    ..... ..... Adult male
```

[Dated comments, photo frames.]

6. For those who prefer to keep notes by hand, rather than on a computer, a form such as this can be typed and drawn up for xeroxing.

Lep. Family: species: Det. by: Lot #

Collected as: Date: By: Locality:

Hostplant Order: Family: species: Det. by:

Parasitoid Order: Family: species: Det. by:

Duration	01	02	03	04	05	06	07	08	09	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24	25	26	27	28	29	30	31

Notes: Photo Date Frame

Appendix B

The Write Stuff: Preparing Manuscripts for Publication

Boyce A. Drummond

If you take time to make careful observations or perform experiments on Lepidoptera, the data collected and conclusions drawn from your studies are much more valuable if published. Publication makes your work available to others who share your interests and would benefit from your findings. The following suggestions are intended to introduce you to the format required for publishing your scientific work.

Scientific writing is characterized most of all by being concise and to the point. By eliminating unnecessary words and phrases in your paper you (1) reduce the time required for reading it (which allows the reader to extract the important information more quickly) and (2) save money (many journals require you or your institution to pay for each page of print required by your article).

Most journals require manuscripts to adhere to strict guidelines governing the organization, punctuation, and spatial arrangement of text. For details, consult the "Notice to Contributors" published in each issue of the journal to which you plan to submit. The Journal of the Lepidopterists' Society *uses the format presented below.*

1. Title

The title should be brief but descriptive of the content of your paper. Careful choice of words is important, because abstracting services use words in the title to compile their subject indices. (A set of up to five "additional key words" should be provided to further facilitate bibliographic retrieval.)

2. Abstract

Briefly summarize your findings. The abstract should be in complete sentences and report what you did, found, and concluded. Many journal subscribers will have time to read only a few papers in a given issue, but will read most of the abstracts.

3. Introduction

The introduction tells why you did the experiment. This often involves a brief review of previously published studies to put your experiment into historic and scientific perspective. Outline the nature of the problem under investigation and the objectives of your observations or experiments. Be sure to state the hypothesis or hypotheses you are testing. Although the introduction should be brief and to the point, it must adequately prepare the reader for what is to come.

4. Materials and Methods

If the introduction answers the question "Why?", the materials and methods section should answer the questions "What?, How?, Where?, and When?" Briefly describe the techniques and equipment used in your study; standard or commonly used methods or instruments need not be described in detail. This section should always state the number of samples, specimens, or

measurements taken so that the basis for the results is known. Be sure to give the locality, dates, times and conditions of the experiments or observations. Be brief with this section, but remember that enough information must be given to allow someone else to duplicate your methodology.

5. Results

The results section should contain only what the heading states—results; no interpretation should be included. Results should be presented in one of two ways, depending on the type and amount of data. First, the data may be incorporated into the text. For example, to report a single observation it may be best to state simply *"The mated pair, perched on a blade of grass 10 cm above the ground, remained motionless for 47 min., after which the female flew out of sight to the northwest while the male continued to perch for another 30 min."* Second, if you have a body of similar data, (e.g., comparing forewing lengths to body weight for several species of moths), incorporate it into a table or graph (see examples in current issues of the *Journal).*

6. Discussion

In this section pull together your results, draw conclusions, and point out the significance of your data. State whether your experiment disproved or supported your hypothesis. Indicate the limitations of the available data as well as the general implications. Explore the ramifications of your results: how has knowledge of the field changed as a result of your research? This is an opportunity to exercise your imagination, tempered with logic, to produce well-grounded speculation and generate testable hypotheses for future studies.

7. Literature Cited

When you refer to an idea or data taken from an article or book, you must give credit to the original authors by citing the work from which you obtained the ideas or facts. In this section, list in alphabetical order all published works cited in your paper (see recent issues of the *Journal* for format and style). In the text, references should be cited in either of the following ways: *"Pliske (1972) reported a positive correlation between female age and mating frequency in butterflies."* or *"Mating frequency in females is a function of age (Pliske 1972)."*

This bare bones outline of a scientific paper provides none of the "flesh" that can make a journal article interesting and exciting to read. For further information, consult:

For narrative writing:

Strunk W Jr. & EB White 1979. The elements of style, 3rd ed. New York: MacMillan Pub. Co.

Zinsser WK 1994. On writing well: An informal guide to writing nonfiction, 5th ed. New York: Harper Perennial.

For specific advice on matters of style and format:

Grossman J 1993. The Chicago manual of style: The essential guide for writers, editors, and publishers, 14th ed. Chicago: University of Chicago Press.

Style Manual Committee, Council of Biology Editors, Inc. 1994. Scientific style and format: The CBE manual for authors, editors, and publishers, 6th ed. New York: Cambridge University Press.

For specific instruction on writing scientific papers:

Alley M 1987. The craft of scientific writing. Englewood Cliffs, NJ: Prentice-Hall Inc.

Day RA 1988. How to write and publish a scientific paper. Phoenix: Oryx Press.

Katz MJ 1985. Elements of the scientific paper: Step-by-step guide for students and professionals. New Haven, CT: Yale Univ. Press.

Appendix C

How to Find an Original Description: Lepidoptera

J. Donahue and K. Donahue

(Reprinted with permission from News Lepid Soc 1989 No. 6, p. 82–83.)

Finding an original description, or any other publication on Lepidoptera, can often be a very frustrating and time consuming process. Having access to a good reference library, or good working relations with a colleague who does, greatly reduces the search time. Searching for a reference by randomly searching the literature can waste a lot of time, but may ultimately be the only way to find a particular obscure reference. In most cases, however, assuming you have access to all the references listed, following the sequential steps listed here should result in the most rapid "hit rate."

Note: the most important key to the literature is the name of the author of the taxon (genus or species) and the year in which the name was published. With this information, the Zoological Record *usually quickly yields a citation of the original description, and should be the reference of choice. Table 1, on the next page, gives a graded approach to locating an original description.*

Publications useful for locating references to older literature:

Derksen, W & U Scheiding 1963-1968.
Index Litteraturae Entomologicae, Serie II. Die Weltliteratur ueber die gesamte Entomologie von 1864–1900, 4 vols. Deutsche Akademie der Landwirtschaftswissenschaften zu Berlin. Vol. 1 (1963), A–E; vol. 2 (1965), F–L; vol. 3 (1968), M–R; vol. 4 (1968?), S–Z [A world bibliography of entomological publications from 1864 to 1900, to complement the two earlier bibliographies of Hagen and Horn & Schenkling (below).]

Hagen, HA, 1862–1863.
Bibliotheca Entomologica: die Litterature ueber das ganze Gebiet der Entomologie bis zum Jahre 1862, 2 vols. Leipzig. [vol. 1, 1862, authors A–M; vol. 2, authors N–Z]. [A bibliography of virtually all entomological works published up to and including 1862. A reprint edition is available.]

Horn, W & S Schenkling 1928–1929.
Index Litteraturae Entomologicae, Serie I: Die Weltliterature ueber die gesamte Entomologie bis inklusive 1863. [Covers the same territiory as Hagen (above), with bibliographic refinements.]

Sources:

Neave, Nomenclator Zoologicus (7 vols., including supplements):
Zoological Society of London, Regent's Park, London NW1 4RY, England.

Zoological Record, Insecta, Part D: Lepidoptera.
Paper copy available from Biosis, 2100 Arch Street, Philadelphia, PA 19103. *Volume 115 (1978) to present is available on-line through* Dialog *Information Retrieval Services, Inc., 3460 Hillview Avenue, Palo Alto, California 94304.* Dialog *maintains and constantly updates a substantial number of computer databases which may be accessed by telephone and a modem from a terminal or personal computer. The fee is based on connection time.* Dialog *training is available.*

Table 1. The route to an original description.

Information one has:	***References to consult:***
1. Any amount	*Someone who knows or who can find out (such as a friendly curator with access to a good library). [Caution: although this technique is easiest for the questioner, your source will have to do all the legwork outlined below; be prepared to return the favor if you want to cultivate a continuing relationship.]*
2. Name, author, year (genus or species)	Zoological Record *(for all the names published 1864 to present);* Berichte ueber Ent. *(for names published 1834 to 1863);* *Sherborn,* Index animalium *(1801–1850).*
3. Generic name (family known or unknown)	*Any animal:* *Neave,* Nomenclator zoologicus. *Butterfly:* *Heming,* Generic names of butterflies; *Kirby,* Synonymical Catalog of diurnal Lepidoptera; *Beattie,* Rhopalocera directory. *Moth:* Generic names of moths of the world; *Kirby,* Syn. Cat. Lepidoptera Heterocera (part).
4. Generic name (family known)	*In addition to references in #2* Genera Insectorum *(not complete);* Lepidoptorum Catalogus *(nearly complete).* *Also worldwide family catalogs and monographs, such as Bleszinski,* Crambinae; *Rothschild & Jordan,* Sphingidae; *Hampson,* Noctuidae and Arctiidae *(incl. Ctenuchidae); Evans,* Hesperiidae of the World *(four regional works).* *Also regional catalogues, such as Hodges,* Checklist of Lepidoptera of America north of Mex.*;* *Staudinger & Rebel,* Cat. Palearctischen Lepidopteren.
5. Species known (family known or unknown)	*Butterfly:* *Beattie,* Rhopalocera Directory.
6. Species known (family known)	Lepidoptorum Catalogus; *Any appropriate world and regional catalogs (#3 above);* *Hesperiidae: Bridges,* Notes on Species-group Names; *Lycaenidae, Riodinidae:* List of Original Descriptions.
(Region also known)	*Seitz,* Macrolepidoptera of the World *(incomplete).*

Appendix D

Searching for Papaipema *Larvae*

(From S. A. Hessel, A guide to collecting the plant-boring larvae of the genus Papaipema *(Noctuidae), Lepid News 8:57–63, 1954)*

Hessel (1954) provided the following information on larvae in a "readily referable form in order to stimulate members to ... study the fascinating and beautiful North American Noctuid genus *Papaipema* Grote." E. Quinter (pers. comm.) has since found ten new species (not included here), and emphasizes that he has not had the opportunity to search in western North America, where many may remain to be discovered. Colored plates of most adults can be found in Holland (1958), Rockburne and Lafontaine (1976), Covell (1984), or Rings at al. (1992); some species names are obsolete (references in this appendix are listed in Appendix J: Books). The date ranges in the table are too narrow: pupation in the South is much earlier, and flight periods can extend to December in the Gulf states. Quinter states that, rather than late July, late June is the time to begin to search for plant damage and larvae, and as early as April in the South (see Table 1, on the next page).

Collecting equipment includes a narrow trowel with edges sharpened, a stout pocket knife, and containers such as Ziploc bags in which the host plant with its burrow can be kept fresh; moist sphagnum packing can help. Contrary to a statement by Hessel, Quinter finds that a larva that leaves its burrow can be reintroduced if you are persistent enough. If it is difficult to keep the larval foodplant in fresh condition, he recommends substituting roots such as carrot or sweet potato.

Note the following pointers from Hessel:

> "Parasites, too, take their toll, but these should be considered prizes reared out carefully, and passed along to competent authorities."
>
> "For those species pupating in burrows care must be taken that drying and shrinking plant tissues do not pinch or crush the pupa. The latter may be removed for safety and placed on a moist sphagnum bed or, if the plant is maintained in fresh condition, they may, of course, be left undisturbed."
>
> "Be alert for anything queer about the condition of a plant, look for further clues, close in on the culprit, and sentence him to life imprisonment; and don't forget that notebook. Besides a contribution to lepidopterology it is likely that you will find the work the most fascinating form of 'who-dunit' you have yet experienced."

Eggs are generally dropped loose in the vicinity of the foodplant in the fall (Forbes 1954).

REFERENCES

Forbes WTM 1954. Lepidoptera of New York and neighboring states, Part III. Noctuidae. Ithaca, NY: Cornell University Agricultural Experiment Station Memoir 329.

Hessel SA 1954. A guide to collecting the plant-boring larvae of the genus *Papaipema* (Noctuidae). Lepid News 8: 57–63.

Table 1. Papaipema *larval and pupal situations.*

Species	Food plant	Evidence of presence	Position of: larva	Position of: pupa	Date the species: pupates	Date the species: emerges
*P. aerata**	Burdock (*Arctium*)	Wilted branch; native foodplant unknown*	Lower stem	In burrow at times*	Jul 15	Aug 24–Sep 6
*P. appassionata**	Pitcher plants (all *Sarracenia*)	Orange frass	Root; to new root in July	Root, or soil near plant	Aug 1–10	Sep 15
P. araliae	Hercules club (*Aralia spinosa*)	Bored new growth; withered leaves; dead branch	Stem tip; new growth	Soil	Aug 10	Oct 1
P. arctivorens	Thistle (*Cirsium* spp.); burdock (*Arctium lappa*)	Thistle: branching below crown, or crown black; frass	Stem	Soil	Jul 20–Aug 20	Aug 5–Oct 5
*P. astuta**	Horse-balm, stoneroot (*Collinsonia canadensis*)	Dry stem; white frass	Stem; later cell in root	Soil	Aug 1–10	Sep 12–24
*P. baptisiae**	Wild indigo (*Baptisia tinctoria, B. alba*), Indian plantain (*Cacalia tuberosus*), dogbane (*Apocynum*)	Discolored foliage; frass; holes in stem; sometimes fallen plant	High in stem, to upper root	Soil*	Aug 1–Sep 10	Aug 16–Oct 16
*P. beeriana**	Snakeroot (*Liatris* spp.)*	Wilted tip, or brown and dry leaves	Lower stem, root crown	Burrow or near root	Aug 10–Sep 15	Sep 6–Oct 16
P. birdi	Water hemlock (*Cicuta maculata*), water parsnip (*Sium suave*), other umbellates	Bending or fallen top or branch at point of entry	1m from ground, to root	Soil, 4–20" from plant, 1–2" deep	Late July–Sep 5	Early Aug–Oct 16
*P. cataphracta**	General feeder, esp. burdock, thistles, lilies*	Plants stunted, drooping, discolored; swelling; much frass	Stem; at maturity, near ground	Stem, sometimes root	Aug 10–Sep 25	Sep 8–Oct 15
P. cerina	Turk's cap (*Lilium superbum*) et al.†	Small hole; drying or dry stem; little frass	Stem	Burrow	Aug 5–25	Aug 30–Oct 14
P. cerussata	Ironweed (*Vernonia* spp.)*	Plant stunted, broken, or bent at larval entrance; top may be much branched from later boring below	Stem (early), to root	Soil, 2–12" from root	Aug 10	Sep 5–15

* Emendations by E. Quinter, 1995-97.

† Small larvae have been found in starry campion (*Silene stellata*), may apple (*Podophyllum peltatum*) feeding upward, and quite commonly in bottle-brush grass (*Hystrix patula*); will mature in iris but require several stems; will not enter root. — Wyatt.

Table 1. Papaipema *larval and pupal situations.* (continued)

Species	Food plant	Evidence of presence	Position of: larva	Position of: pupa	Date the species: pupates	Date the species: emerges
P. duovata *	Seaside and late goldenrod (*Solidago sempervirens, S. gigantea leiophilla*)	Several openings in stem; often whitish frass on sand	Stem (early), to root	Burrow	Aug 15	Sep 15–Oct 20
P. duplicata	Horse-balm, stoneroot (*Collinsonia canadensis*)	Wilted leaves on dry stem; sometimes white frass	Stem (early), to root	Root	Aug 15	Sep 30–Oct 28
P. eryngii	Button snakeroot (*Eryngium aquaticum, E. yuccifolium*)	Little or none; yellow or dead leaf; bored	Leaf or stem (early), to root	Burrow in root	Aug 15–30	Sep 10–Oct 15
P. eupatorii	Joe-Pye weed (*Eupatorium purpureum*)	Leaning stem usually still living; sometimes frass	Stem to root	Burrow at base	Aug 5–Sep 5	Sep 12–Oct 5
P. furcata	Red, white, and black ash (*Fraxinus pennsylvanicus, F. americana, F. nigra,* probably all spp.)*	Dry branch or blackened tip of shoot; clean hole to later burrow in older growth	First in new growth, later in near wood	Soil	Jul 30–Aug 25	Aug 24–Sep 20
P. harrisii *	Cow parsnip (*Heracleum maximum*), angelica (*Angelica atropurpurea, A. lanatum*)	Yellow or wilted leaf in *Heracleum*, drooping stem in *A. lanatum*	Leaf stem to root crown	Soil	Jul 15–22	Aug 8–Sep 25
P. humuli *	Hop (*Humulus lupulus*)	Cigar-shaped gall	Stem	Soil	(Retrieve larva July 20)	Aug 15
P. impecuniosa	Aster (*Aster puniceus, A. umbellata*), sneezeweed (*Helenium autumnale*)*	Large opening for moth	Lower stem to root	Base of stem or root	Aug 15–Sep 10	Sep 10–Oct 15
P. inquaesita *	Sensitive fern (*Onoclea sensibilis*)	Yellow, brown to dry stem, hole at entry, orange frass	Stem (early), to root	Root	Jul 20–Aug 15	Sep 1–Oct 1
P. leucostigma *	Columbine (*Aquilegia canadensis*, other spp.)	Wilted plant, frass	Root (often needs two)	Soil	Jul 15–Aug 15	Sep 1–19
P. lysimachiae	Loosestrife (*Lysimachia quadrifolia, L. terrestris*)*	Yellow, brown or dry stem	Stem (early), to root	Soil, 2–15" from plant, 1/2–3" deep	Early Aug–Sep 15	Sep 1–Oct 1
(*P. marginidens*)*		(Removed, biology unknown)*				
P. maritima	Giant sunflower (*Helianthus* spp.)*	Frass and gall-like swelling; stem may be broken at top of burrow; pupa near hole	Base of stalk	Base of stalk (in gall)	Aug 15–Sep 2	Sep 5–Oct 20

Table 1. Papaipema *larval and pupal situations.* (continued)

Species	Food plant	Evidence of presence	Position of: larva	Position of: pupa	Date the species: pupates	Date the species: emerges
*P. nebris**	General feeder, esp. ragweed (*Ambrosia artemisiifolia*), burdock (*Arctium*)	Swellings or galls; holes in stem	Lower stem	Burrow or soil	Aug 10–mid-Sep	Sep 5–Nov 10
*P. necopina**	Sunflower (*Helianthus* spp.), Indian plantain (*Cacalia tuberosus*)*	Elongate enlargement or gall at base	Stem to root	Root burrow or soil	Aug 5–30	Sep 9–20
P. nelita	Tall coneflower (*Rudbeckia laciniata, Arctium* spp.)*	Gall	Base or root	Soil	Jul 20	Sep 1–15
*P. nepheleptena**	Turtlehead (*Chelone glabra*)	White frass	Stem	Soil	Aug 10–15	Oct 1
P. polymniae	Leafcup (*Polymnia uvedalia*)	Irregular swelling	Mid-stem to base	Soil	Aug 5–10	Sep 2–23
*P. pterisii**	Common brake (*Pteridium aquilinum*)	Yellow or brown frond, orange frass	Stem to upper root	Soil near root	Jul 25–Aug 15	Aug 7–30
P. rigida	Golden alexanders (*Zizia aurea*), oxeye (*Heliopsis helianthoides*), sunflower (*Helianthus* spp.)*	Holes in stem near base, slight swelling, frass	Stem (early), to root	Soil	Jul 25–Aug 20	Aug 30–Oct 11
*P. rutila**	May apple, mandrake (*Podophyllum peltatum*)	Yellow leaf and much frass	Root	Soil	Aug 10–Sep 15	Sep 7–Oct 5
P. sciata	Speedwell (*Veronicastrum virginicum*)	Dry, black dead stem, sometimes broken; frass	Long roots	Soil	Aug 10–Sep 10	Sep 10–Oct 10
P. silphii	Prairie dock, rosinweed, cup-plant (*Silphium* spp.)	Brown leaf or two, frass	Root	Soil	Aug 1–30	Sep 16–Oct 20
P. speciosissima	Cinnamon, interrupted fern (*Osmunda cinnamomea, O. claytoniana*), but esp. royal fern (*O. regalis*)*	Yellow (June) to dry (Aug); frond bends as larva matures; frass	Stem through root stock	Burrow or fibers of root	Aug 10	Aug 25–early Oct
*P. stenocelis**	Chain-fern *(Woodwardia virginica)**	Brownish to dry frond, orange frass	Stem (near tip of frond) to root	Soil	Aug 15–Sep 15	Sep 10–Oct 10
*P. unimoda**	*Meadow rue (*Thalictrum* spp.)	Slightly dwarfed plant; stem yellow or bending; hollow stem always blackened	Tip of stem (early), to root	Soil, 1–15" from plant, 1/2–3" deep	Aug 10–Sep 15	Sep 1–Oct 10

* Emendations by E. Quinter, 1995-97.

Appendix E

Lepidoptera Philately

Charles V. Covell Jr.

If you like collecting but for some reason do not wish to collect butterfly and moth specimens, you might consider "philatelic lepidopterology." This is collecting stamps and related postal items that depict Lepidoptera. There are several thousand issued by many countries. Some are or were valid for postage, and are considered true postage stamps. Others have been printed just for collectors, and are known as "Cinderellas." Many of these appear to be canceled (have rubber stamp markings), but such postmarks are usually a portion of a circular cancel in one corner. These are called CTOs, meaning that the stamps have been "canceled to order." The cancels were put on for collectors and did not result from true postal usage. While these are collectibles, they are usually avoided by those collecting mint (unused) or truly "postally used" stamps, because they are "contrived" for collectors.

Figure 1. Issued by Hawaii in 1890, showing a butterfly barrette in the hair of Queen Liliuokalani.

Besides mint and used stamps there are other postal items related to Lepidoptera that philatelists also seek. These include special cancellations ("cancels") with butterfly or moth pictures or words (some from Pacific Grove, CA, where the Monarch Festival is held annually); postal meters with references or illustrations of Lepidoptera (like "Caterpillar tractors"); corner cards (return addresses from Lepidoptera-related businesses or organizations, with or without Lepidoptera pictures, such as the Lepidopterists' Society); essays (artwork submitted for possible or actual use in producing stamps); proofs (sample printings to test images or colors); "EFOs" (errors, freaks and oddities, such as the US 1977 butterfly stamps with perforations through their middle); commercial covers (any envelopes or pieces of package covering with used Lepidoptera stamps); FDCs (first day covers) of Lepidoptera issues; postal stationery (envelopes or postal cards with Lepidoptera related "stamps" printed by postal services, such as air letters issued by Belize and postal cards issued by Poland); and cancels from places with Lepidoptera names such as "Butterfly, KY," "Butterfly, NY," and "Mariposa, CA."

Figure 2. Sarawak 1-cent issue of 1950, depicting the Rajah Brooke birdwing, Trogonoptera brookiana.

So, where can you start your collection of paper Lepidoptera? I suggest purchasing a copy of *Linn's Stamp News* at your book store, and taking out a year's subscription. In it you will find ads for dealers who sell topicals—stamps dealing with such topics as butterflies and moths. Next, join the ATA (American Topical Association, PO Box 50820, Albuquerque, NM 87181-0820 USA), made up of philatelists specializing in topical philately, including insects. Their annual surveys typically

show butterflies and moths to be the 8th or 9th most popular topics. There are lots of dealers offering material, and they have ads in *Topical Time*, the bimonthly magazine of the ATA. Finally, join the PLA – Philatelic Lepidopterists Association (C.V. Covell Jr., Treasurer, 2333 Brighton Drive, Louisville, KY 40205-3023 USA) and see articles on butterfly and moth philately, and make contact with other collectors of this theme. Ads for exchanging, buying and selling philatelic Lepidoptera items are included in the quarterly newsletter, *The Philatelic Aurelian*. New issues are listed there to inform you of "what's new."

Figure 3. Old World swallowtail, Baltimore checkerspot, California dogface, and falcate orange tip, issued in 1977.

Figure 4. The luna moth, eastern tiger swallowtail, and monarch, issued in 1983.

Before long you will have the oldest butterfly stamps (Figure 1), and the first stamp showing an identifiable butterfly (Figure 2).

From there the sky's the limit, with more than 1000 identifiable butterflies and moths on stamps, and more issued each year. So far the United States has issued seven Lepidoptera stamps: four butterfly stamps in 1977 (Figure 3), and two more, plus a luna moth in 1983 (Figure 4). Most butterfly and moth stamps are available and inexpensive.

Happy hunting!

Appendix F

A Technique for Setting and Mounting Microlepidoptera

Jean-François Landry and Bernard Landry
(Reprinted with permission from J. Lepid Soc 48:205-227, 1994.)

During the past 75 years, several papers have presented, with various amounts of detail, techniques for preparing (pinning, setting, and mounting) microlepidoptera (e.g., Kearfott 1904, Calmbach 1921, Lhomme 1926, 1927a, 1927b, Amsel 1935, Holland 1937:19, Janse 1939, Janmoulle 1943, Charlson 1945, Lindquist 1956, Hodges 1958, Lewis 1965, Tagestad 1974, Zimmerman 1978, Sokoloff 1980, Mikkola 1986). However, our contacts with many lepidopterists indicate that, at least in North America, good and simple techniques for preparing microlepidoptera are not well known. In fact, many North American lepidopterists do not even collect microlepidoptera as routinely as other Lepidoptera, in part because of the perceived inconvenience of preparing them. Microlepidoptera that are collected are often only the larger specimens, in groups such as pyraloids, tortricoids, and large gelechioids.

The paucity of good-quality microlepidoptera from North America in many collections is one of the causes for the very slow progress in systematic studies of the Nearctic fauna. Our knowledge of the taxonomy and faunistics of many families of microlepidoptera is shockingly poor. A plea recently has been made for North American lepidopterists to take on the collection and study of microlepidoptera (De Benedictis 1993). Of course the first step in this endeavour is to acquire a good and efficient technique for preparing specimens.

There are probably nearly as many ways of preparing microlepidoptera as there are individuals collecting them. The basic method of spreading microlepidoptera is the same as for larger Lepidoptera. However, some adjustments, both at the time of collecting and of preparation, and in the equipment, are needed because of the small size and fragility of microlepidoptera.

Over the years we have tried every different method and variation of preparation for microlepidoptera that we came to know. While most techniques can yield high-quality specimens, many suffer from being relatively slow or requiring somewhat cumbersome equipment (e.g., spreading boards) ill-suited for prolonged field work under difficult conditions. We sought to develop a technique that offsets these problems, i.e. one that is rapid, usable under any condition with equal efficiency, yet versatile with respect to the quality of preparation desired by the collector. An earlier version of the technique described here was published in French by Landry (1991) but we have modified it slightly, with some additions.

Our method actually combines elements from other methods employed by microlepidopterists, with added refinements. It is based on the concept of setting microlepidoptera on the bottom of a box, which can be traced back at least to Amsel (1935). Modern materials, especially dense polyethylene foam, dramatically enhance the results of Amsel's method. Partial spreading in such boxes is now used by many microlepidopterists on collecting trips (Zimmerman 1978:50-59, Nielsen 1980). The main shortcoming of partial spreading is that special specimens, such as types of new species or those needed for photography, may need subsequent relaxation for final spreading. The technique exposed here offers the possibility of a full range of quality of preparations, from unspread to fully spread with as much care as a perfectionist may wish, all with the same equipment and with hardly any extra time. The technique may be used in the field, in the lab, or at home. The necessary equipment is very compact, lightweight, inexpensive, and easily made. We have tested the method with tens of

thousands of microlepidoptera over the past few years, under conditions varying from local, day trips to month-long expeditions in the tropics (including camping).

In addition to the actual technique of setting microlepidoptera, we offer some suggestions for handling specimens when they are collected in the field, and for staging (double-mounting) spread specimens. Appropriate handling of collected microlepidoptera is as critical as the actual setting in obtaining high-quality specimens, and so is the final staging to insure safe preservation in subsequent handling.

Collecting

The facility and rapidity of the technique outlined here rests on working with the freshest specimens possible. Moths are placed individually in glass vials upon collecting and kept alive until the time of pinning and setting. Upon returning from the field, vials are stored in a cool, dark place if the specimens cannot be prepared immediately. The ideal place is the refrigerator, or a cooler box if one is on a prolonged field trip. We have been able to keep specimens alive for up to five days in this manner, although we recommend delaying as little as possible (some moths will begin to show some wear even after 1-2 days in the refrigerator). Refrigeration is particularly useful if one has had a large catch on one day and there is not enough time to prepare all specimens immediately after they have been collected. We recommend preparing the smallest microlepidoptera as soon as possible, as they will die more quickly from dehydration. Once dead, small moths tend to dry very quickly and become difficult to relax and spread. In the humid tropics, small microlepidoptera will dehydrate quickly inside vials (often in just a few hours) and are best set as soon as possible. Always begin by preparing the smallest specimens first, working up to larger ones. Refrigeration, even if available, should probably not be used for tropical microlepidoptera from those regions that hardly ever experience temperatures below 10°C, because the relative cold will kill many of them.

Vials. Collecting vials should preferably be made of glass and closed with an easily removable stopper that can be opened with a single hand (the other may be busy holding a net). We use glass vials that are 65 mm long and 19 mm in diameter, closed with a rubber stopper. Stoppers should be as little wedge-shaped as possible, otherwise smaller microlepidoptera will crawl in the space between the stopper and the vial neck and damage themselves. We carry about 100 vials for most daytime collecting, at least twice as many for nighttime collecting at a light. Experience will dictate the adequate supply. During daytime collecting, care must be taken that the vials are not exposed directly to or heated by sunlight, otherwise the moths will quickly die and dry. If possible, avoid plastic vials (snap-cap type), especially with the smaller specimens, because the static charge that such vials accumulate through handling and friction will damage the squamous cover of the moths and increase the rate of wear.

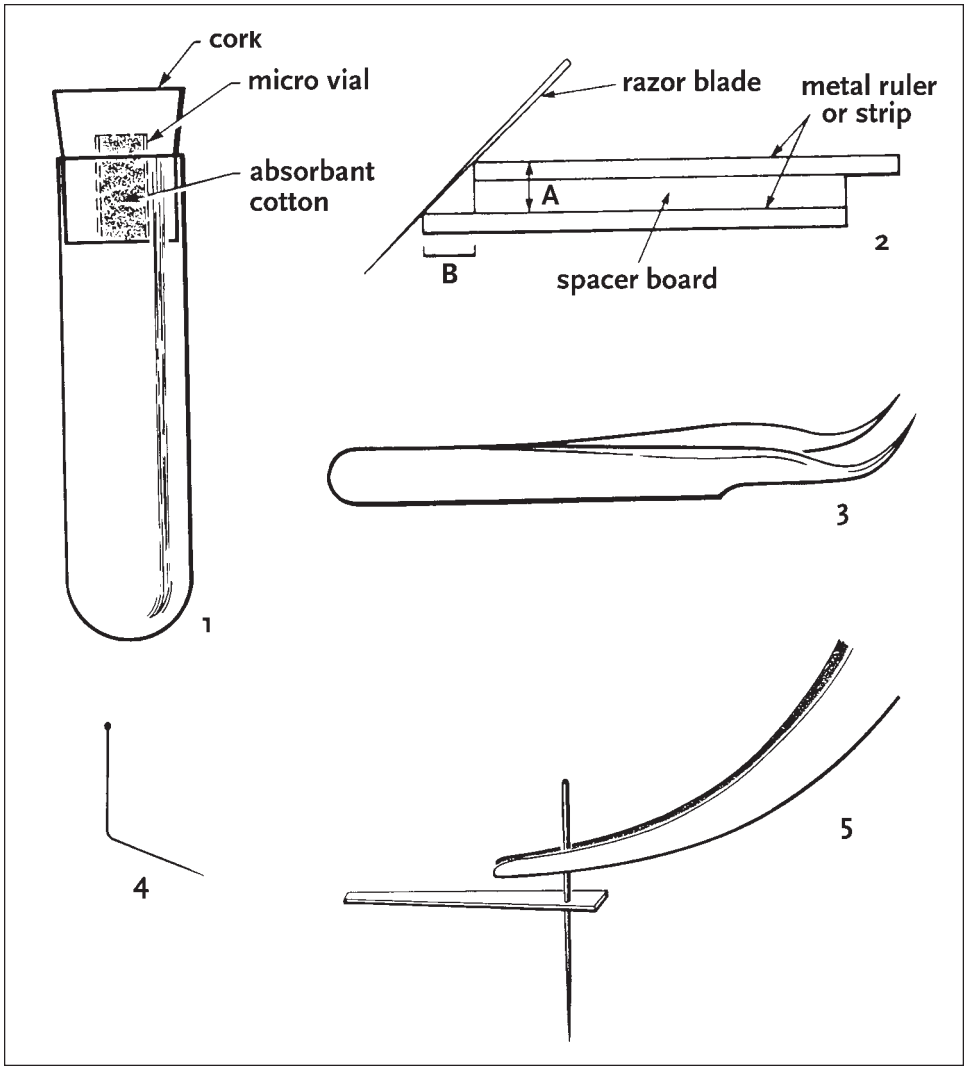
A word of caution is necessary if one is setting reared specimens: never set a freshly emerged moth. Allow at least 24 hours (longer if a genitalia dissection may be required) for the moth to harden sufficiently. Without this precaution, wings may curl, crumple, or droop after removal from the setting box, and if the genitalia are later dissected, structures will be insufficiently sclerotized and difficult to prepare adequately.

Poison and Killing Tubes. The choice of poison is, of course, a matter of personal preference, availability, etc. We strongly recommend ammonia (ammonium hydroxide): it has a quick knockdown action and leaves freshly killed specimens beautifully relaxed and ready to be spread immediately with the greatest ease. We have tried other killing agents and methods, but ammonia is the one that has given us the best results. The ammonium hydroxide solution should be as concentrated as possible. A laboratory-grade solution containing about 30% ammonia and 70% water is preferable because it has a very fast knockdown action. Household

ammonia, generally a murky liquid, is weaker and unsuitable.

For killing tubes, we use glass tubes closed with cork stoppers into which a small microvial is inserted, loosely stuffed with cotton (Figure 1).

Five to ten minutes before using a tube, the cotton is imbibed with a few drops of ammonia solution, and the tube closed to let the ammonia concentration rise. This type of killing tube offers nothing inside against which struggling moths may rub; the disadvantage is that the tubes need to be recharged more frequently, approximately once every 2-3 hours of continuous



Figures 1-5. Materials required for preparing microlepidoptera. 1, Killing tube; 2, superimposed, offset rulers to cut symmetrical V-shaped grooves, A = B for 45° grooves; 3, curved forceps used to handle minutens; 4, bent standard pin used to assist pinning and spreading; 5, card triangle mounted on shortened pin used to hold set wing in the point method.

use (when opened several times periodically). When setting large numbers of microlepidoptera, we use up to 10 tubes at a time to minimize recharging, and place only 2-3 moths per tube at a time. It is essential to check for and wipe traces of moisture or sweating on the walls of the killing tubes. Charged tubes may be laid on their side to prevent any ammonia from possibly running down the sides, although this will not be a problem if a modest quantity is used. When tubes are not in use for more than a day or so, it is preferable to leave them open and remove the cotton swab from the stopper to allow then to dry thoroughly.

Ammonia has a few disadvantages: it tends to sweat in a tube if an excessive quantity is used or if it is too warm (tubes must not be exposed to heat or direct sunlight) — but this is a disadvantage common to most liquid poisons; it loses strength relatively rapidly in a frequently open tube; and fumes are choking, irritating. Weak ammonia must not be used for moths with green, red, or orange pigments because the long exposure needed to kill them may cause discoloration. If the ammonia is strong though, this is not a problem providing that the moths are removed as soon as they are dead. In case of doubt about possible discoloration, one should use another poison, preferably ethyl acetate (subsequent relaxation may be necessary). With strong, concentrated ammonia, we have not had discoloration problems. Generally we have found that the advantages of ammonia far outweighed its disadvantages, none of which presented a real problem if it was used with the precautions outlined above, and that it was no more inconvenient to use than any other poison.

Recently we have experimented with a solid form of ammonia, ammonium carbonate, a salt with the appearance of cyanide crystals. Upon contact with the ambient humidity, the crystals decompose into gaseous ammonia, carbon dioxide and water vapor (Gilligan and Gilligan 1990). Killing tubes are made simply by packing a 1-2 cm thick layer of crystals in the bottom and covering them with a smooth, porous material [e.g. artificial foam sponge (Gilligan and Gilligan 1990)]. We used plastic caps (from snap-cap vials) punctured with many minute pin holes to cover the crystals. Plaster cannot be used because the water it contains will instantly dissolve all the crystals and produce all the ammonia at once. We obtained satisfactory results with ammonium carbonate if used for small numbers of specimens. Disadvantages are that the rapidity of killing decreases markedly compared to liquid ammonia if one opens the tubes frequently; also if there are too many specimens in a tube and it is warm, the moisture content may rise to the point where, upon cooling, crystals may form on the specimens; such crystals are then very difficult to remove. For these reasons we find ammonium carbonate less satisfactory than ammonium hydroxide.

Ethyl acetate also works well but we found that it has a tendency to stiffen many microlepidoptera if they are left in the killing tube a few minutes too long; hence, some relaxation is sometimes necessary. Like ammonia, it is volatile, and tubes need frequent recharging and may "sweat" if heated. It is also flammable and will dissolve some plastics. Generally, we have found ethyl acetate to be less satisfactory than ammonia in quickly producing ready-to-spread specimens.

Killing. Remove the cork, insert one moth, close the cork. Repeat with other tubes. When there is one moth in each tube, start again with the first tube, ensuring that the moth is stunned. Continue until there are 2-3 moths per tube. Stunning takes less than five seconds when the ammonia is strong but may stretch to 10-15 seconds after tubes have been opened several times. Moths should be left in the killing tubes for at least 15 minutes to ensure they are dead. Very small moths (Nepticulidae, small Gracillariidae, for example) can be removed sooner. A time-saving strategy in the subsequent setting operations is to segregate specimens by size at the killing stage. This way, at the setting stage, one does not have to switch back and forth among various spreading boxes with different groove widths.

Setting Equipment

Spreading boxes (Figure 6). We use shallow, clear polystyrene plastic boxes; currently we have two sizes, 11cm X 11cm X 2cm, and 12cm X 8cm X 2cm, obtained from different suppliers. Actual dimensions are not important, as long as boxes are relatively small, preferably shallow (for compactness) with a low-edge lid, rigid, and relatively airtight (or pest-proof).

For a spreading surface we use Plastazote, a dense, smooth polyethylene foam. We found this material best because it affords the following advantages: the surface acquires a small static charge through handling, which helps wings cling slightly and facilitates spreading; it grips the pins firmly and leaves no pin holes; it sustains hardly any wear.

A 1-cm-thick piece of Plastazote is glued inside the lid of a spreading box (we use all-purpose, nontoxic white glue). Gluing the foam inside the lid (using the bottom as lid) eliminates edges to the spreading surface, greatly facilitates work of the hands, and maximizes use of the spreading surface.

Before gluing the foam into the boxes we cut three or four V-shaped grooves with a razor blade. To obtain grooves with perfectly symmetrical sides, we use two metal rulers or strips, with one being taped on top of the other and propped up by a thin board; the edge of the top ruler is offset from the edge of the lower one by a distance equal to that of the ruler + board thickness (Figure 2). To cut, the blade is slanted and abuts both edges. Symmetrical grooves facilitate spreading. We use a series of spreading boxes with various groove widths, these varying from 1-5 mm (2 mm and 3 mm are the most frequently used widths). It is not necessary to have square grooves with vertical sides, as on standard spreading boards. In fact, the sides of V-shaped grooves often provide direct support for the abdomen.

Minuten pins. Use of minuten pins involves subsequent staging or double-mounting, so this is distasteful to many a lepidopterist. Whatever the perceived difficulty, inconvenience, or time factor involved, we emphasize that this is by far the best and safest way of obtaining fine-quality microlepidoptera. Double-mounted specimens can sustain rougher handling without damage and are far less likely to lose their abdomen, a very frequent problem with microlepidoptera that are mounted on fine standard pins (00 or 000), which are very springy. The genitalia are critical for the specific determination of numerous species of microlepidoptera, hence the abdomen must not be lost.

There are different qualities of minutens available on the market. For the best results, and to avoid frustration, one should use the best quality stainless steel minutens. Avoid black-

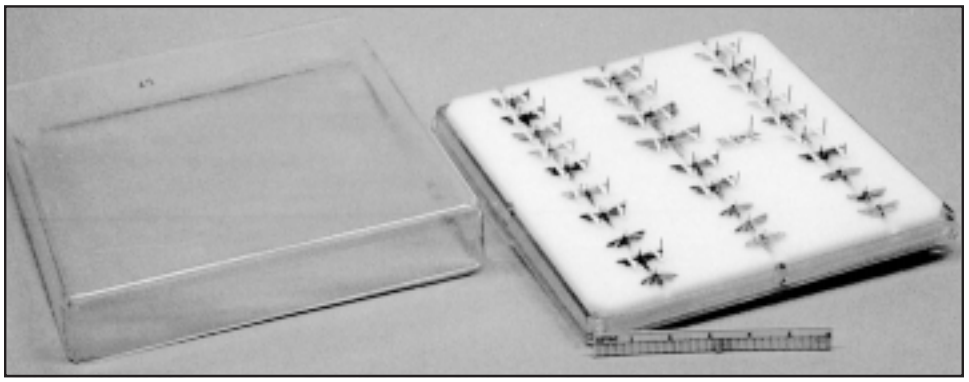


Figure 6. Spreading box. The actual lid is used as bottom on which the Plastazote is glued. Scale in cm.

enameled minutens, which have a tendency to rust (guaranteed if one is in the tropics) and have tips that more easily "hook" (being made of softer metal). The difference in price between stainless steel and black-enameled minutens is small. Diameters of the most useful sizes are 0.20 mm, 0.15 mm, and more rarely 0.10 mm (for nepticulids and other tiny microlepidoptera); some British brands label their minutens A1 (0.14 mm) and B1 (0.19 mm) (1 referring to the shortest length, usually 10-12 mm).

Most minutens are excessively long and must be shortened down to no more than about 1 cm for the larger ones (0.20 mm) or 6-7 mm for the finer ones (0.15 mm and 0.10 mm). If minutens are not shortened, the excess length jutting either above or below the specimens will greatly increase the risk of breakage or damage during handling of the double-mounts (fingers pinching the minuten while grasping the stage-supporting pin will spring the specimen and likely send parts flying, most commonly the weakly-attached, all-precious abdomen). A rapid method of shortening a large number of minutens is to cut narrow strips of Plastazote (often the latter's thickness is conveniently 1 cm or 7 mm), to insert minutens all the way through the strips (ensuring that their tips do not extrude), and trimming off the excess length close to the strip surface with good scissors or pin cutters. To maximize efficiency later in the setting process, we prepare large quantities of trimmed minutens in advance. Minuten-loaded Plastazote strips can be packed side by side in an insect mounting tray or small shallow cardboard box. A protective layer of Plastazote is glued on the bottom of the tray or box. Strips are then laid upright, side by side, and held in place with pins inserted through the sides of the tray or box; any remaining space can be filled with Plastazote. Use a box narrow enough for the holding pins to pierce through at least half of the strips from one side.

Tools. We use curved forceps for handling minuten pins (Figure 3). The inner surface of the grasping end must be smooth (not striate). While fine straight forceps could be used, we found curved forceps to be a much more versatile tool for the task. A large standard pin (e.g. no. 4) bent at an obtuse angle (Figure 4) provides an inexpensive tool, instead of a second pair of forceps, to help in holding the specimens or the wings during pinning and setting. It is important to use a pin that is not too fine because the point may catch and rip into the wings too easily.

Setting triangles (Figure 5). Triangles are used to hold the wings in place once they are spread. They are made with a point punch (of the type commonly used for mounting small insects) from moderately thick, very smooth or glossy card and inserted no more than half way up on short pins. We use two sizes of triangles [7 mm long with pointed end (Figure 17) and 10 mm long with truncate end (Figures 10-12)] for different sizes of microlepidoptera. We mount them on pins no. 00 cut down to 1 cm in length (trimmed the same way as the minutens). Do not use minutens for mounting triangles because they are too fine to insert easily into the relatively thick card stock of the triangles. When mounting triangles, check that the side with rough edges (produced by the punch on the underside of the paper) is turned upwards (check with a magnifying lens if necessary). If this simple precaution is not taken, much damage to the wing scales will occur because of the rough edges of the triangles. As for minutens, a large supply of mounted triangles should be readied, pinned in shallow boxes. Triangles are reused indefinitely or until they become loose on the supporting pins.

Pinning pad. White cotton fabric folded several times into a pad about 1 cm thick and 10 cm X 10 cm makes an ideal surface to pin microlepidoptera. The fabric must be as soft as possible. This surface prevents specimen compression when pinning and the fabric fibres tend to hook the moth claws, thus reducing slippage. Avoid paper towels of any kind; they are usually too rough. The thickness of the pad must be greater than the length of the minutens so that the pad does not have to be lifted up when pushing the minutens through the specimens.

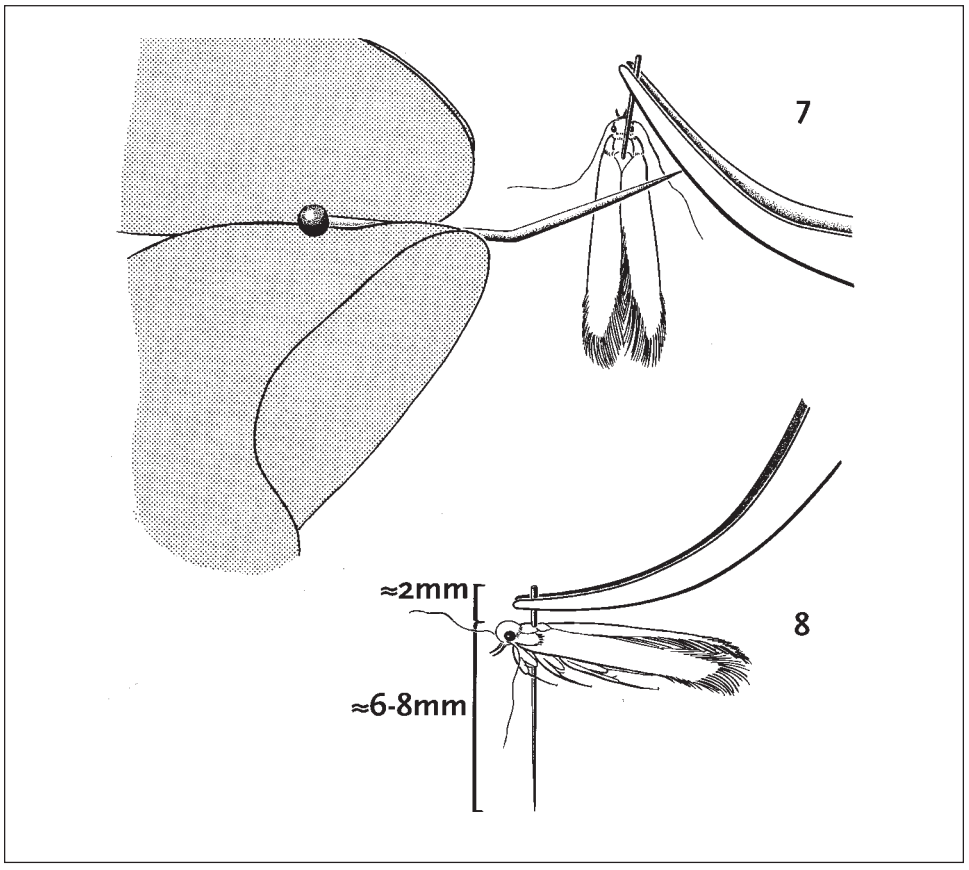
Humid container. This serves to hold pinned specimens to prevent them from drying

while they await setting. If this precaution is not taken, the smaller microlepidoptera will begin to dry in a mere few minutes in dry air and become difficult to spread by the time one gets to the last few of a batch. The container is simply made from a plastic petri dish or similar small plastic dish or box with a loosely fitting lid. The bottom of the dish is lined with wetted tissue or filter paper. A small Plastazote pad serves to hold specimens. In very dry conditions, the inside of the lid may be lightly misted to increase ambient moisture in the container (too much moisture could drip on the specimens).

Pinning and Setting

For best results (and less eye strain) the pinning and setting operations should be done with magnifying lenses or under a low-power stereoscope (up to about 5X, maximum).

Pour the freshly killed moths on the cotton pad and pin them. Insert the minuten through the center of the mesothorax (mesoscutum) or at the suture between the mesoscutum and mesoscutellum (the mesoscutellum is the roughly triangular or diamond-shaped area behind the center of the mesothorax). Try to keep the pin in line with the center of the thorax, otherwise the wing muscles may become transfixed, which renders spreading more difficult. To



Figures 7–8. 7, Inserting the minuten while holding the body with a bent standard pin; 8, Minuten-pinned specimen, showing approximate height on 1-cm-long minuten.

ensure that a specimen is squarely pinned, apply very slight pressure on its dorsum with the tip of the bent pin (or another curved forceps) to prevent the body from rolling sideways while the minuten is inserted into the mesothorax (Figure 7). The minuten must be inserted far down so as to leave no more than about 2 mm protruding above the moth, enough to manipulate it comfortably with forceps (Figure 8). Of course, the height of specimens with unusual structural modifications such as long palpi recurved over the body or thoracic crests should be adjusted appropriately in order to leave sufficient minuten length for the forceps; such specimens may require longer (untrimmed) minutens.

Place pinned specimens in the humid container. Prior to this, if one wishes, the wings may be partly opened by gently blowing on them from behind the moth with a slight puff of breath. Before proceeding with setting, another series of specimens is transferred to the killing tubes. Hence, there will be specimens ready for pinning when the first batch has been set. We usually proceed in batches of no more than 15-25 moths.

Take specimens out of the humid container singly for setting. If the wings are still closed, gently blow on them from behind, then insert the specimen into the groove (Figure 9). Lift the wings and partly push them forward with the tip of the closed curved forceps inserted beneath the wings. Tuck the legs into the groove. With a minuten position the antennae so that they form a widely obtuse V, holding them temporarily by placing minutens behind their base. If the fringes are matted, lift the wings a little and comb the fringes by brushing them with the tip of the triangle's pin in a movement going from the apex of the wings toward the body.

To fix the wings into position, we use two different procedures.

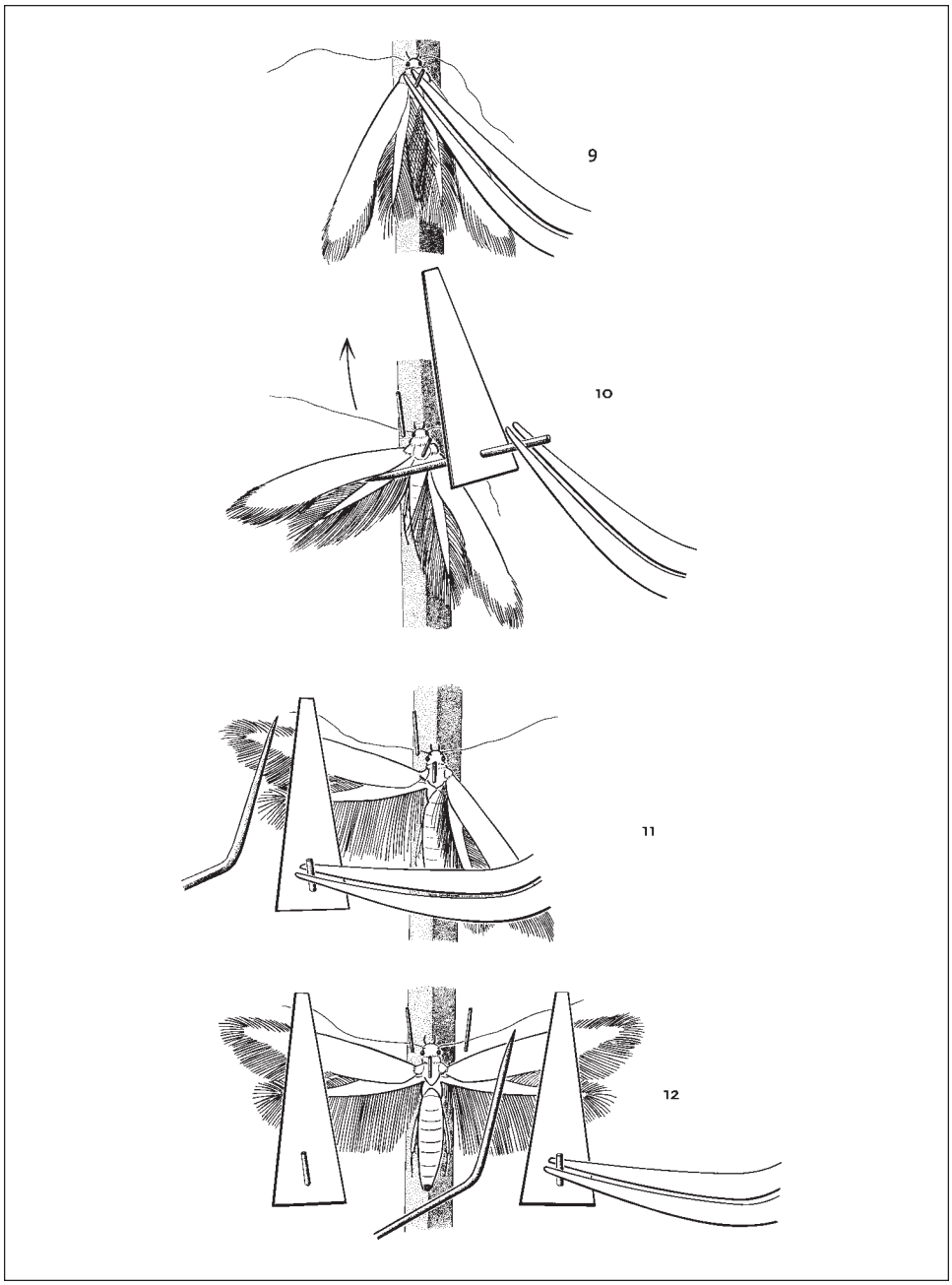
(1) Points method (Figures 10-12). This method may be a bit faster than the paper method (see below). Although excellent, it sometimes gives slightly inferior results, and makes it more difficult to set the antennae properly.

Using a mounted triangle, bring one pair of wings forward by pushing on the hind margin of the forewing with the tip of the pin. Usually, if this movement is delicately executed, both wings will move together because of the coupling. Do not pierce the wings. While holding the wings into position with a slight pressure of the bent pin held in the other hand, put the triangle on top of the wings as close to the apex of the hindwing as possible and push it down sufficiently to immobilise the wings. The triangle must lie flat against the wing surface and must not be pressed down too strongly or it will leave a mark. It may be necessary to adjust the position of the hindwing slightly, which sometimes will be a little too far back or too far forward. One or more triangles may be added to better hold the wings of larger or broad-winged microlepidoptera or to prevent them from curling up.

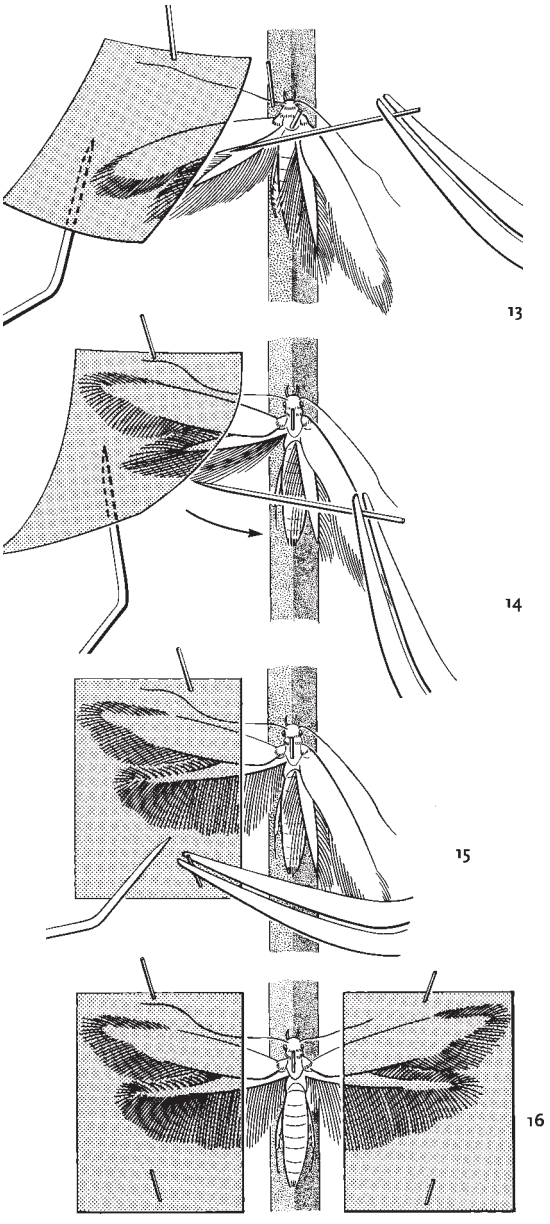
Repeat the procedure for the other side. To prevent set specimens from hindering hand work over the spreading box surface, it is best to proceed in transverse rows instead of filling one groove after another.

For someone having difficulty using both hands simultaneously, the following variation may be applied: using a mounted triangle as outlined above, move a pair of wings only halfway forward then insert the triangle over the wings just sufficiently to prevent the wings from slipping back but ensuring that they can still be moved; with a minuten move the wings into their final position (the wings should stay in place) and with the forceps maintained closed, gently push down the top of the pin holding the triangle until the wings are flat. Positioning of the wings in this way may have to be done in several stages for some specimens. The other hand may hold the spreading box. With this variation, one can proceed by filling one groove after another if desired.

(2) Paper method (Figures 13-16). The second procedure uses small strips of thin, translucent setting paper and is essentially similar to the standard technique used to spread larger Lepidoptera on a normal setting board. The paper strips are held down with 0.20 minutens.



Figures 9-12. Setting with the point method. 9, Inserting the specimen into the groove with the wings partly opened; 10, Moving one set of wings forward with the point-holding pin; note the antenna held in position with a minuten; 11, Setting the wing into position with the mounted point while holding it with the bent pin; 12, Repeating the operation with the other side.



Figures 13-16. Setting with the paper method. 13, Moving one set of wings forward with a minuten while lifting the paper strip with the bent pin; note the minuten holding the antenna; 14, Combing the fringe; arrow indicates direction of combing movement; the combing minuten touches the tip of the fringe lightly; 15, Pinning the paper strip down to secure the wing into position; the minuten holding the antenna may be removed as it is usually no longer necessary; 16, Set specimen.

This technique can yield the finest specimens because the entire surface of the wings is held flat, and the antennae can be set properly with ease. It is a little more cumbersome and may take a little more time depending on individual ability. In our own experience, however, it takes about the same amount of time as the triangle technique, if one has prepared and has ready the necessary materials, such as pre-cut pieces of setting paper and minutens.

Cut many small pieces of setting paper, just long enough to cover the antennae and one set of wings, prior to spreading. For most microlepidoptera, the strips we use are about 1–1.5 cm long and about 5 mm wide. Four minutens are usually needed to spread one moth. For increased speed, sets of four paper-holding minutens may be pinned beside each groove of an entire row before the setting begins. When one row has been filled with specimens, another series of minutens is placed along the next row, and so on.

After pinning the moth and having set the antennae as described above, pick up a paper strip by stabbing it with a minuten and pin it just ahead of the antenna to cover the half-opened wings. Check that the curvature of the paper faces upward. With the bent no. 4 pin (or another pair of curved forceps) held in one hand, slightly lift the posterior end of the paper from beneath. With another minuten held with curved forceps in the other hand and working from behind the hind margin of the forewing, push the wings into position. When both wings are positioned, drop the paper strip, hold it down with the tip of the bent no. 4 and pin it behind the hindwing with a second minuten. Repeat on the other side.

Choosing the appropriate groove width will facilitate spreading. A groove too narrow will force the legs up and put pressure on the thorax, thus hindering wing movement. A groove too wide will result in either the specimen swinging on the pin when the wings are pushed on one side, or in an insufficient portion of the wing surface resting flat.

Before placing the specimen into the groove, the Plastazote surface may be gently rubbed with the tip of the closed forceps to create a charge of static electricity which will help in spreading the wings. This is not necessary, however, if one is using the paper strip method, and it is not recommended with very small microlepidoptera such as nepticulids because the charge will be too strong and may push the wings up vertically.

With a fresh, fully relaxed moth and some practice, the whole operation of pinning and setting takes no more 30-60 seconds. With practice, specimens can be set quite closely behind one another into the grooves to conserve space (Figures 17-18).

Stunning prior to spreading is sometimes used, instead of killing, when time is short (Sokoloff 1980). If specimens are only anesthetized (stunned) prior to spreading, it is necessary to pin a small cotton swab imbibed with ammonia into the spreading box and close it for about 15-20 minutes to kill the moths. If the spreading box is made of polystyrene-base plastic, avoid ethyl acetate because it will dissolve the plastic. We do not use the stunning method because we find it inconvenient, especially in the field.

Label the specimens as usual and leave them in the spreading boxes in a dry place for at least two weeks, or preferably for as long as possible. If one does not provide enough time for the specimens to dry, the tips of some wings may curl up or droop. In humid regions, it is advisable to secure a few crystals of 4-chloro-m-cresol in the boxes to prevent molding. Once the moths are dry, full

Figures 17. Filled spreading box with point-set specimens. Scale in cm.

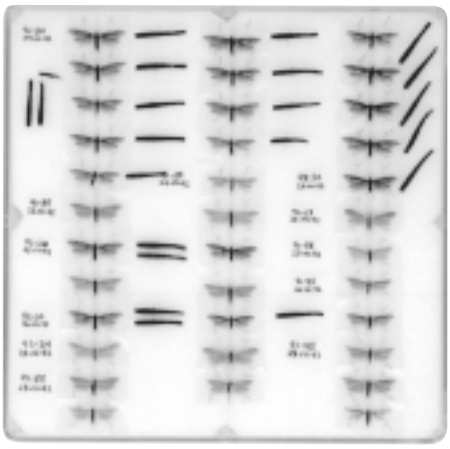


Figure 18. Filled spreading box with paper-set specimens (larval cases beside reared specimens).

boxes should be sealed tightly with tape until ready for staging.

When in the field for an extended time and spreading boxes are in short supply, or to reduce the number of boxes being transported, space can be saved by removing specimens from the grooves after drying and packing them somewhat like shingles (Figure 19). Specimens are pinned slanted in transverse rows, with the left wings of a specimen partly overlapping the right wings of the preceding one. This allows for large quantities of specimens to be stored in little space. An entire collection of several thousands of microlepidoptera can be carried in this way in a handbag on an airplane instead of being placed in regular baggage, thus maximizing the safety of specimens that may represent months of field work in a remote region.

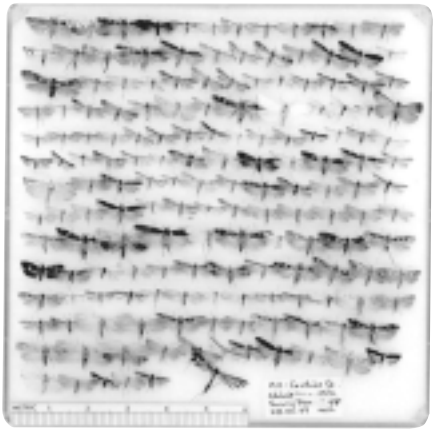


Figure 19. Example of packed spreading box in which previously set and dry specimens are overlapped like shingles to conserve space. This 11 X 11 cm box contains 166 specimens.

Some authors have recommended heat-drying because, supposedly, moths that have been heat-dried will never have drooped wings (Amsel 1935). This is, however, a delicate and risky operation that must be done very carefully with very low heat (no more than 40° C). We have tried drying on a few occasions and are rather wary of it. We have noticed that several microlepidoptera tend to become a little greasy when dried with heat (noticeable under magnification). Another problem is that the Plastazote of the spreading boxes may warp slightly from being heated. We think that it is preferable to see some wing drooping occur later in the collection than risk damaging specimens in heat-drying. Wing drooping will be minimized or virtually eliminated if specimens are allowed to remain set in the spreading boxes for an extended period.

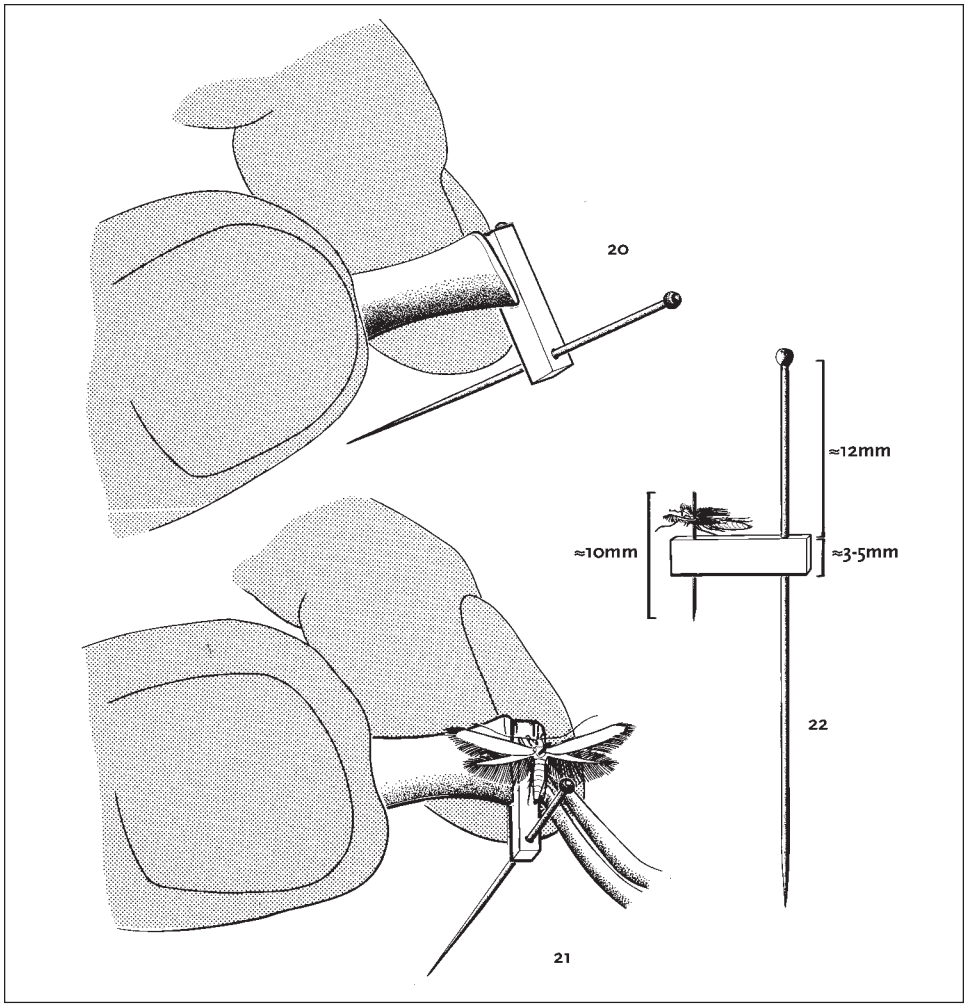
Staging

To be placed in collections, dry minuten-pinned microlepidoptera must be mounted individually on small rectangular blocks, which are inserted on standard (# 3 or 4) insect pins. This is referred to as staging or double-mounting. Specimens should always be mounted singly on a block, complete with all necessary labels on the supporting pin, except perhaps in cases of mated pairs which may be staged together. It is very annoying to find two or more microlepidoptera belonging to different but superficially similar species that have been staged together

with a single label; such specimens have to be remounted separately and new labels produced. Multiple mounts also increase the risk of misassociation of subsequently made genitalia slides.

Staging blocks. It is more efficient to prepare large quantities of blocks in advance. Traditionally, blocks have been cut from strips of polypore fungi (especially from birch bracket fungus). Normally it is easy to procure polypore strips from naturalist supply houses, but periodically they tend to become very difficult to obtain.

Plastazote provides a superior substitute. It is comparatively inexpensive, available in practically infinite supply, extremely regular in density, practically unalterable, and pest proof (we once had a supply of polypore strips heavily infested with ciid beetles). Plastazote allows the finest minutens to be inserted without effort and provides remarkable protection from shocks



Figures 20-22. Staging or double-mounting. 20, Holding with flat-tipped forceps a staging block mounted on a standard pin; 21, Inserting the specimen on the stage, clasping the minuten from below the specimen; 22, Staged specimen, showing good heights for safe subsequent handling.

and vibrations. Other materials such as balsa, cork, and polystyrene-based foam ("styrofoam") should be avoided because they are either too hard to insert the minuten without risking damage or are not rubbery enough to hold firmly the pin and the minuten (the latter is a problem of balsa and polystyrene-based foams, on which minutens frequently become loose). Blocks made of a silicon rubber compound are used by some but their durability is uncertain in insect drawers where they may be affected by fumigants; we have seen a set of such blocks that were about 15 years old and that exuded a greasy substance which seeped up the minutens and coated the specimens. It is also harder to insert a minuten into silicon rubber, which is a springy material.

The length of the blocks varies with the size of the specimens. Ideally, we think that they should be about as long as the length of the moth from its head to the tip of its abdomen plus about 3 mm to provide space for the legs and the supporting pin. The width and height vary little and are from 2-3 mm (width) and from 2-4 mm (height). We recommend the use of as few sizes of blocks as necessary to maintain some uniformity to the collection. A cutting board with preset guides and mounted razor blade can be made to speed the cutting of large numbers of uniformly sized blocks. It is essential to mount the blocks on standard pins prior to double-mounting the moths. Staging blocks must be inserted up to a height that will leave adequate clearance between the specimen and the head of the supporting pin to allow for safe handling of the whole mount (Figure 22); we recommend at least a 1-cm clearance.

Staging procedure (Figures 20-22). To facilitate staging, use one pair of forceps with curved tips and another with broad, flattened tips. With the flat-tip forceps, hold the pinned block in front of you. With the curved forceps, take the specimen by holding the minuten from beneath the specimen and insert slightly into the block. Check that the plane of the wings is perpendicular with the axis of the pin and adjust the inclination if necessary. Still grasping the minuten from beneath the specimen, pull it down into the block to the point where the venter of the moth is about 1 mm from the surface of the block.

Holding the minuten from beneath the specimen for insertion is especially critical if one is using polypore blocks. Polypore blocks vary markedly in hardness and pushing the minuten down while grasping it from above the specimen may cause the minuten to bend or spring, usually resulting in damage to the moth. Using Plastazote blocks generally obviates this danger but grasping the minuten below the moth reduces the risk of damage in case of slippage of the forceps.

It is important to insert the minuten as far down as possible, while not touching the stage, in order to secure the specimen (Figure 22). Specimens protruding high on the block risk getting damaged in subsequent handling as much as those with overly long minutens jutting high above the body.

Final Remarks

The techniques described above may seem laborious, but what takes many words to explain is actually executed in just a few seconds. With some practice, one can easily pin and set up to 30-40 microlepidoptera of fine quality per hour.

If there is no time or desire to fully spread all the moths that are collected, one may at least spread the wings partly and brush the fringes. Provisional spreading (Amsel 1935; Zimmerman 1978: pp. 48-ff; Nielsen 1980; Mikkola 1986), with subsequent relaxation and spreading if necessary or desired, is a good compromise where time is short such as during expeditions aiming at sampling as many specimens as possible. Damaged or rubbed specimens that may be worth collecting for some reason may be partially spread to save time.

Generally we do not use light traps and prefer to collect microlepidoptera at light on a sheet. Although light traps afford several advantages in sampling and are often necessary for surveys,

we find that one is easily overwhelmed by the abundance of specimens so obtained, that a significant amount of time is necessary to sort the microlepidoptera from other Lepidoptera and insects, and that most specimens sustain a certain amount of rubbing and damage. If there is no time to relax and set trap-collected specimens right away, they should be placed on slightly damp cotton in tight containers and kept in a freezer.

Methods that involve killing the specimens immediately upon capturing them (as in light traps) and storing them for an indeterminate period of time (e.g. by freezing), generally necessitate some period of relaxation in a humid chamber before proper setting can be performed. Such specimens are usually not quite as easy to spread as freshly killed specimens and are not ideal for the point-setting technique described above, although satisfactory results can be obtained with adequate relaxation and using the paper-strip technique. Specimens that have dried unspread usually cannot be subsequently relaxed and spread. Some lepidopterists who have tried our technique complained that it was not quite as easy as we told them but, when pressed for details of how they proceeded, most conceded that they had killed their specimens upon collecting and spread them a little later without relaxation. We reiterate that working from fresh, live specimens killed just before setting is central to the ease and rapidity with which microlepidoptera can be set with the technique described here, and to obtaining high-quality specimens. Of course, some experience is necessary to achieve the best results; one is unlikely to obtain perfect microlepidoptera after attempting to set only a dozen specimens.

It is a truism that fine, well-prepared specimens are easier to identify. This is particularly true for microlepidoptera, whose small size puts them at a disadvantage over the larger Lepidoptera when it comes to studying them (incidentally, lepidopterists facing space limitations to house their collection of macros should seriously consider taking up the collection of microlepidoptera!). Many well prepared microlepidoptera can be recognized at a glance. On the other hand, rubbed, damaged, or badly mounted specimens may be quite difficult to recognize, even to family, particularly if they are unspread.

Unavoidably, processing microlepidoptera soon after their collecting will take more time and seem more laborious than for larger Lepidoptera that are simply papered or pinned for subsequent setting. It can be argued, however, that the time involved strictly in spreading microlepidoptera is no more than for spreading macros; in fact spreading microlepidoptera is faster. The main difference is that one should do it right at the time of collecting for best results. The resulting quality of the specimens makes it well worth the effort.

Acknowledgments

Several lepidopterists made suggestions and comments on our technique and on spreading microlepidoptera in general, and/or have encouraged us over the years to publish our method. In particular, we are indebted to Vitor Becker, Bengt Bengtsson, Don Davis, John De Benedictis, Michael Fibiger, John Grehan, Ron Hodges, Ole Karsholt, Eric Metzler, Kauri Mikkola, John Morton, Jerry Powell, Tony Roberts, Klaus Sattler, Dave Wagner, Monty Wood, and Don Wright. We thank Eric Metzler for making us aware of ammonium carbonate, and Cees Gielis for testing it under harsh field conditions. We thank Dave Moorehouse for his assistance in preparing the figures. Ole Karsholt, Jeff Cumming, Mike Sharkey, Kevin Tuck, and an anonymous reviewer, provided many useful comments on the manuscript.

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Appendix G

Cutting Polyethylene Foam into Strips for Staging Microlepidoptera.

D. J. Wright

(Reprinted with permission from Ohio Lepid 17:3–4, 1995.)

For the last few years Ohio collectors of microlepidoptera have been using polyethylene foam as a staging medium for specimens mounted on minutens. The advantages of using this material are discussed in J-F and B Landry, J Lep Soc 48 (3) 1994, 205–277. [Reprinted here as Appendix F.] The purpose of the present note is to describe a jig for cutting 1/4" foam sheets into uniform strips of 1/8 x 1/8" square cross-section.

The cutting jig consists of a table and a cutter. The table has a platform that supports the foam, a fence which positions the foam during the cut and provides support for the cutter, and a second fence that serves as a guide for the cutter. The cutter itself is just a block of wood on which is mounted a single-edge razor blade. The block, when positioned on the first fence, holds the blade perpendicular to the platform (see Figure 1).

Two passes with the cutter are required to produce the desired strips. The first yields a strip of 1/4 x 1/8" rectangular cross-section, and the second pass divides the strip into two 1/8 x 1/8" cross-sectional strips. For the first pass a piece of 1/4" foam is placed on the platform with one edge snug against fence #1. The friction between the edge of the sheet and the first fence stabilizes the foam during the cut. The second cut is a little trickier. Place the 1/4 x 1/8" strip on the platform with the narrow edge against fence #1. To get a clean cut you need to hold the strip

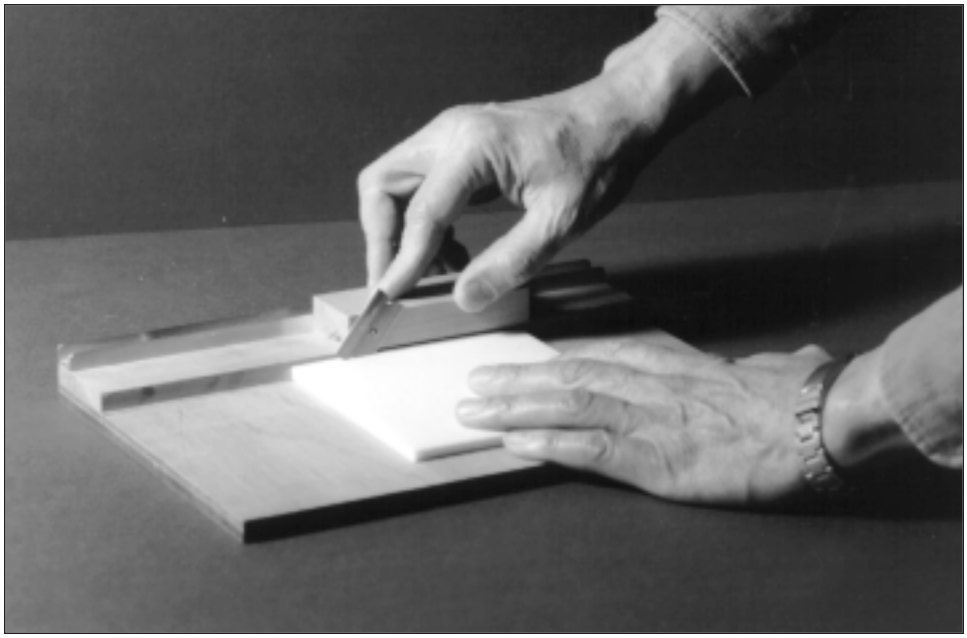

Figure 1. The cutting jig. Making first cut.

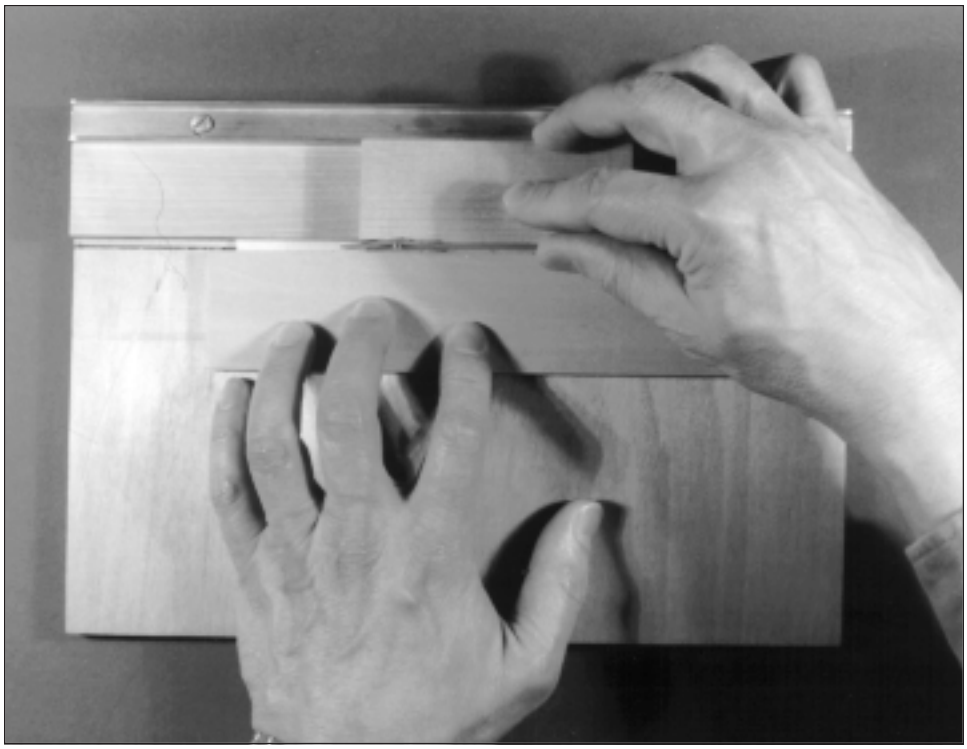

Figure 2. The cutting jig. Making second cut.

firmly against both the platform and the first fence. Given the size of the strip, this can be a little hazardous to the fingers, so I use a thin sheet of wood to hold the strip in place (see Figure 2).

Figure 3 shows a cross-section of the table perpendicular to the fences. The platform is an 8 x 12" piece of 1/2" plywood. Note that it has a 1/16 x 1/8" slot running parallel to the fence. The center of the slot should be located 1/8" from fence #1. When making a pass with the cutter, the tip of the razor blade fits in the slot. Affixed to the platform is a 3/8 x 2 x 12" piece of wood that serves as the first fence. It is crucial that the long edge of this fence be parallel to the slot in the platform. The second fence is mounted on the first. In the prototype, illustrated in Figure 1, it was made from a scrap piece of aluminum channel. The 1/2 x 1/2" aluminum L angle depicted in Figure 3 should also work nicely. Fence #2 is attached with two round-head screws, located 2" from each end. Drill elongated slots in the fence to accommodate the screws and assemble with a flat washer between the screw head and the fence. By loosening the screws a little you can fine tune the position of the fence so as to get it perfectly parallel to the slot in the platform. Note that the second fence is mounted so that the guide edge is 11/2" from the edge of fence #1.

Finally, the cutter is a 3/4 x 1 5/8 x 4" block of wood (Figure 4). The end of one of the long edges is relieved a little to accept the thick edge of the razor blade. The relief allows the blade to lie flat against the edge of the block. The blade is attached to the block with a small round head screw. Position the blade on the block so that its thickened edge is snug against the relieved angled edge of the block and so that the cutting corner extends 7/16" below the bottom surface

of the block. Then trace on the block the central slot in the razor blade. Remove the blade and drill a pilot hole for the mounting screw. If the screw is placed at the bottom of the blade slot, it will hold the thickened edge of the blade against the angled edge of the relief and thereby prevent the blade from turning during the cut. Tighten the mounting screw just enough to hold the blade firmly. Overtightening can cause the blade to warp. Change blades frequently. You will need a sharp blade to get a clean cut on the second pass.

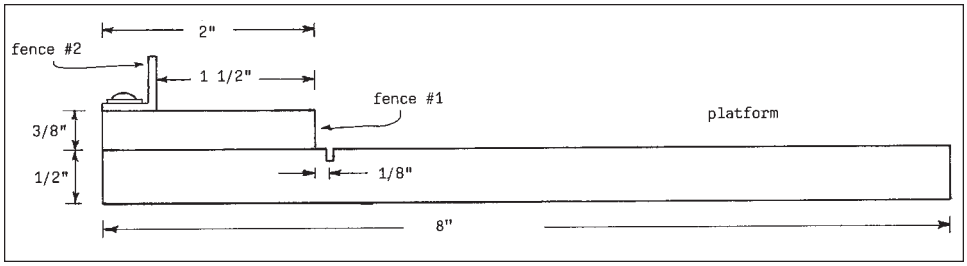

Figure 3. Cross-section view of the cutting jig.

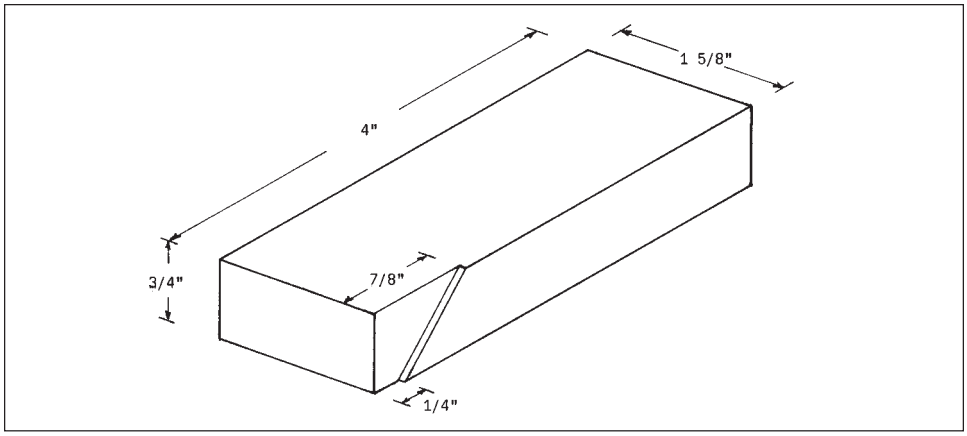

Figure 4. Detail of the cutter block of wood.

Appendix H

Sesiid Pheromones

Chemically speaking, what are the sesiid pheromones, and how do you digest the alphabet soup used to name them? They are variations on an 18-carbon, straight chain compound, "octadecadien", that has one "double bond" after carbon #2 or #3, and a second one after carbon #13. So "2" vs. "3" is one variable (13 does not vary). Next, the hydrogen atoms attached to the carbons on each side of the double bond may be side-by-side (German *zusammen* or together) or rotated away from one another (*entgegen* or opposite). Another variable is therefore "Z" vs. "E." Finally, an appendage at the #1 end of the octadecadien chain can be either an acetate or an alcohol group, shorthanded as "A" and "OH," respectively. When the characteristics of the commonest pheromone are strung together it is named "Z,Z 3,13-octadecadien-1-ol acetate" (Fig. 1). For convenience, the contraction Z,Z 3,13-ODDA is commonly used. Variation at position 3 gives E,Z 3,13-ODDA, and substitution of the alcohol for the acetate radical gives E,Z 3,13-ODDOH. If the double bond is at position 2 instead of 3, the shorthand is E,Z 2,13-ODDOH (Figure 2). Because the configuration at position 13 is always "Z," suppliers often employ even shorter acronyms, such as "ZZA," "EZA," "EZOH," and "EZOH 2,13," respectively. Flaschka's paper (1992), from which this summary was derived, provides a more detailed exposition of the chemical configurations.

The primary references for the Sesiidae (clearwing borers) are Engelhardt (1946); Eichlin and Duckworth (1988); and Taft et al. (1991).

Table 1, below, lists lures retailed by two manufacturers. The lure acronym is follwed by the manufacturer's statement as to target species, retailer, apparent pheromone composition, and the common name of each species.

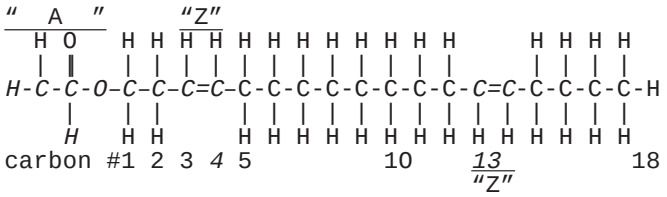

Figure 1. "Z,Z 3,13-octadecadien-1-ol acetate," "Z,Z 3,13 ODDA," or "ZZA." Variable areas: italics.

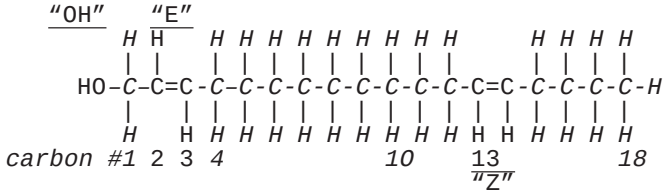

Figure 2. "E,Z 2,13-ODDOH," or "EZOH 2,13." Variable areas: italics.

Table 1. Commercially Available Pheromones

Lure name	*Target species*	*Available from*	*Compound(s), inferred from Eichlin & Duckworth (1988) unless qualified*	*Common name*
(?)[1]	Melittia cucurbitae	*IPM*	*EZA 2,13/ZZA 99:1*[2]	*squash vine borer*
CWB	Paranthrene simulans	*IPM*	*ZZA/EZA 97:3*[2]	*oak borer*
CWB	Podosesia aureocincta	*IPM*	*ZZA/EZA/ZZOH*	*banded ash borer*
CWB		*IPM*	*ZZA*	
LILA	P. syringae	*Trécé*	*ZZA*	*lilac/ash borer*
SCM	Synanthedon bibionipennis	*Trécé*	*EZA/EZOH 2:1*	*strawberry crown moth*
CWB		*IPM*	*ZZA*	
GTPB	S. exitiosa	*Trécé*	*ZZA*	*greater peachtree borer*
LPTB	S. pictipes	*Trécé*	*EZA*[2]	*lesser peachtree borer*
CWB	S. rhododendri	*IPM*	*ZZA*	*rhododendron borer*
DWB	S. scitula	*IPM, Trécé*	*ZZA, 100*[2]	*dogwood borer*
SPM	S. sequoiae	*Trécé*	*ZZOH*	*sequoia pitch moth*
(?)[1]	Vitacea polistiformis	*IPM*	*EZA 2,13/ZZA 99(or less):1(or more)*[2]	*grape root borer*

[1] available catalog showed no acronym.

[2] information from manufacturer, as obtained by H. Flaschka, pers. comm.

Scentry *and* ***Trécé*** *lures are obtainable from Great Lakes IPM, 10220 Church Road, NE, Vestaburg, MI 48891; phone 517-268-5693 or -5911. They also carry the durable and effective Universal Moth Trap (Unitrap) and DDVP toxicant strips (Hercon Vaportape).*

Trécé *lures are obtainable from Trécé Inc., 1143 Madison Lane, Salinas, CA 93912; phone 408-758-0205.*

Table 2. Sesiid pheromones, listed by species

Table 2 should simplify finding the attractant pheromone for a given species of clear-wing borer. In this list "–,Z -ODDA" or "–,Z -ODDOH" implies the "3, 13" form unless otherwise clarified.

Species	*Pheromone*
Penisetia marginata	*E,Z-ODDOH*
Sophona greenfieldi	*Z,Z-ODDOH ± E,Z-ODDA*
S. snellingi	*Z,Z-ODDA*
Zenodoxus canescens	
Z. heucherae	
Z. maculipes	
Z. mexicanus	
Z. palmii	
Z. rubens	*Z,Z-ODDA*
Z. sidalceae	
Cissuvora ampelopsis	
Paranthrene asilipennis	*Z,Z-ODDA*
P. dollii	*Z,Z-ODDOH/E,Z-ODDOH*
P. tabaniformis	*E,Z-ODDOH*
P. fenestrata	*Z,Z-ODDA*
P. robiniae	
P. simulans[1]	*Z,Z-ODDA*

Table 2, continued.

Species	*Pheromone*
Paranthrene pellucida	Vitacea admiranda
Vitacea cupressi	
V. polistiformis	
V. scepsiformis	*Z,Z-ODDA; /E,Z-ODDA, or E,Z-ODDOH*
Albuna fraxini	
A. pyramidalis	
Euhagena emphytiformis	
E. nebraskae	
Melittia cucurbitae	*E,Z-2,13 ODDA/Z,Z-3,13 ODDA 99:1*
M. calabaza	
M. snowii	
M. grandis	*E,Z-2,13 ODDA/Z,Z-3,13 ODDA 99:1*
M. gloriosa	
M. magnifica	
Sesia apiformis	
S. tibialis	*Z,Z-ODDA*
Calasesia coccinea	
Osminia ruficornis	*E,Z-2,13 ODDOH*
O. donahueorum	
Synanthedon acerrubri	*E,Z 2,13-ODDA*
S. geliformis	
S. richardsi	
S. scitula	*Z,Z-ODDA*
S. pictipes	*E,Z 3,13-ODDA*
S. rhododendri	*Z,Z-ODDA*
S. rileyana	*E,Z-ODDOH, ± E,Z-ODDA*
S. tipuliformis	*E,Z-2,13 ODDA/Z,Z-3,13 ODDA*
S. acerni	*Z,Z-ODDA*
S. fatifera	*Z,Z-ODDA*
S. viburni	*E,Z-ODDA*
S. alleri	*E,Z-ODDA*
S. arctica	
S. bolteri	
S. canadensis	
S. culiciformis	*Z,Z-ODDA*
S. dominicki	*E,Z 2,13-ODDA*
S. fulvipes	*Z,Z-ODDA*
S. helenis	
S. pyri	*E,Z 2,13-ODDA/Z,Z 3,13-ODDA 99:1*
S. refulgens	*E,Z 2,13-ODDA/Z,Z 3,13-ODDA 99:1*
S. rubrofascia	*Z,Z-ODDA, ± E,Z-ODDA 50:50*
S. saxifragae	*Z,Z-ODDA*
S. sigmoidea	
S. albicornis	*Z,Z-ODDA*
S. decipiens	*Z,Z-ODDA, ± E,Z-ODDA or E,Z-ODDOH*
S. proxima	
S. sapygaeformis	*Z,Z-ODDA*
S. arizonensis	

Table 2, continued.

Species	*Pheromone*
Synanthedon arkansasensis	*E,Z-ODDA or E,Z-ODDOH*
S. bibionipennis	*E,Z-ODDA/E,Z-ODDOH 2:1*
S. castaneae	*E,Z-ODDA*
S. chrysidipennis	
S. kathyae	*Z,Z-ODDA*
S. mellinipennis	
S. polygoni	
S. resplendens	
S. exitiosa	*Z,Z 3,13-ODDA*
S. novaroensis	*Z,Z-ODDA*
S. pini	
S. sequoiae	*Z,Z-ODDOH*
Palmia praecedens	
Podosesia aureocincta	*Z,Z:E,Z:Z,Z-ODDOH*
P. syringae	*Z,Z-ODDA*
Sannina uroceriformis	*E,Z-ODDOH/Z,Z-ODDA 90:10 or 3,13 E,Z-ODDOH ± 3,13 Z,Z-ODDOH*
Carmenta albociliata	*Z,Z-ODDA*
C. anthracipennis	*Z,Z-ODDA ± E,Z-ODDA 1:1*
C. apache	
C. arizonae	
C. armasata	
C. auritincta	
C. bassiformis	*Z,Z-ODDA/Z,Z-ODDOH*
C. corni	*Z,Z-ODDA*
C. engelhardti	*3,13 Z,Z-ODDA/3,13 E,Z-ODDA/3,13 Z,Z -ODDOH 20:1:3*
C. giliae	
C. ithacae	*Z,Z-ODDA/E,Z-ODDA*
C. laurelae	*Z,Z-ODDA*
C. mariona	
C. mimuli	*Z,Z-ODDA*
C. odda	*Z,Z-ODDA*
C. ogalala	
C. pallene	*Z,Z-ODDA*
C. phoradendri	
C. prosopis	
C. pyralidiformis	*E,Z-ODDOH or ZZ-ODDA*
C. querci	
C. rubricincta	
C. subaerea	
C. suffusata	*E,Z-ODDOH ± Z,Z-ODDA 1:1*
C. tecta	
C. texana	*Z,Z-ODDA or E,Z-ODDOH*
C. verecunda	
C. welchelorum	*Z,Z-ODDA*
C. welleri	
C. wielgusi	*E,Z-ODDA ± Z,Z-ODDA*
Penstemonia clarkei	

Table 2, continued.

Species	*Pheromone*
Penstemonia dammersi	
P. edwardsii	
P. hennei	
P. pappi	
Alcathoe carolinensis	*E,Z-ODDA or E,Z-ODDA/Z,Z-ODDA 3:1*
A. caudata	
A. autumnalis	
A. pepsioides	
A. verrugo	
Hymenoclea palmii	

1, Sibling species may be involved (Eichiln & Duckworth 1988).

Table 3. Pheromones and the sesiid species they attract

The primary data were taken from Eichlin and Duckworth (1988). Additional information, [in brackets], using the shorter acronyms, is from Taft et al. (1991). Species are grouped by the pheromone or combinations to which they have been observed to respond. Using this table should make it easy to define whether an attraction you have effected is already known or might be new.

Pheromone	*Species*
E,Z 2,13-ODDA;[EZA2,13/ZZA 99:1 or blend]	Synanthedon acerrubri
E,Z 2,13-ODDA	S. dominicki
E,Z 2,13-ODDA/Z,Z-ODDA 99:1; [EZA2,13/ZZA 99:1 or blend]	S. pyri
E,Z 2,13-ODDA/Z,Z-ODDA 99:1 [EZA2,13/ZZA 99:1 or blend]	S. refulgens
E,Z-2,13 ODDA/Z,Z ODDA 99:1; [EZA2,13/ZZA 99:1 or blend]	Melittia cucurbitae
E,Z-2,13 ODDA/Z,Z-ODDA	Synanthedon tipuliformis
E,Z-2,13 ODDA/Z,Z-ODDA 99:1	Melittia grandis
E,Z-2,13 ODDOH	Osminia ruficornis
[EZA or blend]	Albuna pyramidalis
[EZA/2,13/ZZA 99:1 or blend]	Vitacea polistiformis
[EZA/2,13/ZZA 99:1 or blend]	Synanthedon sigmoidea
E,Z-ODDA	S. alleri
E,Z-ODDA	S. castaneae
E,Z-ODDA; [EZA or blend]	S. viburni
E,Z-ODDA; [EZA or blend]	S. pictipes
E,Z-ODDA or E,Z-ODDA/Z,Z-ODDA 3:1	Alcathoe carolinensis
E,Z-ODDA or E,Z-ODDOH	Synanthedon arkansasensis
E,Z-ODDA ± Z,Z-ODDA	Carmenta wielgusi
E,Z-ODDA/E,Z-ODDOH 2:1	Synanthedon bibionipennis
E,Z-ODDOH	Penisetia marginata
E,Z-ODDOH	Paranthrene tabaniformis
E,Z-ODDOH or ZZ-ODDA; [ZZOH or blend; EZOH or blend]	Carmenta pyralidiformis

Table 3, continued.

Pheromone	*Species*
E,Z-ODDOH ± Z,Z-ODDA 1:1	Carmenta suffusata
E,Z-ODDOH, ± E,Z-ODDA; [EZOH or blend; ZZA/EZOH 50:50]	Synanthedon rileyana
E,Z-ODDOH/Z,Z-ODDA 90:10 or E,Z-ODDOH ± Z,Z-ODDOH	Sannina uroceriformis
[ZZA or blend]	Paranthrene pellucida[1]
[ZZA or blend]	Synanthedon proxima
Z,Z-ODDA	Sophona snellingi
Z,Z-ODDA	Zenodoxus rubens
Z,Z-ODDA; [EZA2,13/Z13-ODA 96:4]	Paranthrene asilipennis
Z,Z-ODDA	P. fenestrata
Z,Z-ODDA; [EZA2,13/ZZA 99:1 or blend]	P. simulans[1]
Z,Z-ODDA	Sesia tibialis
Z,Z-ODDA; [ZZA or blend]	Synanthedon scitula
Z,Z-ODDA	S. rhododendri
Z,Z-ODDA;[ZZA/ZZOH 50:50]	S. acerni
Z,Z-ODDA; [ZZA or blend]	S. fatifera
Z,Z-ODDA	S. culiciformis
Z,Z-ODDA [ZZA or blend]	S. fulvipes
Z,Z-ODDA	S. saxifragae
Z,Z-ODDA	S. albicornis
Z,Z-ODDA	S. sapygaeformis
Z,Z-ODDA	S. kathyae
Z,Z-ODDA; [ZZA or blend]	S. exitiosa
Z,Z-ODDA	S. novaroensis
Z,Z-ODDA; [ZZA or blend]	Podosesia syringae
Z,Z-ODDA	Carmenta albociliata
Z,Z-ODDA; [ZZA or blend]	C. corni
Z,Z-ODDA	C. laurelae
Z,Z-ODDA	C. mimuli
Z,Z-ODDA	C. odda
Z,Z-ODDA	C. pallene
Z,Z-ODDA	C. welchelorum
Z,Z-ODDA or E,Z-ODDOH	C. texana
Z,Z-ODDA, ± E,Z-ODDA 1:1; [ZZA or blend; ZZA/EZA 50:50]	Synanthedon rubrofascia
Z,Z-ODDA, ± E,Z-ODDA 1:1; [ZZA/EZA 50:50]	Carmenta anthracipennis
Z,Z-ODDA, ± E,Z-ODDA or E,Z-ODDOH; [ZZA or blend; ZZA/EZA 50:50]	Synanthedon decipiens
Z,Z-ODDA/E,Z-ODDA; [EZA or blend]	Carmenta ithacae
Z,Z-ODDA/E,Z-ODDA/Z,Z-ODDOH; [ZZA or blend; ZZA/ZZOH 50:50; ZZA/EZOH 50:50]	Podosesia aureocincta
Z,Z-ODDA/E,Z-ODDA/Z,Z-ODDOH 20:1:3	Carmenta engelhardti
Z,Z-ODDA/Z,Z-ODDOH [ZZA or blend; ZZA/ZZOH 50:50]	C. bassiformis
Z,Z-ODDA; /E,Z-ODDA or E,Z-ODDOH	Vitacea scepsiformis
Z,Z-ODDOH	Synanthedon sequoiae
Z,Z-ODDOH ± E,Z-ODDA	Sophona greenfieldi

Table 3, continued.

Pheromone	*Species*
Z,Z-ODDOH/E,Z-ODDOH	
[EZA or blend; ZZOH/EZA 50:50]	Paranthrene dollii
[Z,Z-ODDOH/E,Z-ODDOH]	*[*Sesia spartani *Eichlin & Taft]*
[ZZOH or blend; ZZA/EZA 50:50]	Alcathoe caudata
[ZZOH or blend]	Albuna fraxini
[ZZOH/EZOH 50:50]	Synanthedon pini
Species with no pheromone recorded	
	Zenodoxus heucherae
	Z. maculipes
	Z. mexicanus
	Z. palmii
	Z. sidalceae
	Cissuvora ampelopsis
	Paranthrene robiniae
	Vitacea admiranda
	V. cupressi
	Euhagena emphytiformis
	E. nebraskae
	Melittia calabaza
	M. snowii
	M. gloriosa
	M. magnifica
	Sesia apiformis
	Calasesia coccinea
	Osminia donahueorum
	Synanthedon geliformis
	S. richardsi
	S. arctica
	S. bolteri
	S. canadensis
	S. helenis
	S. arizonensis
	S. chrysidipennis
	S. mellinipennis
	S. polygoni
	S. resplendens
	Palmia praecedens
	Carmenta apache
	C. arizonae
	C. auritincta
	C. giliae
	C. mariona
	C. ogalala
	C. phoradendri
	C. prosopis
	C. querci
	C. rubricincta

Table 3, continued.

Pheromone	*Species*
	Carmenta subaerea
	C. tecta
	C. verecunda
	C. welleri
	C. armasata
	Penstemonia clarkei
	P. dammersi
	P. edwardsii
	P. hennei
	P. pappi
	Alcathoe autumnalis
	A. pepsioides
	A. verrugo
	Hymenoclea palmii

REFERENCES

Eichlin TD & WD Duckworth 1988.
Sesiodea, Sesiidae. Moths of America North of Mexico Fasc. 5.1. The Wedge Entomological Research Foundation, Washington, DC.

Engelhardt GP 1946.
The North American clear-wing moths of the family Aegeriidae. US Natl. Mus. Bull. 190, Washington, DC.

Flaschka HA 1992. A chat about sesiid pheromones. Idalia 3, No. 3, p. 3–6.

Taft WH, D Smitley, & JW Snow 1991.
A guide to the clearwing borers (Sesiidae) of the north central United States. North Central Regional Publ. No. 394, Michigan State University, East Lansing, Michigan.

Appendix I

Book Condition Standards

(Quoted verbatim from Antiquarian Bookman's Weekly*, 1 May 1995, p. 1947, provided by K. and J. Donahue.)*

Terms used to describe condition of books are as varied and numerous as the creativity and imagination of bookmen can produce. When confusion reigns over descriptions by advertisers or quoters, dissatisfaction is the inevitable result.

In an effort to promote agreement between buyer and seller in the descriptions used for the condition of books, *AB* first proposed in 1949 a set of terms that could serve as a standard in catalogue and mail-order transactions. The list was published again in the 1975 edition of *ABC of the Book Trade* by Sol. M. Malkin (the 1975 *AB Yearbook,* Part Two, is now out of print, but will be revised at a later date). A revised list of terms used in describing books is now published here in each weekly issue of *AB* to serve as a suggested guide and reference for bookmen:

1. **As new** is to be used only when the book is in the same immaculate condition in which it was published. There can be no defects, no missing pages, no library stamps, etc., and the dustjacket (if it was issued with one) must be perfect, without any tears. (The term **as new** is preferred over the alternative term **mint** to describe a copy that is perfect in every respect, including jacket.)
2. **Fine** approaches the condition of **as new**, but without being crisp. For the use of the term **fine** there must also be no defects, etc., and if the jacket has a small tear, or other defect, or looks worn, this should be noted.
3. **Very good** can describe a used book that does show some small signs of wear—*but no tears*—on either binding or paper. Any defects must be noted.
4. **Good** describes the average used and worn book that has all pages or leaves present. Any defects must be noted.
5. **Fair** is a worn book that has complete text pages (including those with maps or plates) but may lack endpapers, half-title, etc. (which must be noted). Binding, jacket (if any), etc., may also be worn. All defects must be noted.
6. **Poor** describes a book that is sufficiently worn that its only merit is as a **reading copy** because it does have the complete text, which must be legible. Any missing maps or plates should still be noted. This copy may be soiled, scuffed, stained or spotted and may have loose joints, hinges, pages, etc.
7. **Ex library** copies must always be designated as such *no matter what the condition of the book.*
8. **Book club** editions must always be noted as such *no matter what the condition of the book.*
9. **Binding copy** describes a book in which the pages or leaves are perfect but the binding is very bad, loose, off, or nonexistent.

In all cases, the lack of a dustjacket should be noted if the book was issued with one.

These terms may be arbitrary, but whatever terms are employed, they may be useless or misleading *unless both buyer and seller agree on what they mean in actually describing the book.*

When in doubt, describe the book exactly as it is, as to physical condition, textual reading, and edition.

Always bear in mind that a bookseller's reputation and credibility are his most valuable assets, and accurate description preserves that credibility.

Appendix J

Book Lists

The following is a list of many of the books of interest to lepidopterists, arranged alphabetically by senior author under various arbitrary subheadings. Included with descriptive annotation are some books that are listed as references within particular chapters. Bear in mind that a title listed under "Gardening" may also be an outstanding source of information on biology and behavior, or on identification of many commoner species. Perusing all the subgroups in the book lists may lead you to some unexpected pearls.

Also included are a number of valuable but hard-to-find works from the 19th or early 20th centuries, most noteworthy for their larval drawings.

The entries are listed under the following subdivisions:

1. Identification, Broad Area
   (= half the continent or more for North America)
2. Identification, Regional
3. Atlases, Checklists and Lists
4. Butterfly Watching
5. Gardening
6. Rearing & Immatures
7. Not Classified
8. Special Subjects
9. Videos

Some of these books can be found in school and local public libraries, or through the library exchange system. Many of the more recent books are available through bookstores, but check prices before you order—a few of them are rather costly because of extensive color illustration. Some catalogs and book stores offer substantial discounts. These are not necessarily the only sources. For titles not from conventional publishers, an effort has been made to include an ordering source whenever possible. Some books may have gone out of print since this list was compiled.

Prices are noted only to indicate the general range in which a publication falls. They are subject to change over time and do not include shipping charges or taxes unless so stated. An advance inquiry about these additional costs can streamline the ordering process. When a book is available in both hard and soft cover, only the soft cover (lower) price is listed. Prices for Lepidopterists' Society publications are the member-discount prices.

Source abbreviations

BQ = BioQuip
ERS = Entomological Reprint Specialists
LS = The Lepidopterists' Society
XS = Xerces Society
YES = Young Entomologists' Society (addresses in Appendices K or L)
A few specialty book dealers are listed in Appendix L.

1. Identification, Broad Area

Covell CV 1984. A field guide to the moths of eastern North America. Boston: Houghton Mifflin. *Species accounts and color or black & white illus. of most of the commoner and widely distributed eastern moths (Peterson Field Guides). ($16, BQ).*

D'Abrera B 1971–1995.
Butterflies of the world. Victoria, Australia: Hill House. *Color photographs of spread specimens of all species, worldwide, divided by broad regions, some subdivided, to a total of 15 volumes. ($220–$270 per vol., BQ).*

— 1986. Sphingidae mundi. Faringdon, UK: E. W. Classey. *All the sphingids of the world, color illustrations of spread specimens, brief descriptions. ($185, BQ).*

— 1984. Butterflies of South America. Victoria, Australia: Hill House. *Color illustrations of many South American species, with minimal distributional notes. Omitted are species or groups well illustrated in other works. ($19.50, BQ).*

Ehrlich PA & AH Ehrlich 1961.
How to know the butterflies. Dubuque, Iowa: Brown Co. Publishers. *Diagnostic keys, species accounts, detailed line drawings of the butterflies (except skippers).*

Feltwell J & B Hargreaves 1992.
American nature guides: Butterflies of North America. New York: Smithmark. *British treatment of over 350 species of N. A. butterflies. Good illustrations; ranges and flight periods; lacks foodplant information.*

Higgins LG & ND Riley 1970.
Butterflies of Britain and Europe. Boston: Houghton Mifflin. *Color illustrations, descriptions, distribution maps.*

Hodges RW (ed.) 1971 and continuing.
Moths of America north of Mexico. Primarily Wedge Entomological Research Foundation. *Multiple fascicles, with more coming, with extensive and definitive species accounts, unsurpassed color photographs. Groups covered completely or in part to date: Sesiidae, Cosmopterigidae, Oecophoridae, Gelechiidae, Pyralidae, Geometrinae, Mimallonidae, Apatelodidae, Bombycidae, Lasiocampidae; Saturniidae, Sphingidae, Lymantriidae,* Euxoa, *Plusiinae, Cuculliinae, Stiriinae, and Psaphidinae. ($22–$75: Wedge Entomological Research Foundation, 85253 Ridgetop Drive, Eugene, OR 97405-9535 USA).*

Holland WJ 1898; revised 1931.
The butterfly book. New York: Doubleday Doran. *Broad coverage of U.S. species; text and some names out of date, but still very useful. In many public libraries.*

— 1903. The moth book. New York: Doubleday Doran *(also Dover reprint, emended by AE Brower. New York 1958). Available in many public libraries, it is still the only illustrated treatment of many of the North American moths. See Johnson JM, below.*

Howe WH 1975. Butterflies of North America. New York: Doubleday. *Detailed accounts of the species and subspecies of butterflies of the U.S. & Canada, with complete color illustration. ($55, BQ).*

Johnson JM 1994. An update to the nomenclature of Holland's moth book. *Source: J. M. Johnson, 59 East 400 North, Payson, UT 84651. ($15).*

Klots AB 1951. A field guide to the butterflies of North America, east of the great plains. Boston: Houghton Mifflin. *Detailed species accounts, color and black & white illustrations. (Peterson Field Guides, $13, BQ). See revision by Opler & Malikul, below.*

Lewis HL 1973, 1987.
Butterflies of the world. Chicago: Follett Publishing. *Over 5,000 color illustrations, grouped geographically and by families, worldwide, very brief distributional notes. ($30, BQ).*

Mitchell RT & HS Zim 1987.
Butterflies and moths. New York: Golden Press. *Introductory handbook to the commoner species. ($5, BQ).*

Opler PA 1994. Peterson first guides: Butterflies and moths: A simplified field guide to the common butterflies and moths of North America. New York: Houghton Mifflin Co. *($4.95).*

— & GO Krizek 1984.
Butterflies east of the Great Plains. Baltimore: Johns Hopkins Univ. Press. *Detailed accounts of the biology of each species, distribution maps, striking color photographs of living butterflies in the wild. ($49.50, BQ).*

— & V Malikul 1992.
A field guide to eastern butterflies. Boston: Houghton Mifflin. *Complete revision of Klots 1951; more species, all illustrations in color, distribution maps. Second printing, with errors corrected, 1995. ($17, Peterson Field Guides).*

Pyle RM 1981. The Audubon Society field guide to North American butterflies. New York: Alfred A. Knopf. *Keyed by color and silhouette, illustrated by color photos taken in the wild, detailed behavioral and biological treatment for all U.S. and Canadian butterflies and skippers. ($18, BQ).*

Sargent TD 1976. Legion of night: The underwing moths. Amherst, MA: Univ. of Massachusetts Press. *Detailed accounts of the underwing moths (Catocala) of eastern North America, color illustrations, and general discussions of this striking group of moths. ($22.50, BQ).*

Scott JA 1986. The butterflies of North America. Stanford, CA: Stanford University Press. *Species accounts, distribution maps, extensive specific and general biological treatment, color illustrated; includes Bermuda and Hawaii. ($49.50, BQ).*

Smart P 1975, 1989.
The international butterfly book. New York: T. Y. Crowell Co. *Good general information, color photos of representatives of all families, worldwide, over 2,000 illustrated. ($25, BQ).*

Smith DS, LD Miller & JY Miller 1994.
The butterflies of the West Indies and south Florida. New York: Oxford

University Press. *Covers all 350 species of butterflies and skippers known in the region, biology, distribution, influence of islands on biogeography and conservation. ($125, hardcover).*

Tilden JW & AC Smith 1986.
A field guide to western butterflies. Boston: Houghton Mifflin. *A field guide for the West complementing that of Opler for the East. ($15, Peterson Field Guides).*

Tuskes PM, JP Tuttle & MM Collins 1996.
The wild silk moths of North America: A natural history of the Saturniidae of the United States and Canada. Ithaca, NY: Cornell University Press. *Excellent color plates. ($75).*

Tyler HA 1975. The swallowtail butterflies of North America. Healdsburg, CA: Naturegraph. *Species accounts, biology, and color illustrations of swallowtails of North America, including Mexico.*

—, KS Brown & K Wilson 1994.
Swallowtail butterflies of the Americas: A study in biological dynamics, ecological diversity, biosystematics, and conservation. Scientific Publishers, P.O. Box 15718, Gainesville, FL 32604. *($50, hardcover; softcover edition available to students and third-world residents only).*

Williams JG 1971. Butterflies of Africa. Boston: Houghton Mifflin. *Descriptions of 436 species, 283 illustrated in color.*

2. Identification, Regional

Acorn J 1993. Butterflies of Alberta. Lone Pine Publishing, 206, 10426- 81 Avenue, Edmonton, Alberta, Canada T6E 1X5. *Guide to the 156 species known for Alberta, with ranges, behavior, and larval foodplants, emphasis on watching and photography. (≈$13 US).*

Allen TJ 1997. The butterflies of West Virginia and their caterpillars. Pittsburgh, PA: Univ. of Pittsburgh Press. *All the State's species (useful for all middle-latitude U.S, Atlantic to Midwest), larvae, ranges, life histories. ($23).*

Bailowittz RA & JP Brock 1991.
Butterflies of southeastern Arizona. Tucson, AZ: Sonoran Arthropod Studies. *A detailed and well illustrated account of the seasonal occurrence, distribution, and habits of the local butterfly fauna. ($30, BQ).*

Bird CC, GJ Hilchie, N Kondla, EM Pike & FAH Sperling 1995.
Alberta butterflies. *All the province's species, distribution, all color illustration; history of its lepidopterists; hard cover. From Federation of Alberta Naturalists, Box 1472, Edmonton, Alberta, CANADA T5J 2N5.*

Christensen JR 1981.
A field guide to butterflies of the Pacific Northwest. Moscow, ID: Univ. of Idaho Press.

DeVries PJ 1987. The butterflies of Costa Rica. Princeton, NJ: Princeton Univ. Press. *Color plates and biology of the Papilionidae, Pieridae, and Nymphalidae of the region. Useful over a much broader area. ($27).*

Dornfield EJ 1980.
The butterflies of Oregon. Portland, OR: Timber Press.

Ebner J 1970. Butterflies of Wisconsin. Milwaukee, WI: Milwaukee Public Museum. *Species descriptions and black & white illustrations of butterflies and skippers, statewide.*

Emmel TC & JF Emmel 1973.
The Butterflies of southern California. Los Angeles: Natural History Museum of Los Angeles County. *Species accounts, with life histories in detail, covering San Luis Opispo, Kern, and San Bernardino Counties, and southward; color illustrations. ($7.75 BQ).*

—, M Minno & B Drummond 1992.
Florissant butterflies: A guide to the fossil and present-day species of central Colorado. Stanford, CA: Stanford University Press. *Nearly 100 present species illustrated in color; full natural history details; 12 fossil species illustrated in black & white.*

Ferris CD & FM Brown 1980.
Butterflies of the rocky mountain states. Norman, OK: Univ. of Oklahoma Press. *Species accounts, color and black & white illustrations and distribution maps of all the butterflies of the region. ($23, BQ).*

Forbes WTM 1923.
The Lepidoptera of New York and neighboring states, Part I. Primitive forms, microlepidoptera, pyraloids, bombyces. Ithaca, NY: Cornell Univ Agric Exper Sta Mem 68. *(1969 reprint) E. Lansing, MI: ERS.*

— 1948. The Lepidoptera of New York and neighboring states, Part II. Geometridae, Sphingidae, Notodontidae, Lymantriidae. Ithaca, NY: Cornell Univ Agric Exper Sta Mem 274.

— 1954. The Lepidoptera of New York and neighboring states, Part III. Noctuidae. Ithaca, NY: Cornell Univ Agric Exper Sta Mem 329.

— 1960. The Lepidoptera of New York and neighboring states, Part IV. Agaristidae through Nymphalidae, including butterflies. Ithaca, NY: Cornell Univ Agric Exper Sta Mem 371.

Garth JS & JW Tilden 1986.
California butterflies. Berkeley, CA: Univ. of California Press. *Covers more than 230 species and subspecies of butterflies, statewide, with concise descriptions; color and black & white illustrations. ($13, BQ).*

Gerberg EJ & RH Arnett 1989.
Florida butterflies. Baltimore: Natural Science Publications. *Species accounts and color illustrations of Florida butterflies, and a list of skippers; brief treatment of rearing, observing, gardening, etc. ($12, BQ).*

Harris L 1972. Butterflies of Georgia. Norman, OK: Univ. of Oklahoma Press. *Species accounts, distribution, flight periods, color and black & white illus. of all Georgia butterflies. ($8).*

Heitzman JR & JE Heitzman 1987.
Butterflies and moths of Missouri. Jefferson City, MO: Missouri Dept. of Conservation. *Excellent color illustrations of all the butterflies and about 5% of*

the moths of the State, with brief accounts of each species. ($13.50, BQ).

Iftner DC, JA Shuey & JV Calhoun 1992.
Butterflies and skippers of Ohio. Ohio Biological Survey, 1315 Kinnear Rd., Columbus, OH 43212. *A detailed account of the butterflies and skippers of the State, their distribution, and their biology. ($40).*

Kimball CP 1965. Lepidoptera of Florida. Gainesville, FL: Division of Plant Industry. *Color and black & white illustrations of Florida moths & butterflies, with locality records.*

Klassen P, R Westwood, B Preston & B McKillop 1989.
The butterflies of Manitoba. Winnipeg: Manitoba Museum of Man & Nature. *Full species accounts, distribution maps, and color plates of all 145 Manitoba species. ($19, BQ).*

Maza R de la & R Turrent D 1985.
Mexican Lepidoptera: Eurytelinae I. *($25 ERS).*

Minno MC & TC Emmel 1992.
Butterflies of the Florida Keys. Scientific Publishers, P. O. Box 15718, Gainesville, FL 32604. *Color illustrations of 106 species, including many life histories; general background information about the Keys. ($15).*

Morris RF 1980. Butterflies and moths of Newfoundland and Labrador. Ottawa: Agriculture Canada. *Species accounts, color illustrations and occurrence records of Lepidoptera of the region.*

Orsak L 1978. The butterflies of Orange County, California. Irvine, CA: Univ. of California. *A historical and current treatment of butterflies in the rapidly changing ecosystems of Orange County; includes drawings of larval foodplants. ($7.50, BQ).*

Rings RW, EH Metzler, FJ Arnold & DH Harris 1992.
The owlet moths of Ohio. Ohio Biological Survey, 1315 Kinnear Rd., Columbus, OH 43212. *Systematic and annotated checklist of 708 species recorded from Ohio, with color and black & white plates. ($20).*

Rockburne EW & JD Lafontaine 1976.
The cutworm moths of Ontario and Quebec. Ottawa: Agriculture Canada. *Species accounts and color illustrations of the noctuid moths (except deltoids) of the region. ($12).*

Royer RA 1988. Butterflies of North Dakota. Minot, ND: Minot State Univ. *Brief species treatments, distribution maps, color and black & white illus. of the 142 North Dakota species. ($15, BQ).*

Shapiro AM 1974.
The butterflies and skippers of New York State. Ithaca, NY: Cornell Univ. *Distribution and life histories. ($5, BQ).*

Shull EM 1987. The butterflies of Indiana. Indianapolis: Indiana Univ. Press. *Species accounts, color illus., and distribution maps of all the butterflies of the state. ($25, BQ).*

Taft WH, D Smitley & JW Snow 1991.
A guide to the clearwing borers (Sesiidae) of the north central United States. East Lansing, Michigan: North Central Regional Extension Publ.

Michigan State Univ., Rm 10b Ag. Hall, E. Lansing, MI 48824. *($7.50).*

Thomas JA 1986. Butterflies of the British Isles. Twickenham, England: Newnes Books. *A field guide using wild photos for identification, with strong ecological and phenological orientation.*

Tilden JW 1965. Butterflies of the San Francisco Bay region. Berkeley, CA: Univ. of California Press. *Detailed coverage of the area, more than half the species in color.*

Tveten J & G Tveten 1997.
Butterflies of Houston and southeast Texas. Austin: Univ of Texas Press. P.O. Box 7819, Austin TX 78713-7819. *275 color photos, line drawings. ($19.95).*

Woodbury EN 1994. Butterflies of Delmarva [Delaware, Maryland, Virginia peninsula]. Tidewater Publ., Centreville, MD 21617. Tel. 800-638-7641. *Skips skippers. ($13).*

3. Atlases, Checklists and Lists

Calhoun JV 1997. List of the butterflies and skippers of Florida (Lepidoptera: Papilionoidea and Hesperioidea). *Source: J. V. Calhoun, 977 Wicks Dr., Palm Harbor, FL 34684-4656. ($0.78, for postage).*

Ferris CD (ed.) 1989.
Supplement to: A catalogue checklist of the butterflies of America north of Mexico. Lepid Society Memoir 3. *Emends and updates the prior checklist of Miller & Brown 1981. ($6, LS).*

Grehan JR, BL Parker, GR Nielsen, DH Miller, JD Hedbor, M Sabourin & MS Griggs 1995.
Moths and butterflies of Vermont (Lepidoptera): A faunal checklist. Source: J. Grehan, Entomol. Res. Lab., Univ. of Vermont, PO Box 53400, Burlington, VT 05405-3400. *($8).*

Hamel DR 1991. Atlas of insects on stamps of the world. Falls Church, VA: Tico Press. *Black & white illustrations, heavily cross-indexed.*

Hinchliff J 1994. Atlas of Oregon butterflies. Corvallis, Oregon: Oregon State Univ. Bookstores, Inc., P.O.Box 489, 2301 SW Jefferson Ave., McCoy, OR 97339. *Distribution maps, foodplants, flight periods, checklist. ($15).*

Hodges RW et al. 1983.
Check list of the Lepidoptera of America north of Mexico. EW Classey and Wedge Entomol Res Foundation (WERF). *Includes more than 11,000 species of butterflies and moths, scientific names only. WERF, 85253 Ridgetop Drive, Eugene, OR 97405-9535 USA. ($28).*

Holmes AM, RR Tasker, QF Hess, & AJ Hanks 1991.
The Ontario butterfly atlas. Toronto Entomologists' Association, 34 Seaton Drive, Aurora, ON L4G 2K1, Canada. *Distribution maps and life history information for 138 species.*

Iftner DC & DM Wright 1996.
Atlas of New Jersey butterflies. *Checklist and plotted county maps of all NJ butterflies and skippers. D. C. Iftner, 8 Alpine Trail, Sparta, NJ 07871. ($5).*

Miller LD & FM Brown 1981.
A catalogue/checklist of the Butterflies of America north of Mexico. Lepid Soc Mem 2. *Current nomenclature, references to original species descriptions. (Update: see Ferris 1989). ($12, LS).*

Miller JY 1992. The common names of North American butterflies. Washington: Smithsonian Institution Press. *The first formal common names list, including less-used alternates; indexed by both common and scientific names. ($15, BQ).*

North American Butterfly Association (NABA) 1995.
Checklist and English names of North American butterflies. Morristown, NJ: NABA. *($5.00).*

Opler PA 1995. Lepidoptera of North America. 2. Butterflies (Papilionoidea and Hesperioidea) of the eastern United States. Contributions of the C. P. Gillette Museum of Arthropod Biodiversity, Colorado State University (CSU), Fort Collins, CO. Source: PA Opler, Department of Bioagricultural Sciences, CSU, Ft. Collins, CO 80523. *($14).*

— 1995. Lepidoptera of North America. 1. A county atlas of Saturniidae and Sphingidae of the eastern United States. Contributions of the C. P. Gillette Museum of Arthropod Biodiversity, Colorado State University (CSU), Fort Collins, CO. Source: PA Opler, Department of Bioagricultural Sciences, CSU, Ft. Collins, CO 80523. *($12).*

Peigler RS & PA Opler 1993.
Moths of western North America. 1. Distribution of Saturniidae of western North America. Contributions of the C. P. Gillette Museum of Arthropod Biodiversity, Colorado State University (CSU), Fort Collins, CO. Source: PA Opler, Department of Bioagricultural Sciences, CSU, Ft. Collins, CO 80523. *($3.50).*

Powell JA & PA Opler 1996.
Moths of western North America. 3. Distribution of "Oecophoridae" (sense of Hodges 1983) of western North America. Contributions of the C. P. Gillette Museum of Arthropod Biodiversity, Colorado State University (CSU), Fort Collins, Colo. Source: PA Opler, Department of Bioagricultural Sciences, CSU, Ft. Collins, CO 80523. *($6).*

Smith MJ 1995. Moths of western North America. 2. Distribution of Sphingidae of western North America. Contributions of the C. P. Gillette Museum of Arthropod Biodiversity, Colorado State University (CSU), Fort Collins, CO. Source: PA Opler, Department of Bioagricultural Sciences, CSU, Ft. Collins, CO 80523. *($5).*

Stanford RE & PA Opler 1993.
Atlas of western USA butterflies, including adjacent parts of Canada and Mexico. *Distribution dot-maps, by counties, of all species found west of the 100th meridian and within 100 miles beyond the USA borders. Available from RE Stanford, 720 Fairfax St., Denver, CO 80220-5151. ($17 postpaid).*

4. Butterfly Watching

Glassberg J 1993. Butterflies through binoculars. New York: Oxford University Press. *A field guide to butterflies in the Boston-New York-Washington region. Wild color photos of all species. ($16).*

Matthews P 1957. The pursuit of moths and butterflies: An anthology. London: Chatto & Windus.

Pyle RM 1974. Watching Washington butterflies. Seattle: Seattle Audubon Society. *Color photos of live butterflies, good species descriptions, and discussion of faunal zones within the State.*

— 1991. Handbook for butterfly watchers. Boston: Houghton Mifflin. *A personal account by a life-long butterfly watcher ($12). Formerly published as: Pyle RM 1984.* The Audubon Society handbook for butterfly watchers. *New York: Charles Scribner's Sons.*

5. Gardening

Ajilvsgi, G 1991. Butterfly gardening for the South. Dallas, TX: Taylor Publishing Co. *Informative text on both plants and butterflies (applicable to any area), excellent color photographs, resource lists, including suppliers of seeds and plants for the region. ($35).*

Damrosch B 1982. Theme gardens. New York: Workman Publishing. *Contains a section on layouts for butterfly gardens. ($11).*

Dennis JV 1985. The wildlife garden. New York. Alfred A. Knopf Inc. *Includes chapters on butterflies and moths and lists of nectar plants. ($18).*

— & M Tekulsky 1991. How to attract hummingbirds and butterflies. Ramon, CA: Ortho Books. (Chevron Chemical Company, Consumer Products, P.O. Box 5047, San Ramon, CA 94583.) *($11.95).*

Huegel C 1991. Butterfly gardening with Florida's native plants. Florida Native Plant Society, P.O. Box 680008, Orlando, FL 32868. *Covers the butterflies themselves and nectaring and larval food plants. ($6).*

Levicoff J & M Stellen 1994. The butterfly garden. Storytellers Ink, P. O. Box 33398, Seattle, WA. *A participating/story book, pre-kindergarten to 10 yrs. ($10.95).*

Loewer P 1992. The evening garden. New York: Macmillan. *Information on night-fragrant plants and their attraction for moths.*

Newman H 1967. Create a butterfly garden. London: John Baker. *Butterfly gardening principles applicable in any temperate climate.*

Ross GN 1994. Gardening for butterflies in Louisiana: A guide for gardeners, educators and enthusiasts. *LA Dept. Wildlife & Fisheries, NHP, Wildlife Div., PO Box 98000, Baton Rouge, LA 70898-9000. ($5.50).*

Rothschild M & C Farrell 1983. The butterfly gardener. London: Michael Joseph/Rainbird. *An account by two veteran British butterfly gardeners, applicable to any temperate climate; includes greenhouse butterfly gardening.*

Ruffin J 1993. Where are the butterfly gardens? *Lepid. Soc. publication listing butterfly gardens open to the public. ($5.75, LS).*

Sonoran Arthropod Studies Institute (SASI) 1996.
Desert butterfly gardening. SASI, P.O.Box 5624, Tucson, AZ 85703. *Covers the common butterflies of the Southwest and their preferred larval food and adult nectaring plants.*

Sedenko J 1991. The butterfly garden. New York: Villard Books. *By an accomplished gardener with limited butterfly background; good material on choices of garden plants. ($25, YES).*

Stokes D, L Stokes & E Williams 1991.
The butterfly book. Boston: Little, Brown and Co. *A basic guide to butterfly gardening, identification, and an outstanding section on behavior. Excellent color photos. ($11, YES).*

Tekulsky M 1985. The butterfly garden. Boston: The Harvard Common Press. *($10, BQ).*

Xerces Society/Smithsonian Institution 1998.
Butterfly gardening, 2nd ed. San Francisco: Sierra Club Books. *General information on butterfly biology and on gardening for butterflies, by various authors; many color photos. ($24, XS).*

6. Rearing and Immatures

Anderson TE & NC Leppla 1992.
Advances in insect rearing for research and pest management. Boulder, CO: Westview Press.

Barnes WM & J McDunnough 1918.
Illustrations of the North American species of the genus *Catocala*. Memoirs of the American Museum of Natural History, New Series, Vol. III, Part I. *Detailed adult and larval color drawings.*

Collins MM & RD Weast 1961.
Wild silk moths of the United States: Saturniinae. Experimental studies and observations of natural living habits and relationships. Cedar Rapids, IA: Collins Radio Co.

Dirig R 1975. Growing moths. Ithaca, NY: 4-H Members' Guide M-6-6, Cornell Univ. *Procedures for rearing the larger moths. ($2.50, BQ).*

Fitzgerald TD 1995.
The tent caterpillars. Ithaca, NY: Cornell series in arthropod biology, Comstock Publ. Assoc. *Color and black & white illustrations; rearing and classroom advice. (Hardcover, $37.95).*

Friedrich E 1986. Breeding butterflies and moths (English Edition with additional material edited by A. Maitland Emmet). Colchester, England: Harley Books. *A detailed work on breeding and rearing, with information on specific requirements of many European species; techniques are applicable and worth testing on quirkier species worldwide.*

Gardiner BOC 1982.
A silkmoth rearer's handbook. Colchester, England: Amateur

Entomologists' Society. *A study of the world's silkmoths.*

Guilbot R 1982. Élevage des papillons. Paris: Société Nouvelle des Éditions Boubée. *Detailed consideration of rearing and the problems thereof; excellent larval photos; in French.*

McCabe TL 1991. Atlas of Adirondack caterpillars. Albany, NY: New York State Museum Bull. 470. *178 species of New York State caterpillars described and illustrated with drawings and black & white photographs; flight periods, diapause, and food plants accepted and rejected. ($20).*

Packard AS 1876. A monograph of the geometrid moths or Phalaenidae of the United States. US Geological Survey of the Territories. Washington, DC: Government Printing Office.

— 1895. Monograph of the Bombycine moths of America north of Mexico, including their transformations and origin of the larval markings and armature. Part I. Family 1. Notodontidae. National Academy of Sciences. Vol 7. First Memoir on the Bombycine Moths. Washington, DC: Govt. Printing Office. *Outstanding larval color drawings.*

— 1905. Monograph of the Bombycine moths of America north of Mexico, including their transformations and origin of the larval markings and armature. Part II. Family Ceratocampidae, subfamily Ceratocampinae. National Academy of Sciences. Vol 9. Washington, DC, Govt. Printing office. *Outstanding larval color drawings.*

Singh P 1977. Insect colonization and mass production. New York: Academic Press.

Smith CN 1966. Artificial diets for insects, mites and spiders. New York: Plenum.

Stehr FW (ed.) 1987. Immature insects. Dubuque, IA: Kendall Hunt Publishing Co. *Major text on immature insects of all orders, including Lepidoptera in Vol. I. ($92, BQ).*

Stone JLS & HJ Midwinter 1975. Butterfly culture: A guide to breeding butterflies, moths and other insects. Poole, Dorset, U.K.: Blandford Press.

Stone S 1991. Foodplants of world Saturniidae. Lepid Soc Memoir 4. *Foodplant information on more than 500 species and subspecies of worldwide Saturniidae. ($7.20, LS).*

Tietz HM 1972. An index to the described life histories, early stages, and hosts of the macrolepidoptera of the continental United States and Canada. 2 vols. Sarasota, FL: Allyn Museum of Natural History. *A very valuable reference, even though some of the material quoted was erroneous. ($25, ERS).*

Villiard P 1969. Moths and how to rear them. New York: Funk & Wagnalls. *Practical methods for rearing moths of various families.*

Wright AB 1993. Peterson first guide to caterpillars of North America. Boston: Houghton Mifflin. *Illustrations of 120 mostly common U.S. species of butterflies and moth larvae, the adults they produce, and their growth habits and foodplants. ($5, Peterson Field Guides).*

7. Not classified.

Allan PBM 1943. A moth hunter's gossip, 2nd ed. London: Watkins & Doncaster. *A very readable and entertaining account of various aspects of moth collecting.*

— 1948. Moths and memories. London: Watkins & Doncaster.

— 1975 (1943). Talking of moths. Faringdon, UK: E. W. Classey. *Reprint.*

— 1980. Leaves from a moth hunter's notebook. Faringdon, UK: E. W. Classey. *Reprint.*

Beirne BP 1955. Collecting, preparing, and preserving insects. Canad. Dept. Agric. Publ. 932.

Borror DJ, CJ Triplehorn & NF Johnson 1989. Introduction to the study of insects, 6th ed. New York: Holt, Rinehart, & Winston. *A general entomology text delineating the characteristics and interrelationships of the various orders of insects; extensive glossary. ($59.50, BQ).*

Brewer J 1976. Butterflies. New York: Harry Abrams. *Striking photographs by K. Sandved, with very informative text by Brewer; not for identification of butterflies.*

— & D Winter 1986. Butterflies and moths: A companion to your field guide. New York: Prentice Hall. *A detailed introduction to the enjoyment of Lepidoptera. Color and black & white illustrations.*

IC Callaway Foundation 1991. Discover butterflies: An activity book for families, students, and teachers. Callaway Gardens, Pine Mountain, GA 31822. *($7.95).*

Common IFB 1990. Moths of Australia. Victoria: Melbourne Univ Press. *The outstanding source for biologic and taxonomic information on the families of moths, worldwide. (ca $200 US, E.J. Brill).*

Dickson R 1992. A lepidopterist's handbook, 2nd ed. Colchester, England: Amateur Entomologists Society.

Hunt J 1992. A shimmer of butterflies. San Luis Obispo, CA: Blake Publishing. *Brief, well-illustrated description of butterfly biology for the novice. ($8, YES).*

Klass C & R Dirig 1992. Learning about butterflies. Ithaca, NY: Cornell Cooperative Extension Publication, 4-H Member/Leader Guide 139-M-9. *Butterfly structure, biology, behavior, conservation, and "Suggested Projects for Personal Discovery;" NY state butterfly list. ($6.25).*

Nichols SW 1980. The Torre-Bueno glossary of entomology. New York: New York Entomological Society and American Museum of Natural History. *Revised edition of J.R. de la Torre-Bueno's Glossary of Entomology, including Supplement A by G.S. Tulloch.*

Opler PA & S Strawn 1988. Butterflies of the American West: A coloring album. Boulder, CO: Roberts Rinehart. *($5.45, XS).*

— 1989. Butterflies of eastern North America: A coloring album and activity book. Boulder, CO: Roberts Rinehart. *($6.45, XS).*

Pyle RM 1993. The thunder tree: Lessons from an urban wildland. New York: Houghton Mifflin. *Butterfly oriented essays on natural history, focusing on the Highline Canal in Denver. ($20).*

Sbordoni V & S Forestiero 1984 (English reissue 1998). Butterflies of the World. Buffalo, NY: Firefly Books. *Selected species of moths and butterflies and their larvae, color illustrated, broad discussion of all aspects of biology, behavior, distribution, interaction with man; highly informative. ($40).*

Scoble MJ 1992. The Lepidoptera: Form, function, and diversity. New York & London: Oxford University Press.

Swanson HF 1998. Twenty years of butterfly revelations. Privately published. *A running 20-year account of the author's experiences with red admirals in his backyard. Source: Presbyterian Women, First Presbyterian Church, 106 E. Church St., Orlando, FL 32801-3390. ($10).*

Wallace AR 1869. The Malay Achipelago. London: MacMillan & Co. (New York: Dover Publ. reprint, 1962). *Includes accounts of tropical moth and butterfly collecting in the mid-19th century.*

Whalley P 1988. Butterflies and moths. New York: Alfred A. Knopf. *Photos and behavioral details about many species of butterflies and moths, worldwide. ($15, YES).*

8. Special Subjects

Brewer J 1967. Wings in the meadow. Boston: Houghton Mifflin. *A detailed story of the life history of a monarch butterfly (before discovery of the overwintering site).*

Brown GH (ed.) 1996. Chasing butterflies in the Colorado Rockies with Theodore Mead in 1871. *Mead's personal narrative with notes by F. Martin Brown. Colorado Outdoor Education Center, P.O. Box 167, Florissant, CO 80816. ($9).*

Douglas M 1986. The lives of butterflies. Ann Arbor, MI: Univ. of Michigan Press. *Treats the lifestyles, habits, and survival strategies of butterflies. ($45, BQ).*

Dunn GA 1991. International entomology resource guide, 3rd ed. Lansing, MI: Young Entomologists' Society. Spec. Publ. 4.

Feltwell J 1995. The conservation of butterflies in Britain: Past and present. Wildlife Matters, Marlham, Henley's Down, Battle, East Sussex, TN33 9BN, UK. *($17 US).*

Grace ES 1997. The nature of monarch butterflies: Beauty takes flight. Greystone Books, Douglas & McIntyre, 1615 Venables St., Vancouver, BC, Canada V5L 2H1. *Coffee-table book, authoritative text covering biology, attrition, conservation. ($32.50 Canadian, hardcover).*

Malcolm SB & MP Zalucki 1993. Biology and conservation of the monarch butterfly. Los Angeles: Natural History Museum of Los Angeles County. *($90).*

McMahon J. 1991. Studying butterfly populations in urban areas. Lansing, MI: Michigan Entomological Society. Entomology Notes No. 23.

New TR 1991. Butterfly conservation. New York: Oxford Univ. Press. *Discussion of the problems of conservation at the levels of single species, local ecosystems, and entire faunas. Extensive bibliography. ($30).*

Nijhout J F 1991. The development and evolution of butterfly wing patterns. New York: Villard. *($20, BQ).*

Owen DF 1971. Tropical butterflies. London: Oxford Univ. Press. *Extensive treatment of biology, behavior, and ecology of various tropical African butterflies; excellent for general background. (£4.25).*

Scriber MJ, Y Tsubaki & RC Lederhouse 1995.
Swallowtail butterflies: Their ecology and evolutionary biology. Scientific Publishers, P.O. Box 15718, Gainesville, FL 32604. *Consolidation of the primary scientific literature, enjoyable even by nonscientists. ($65, hardcover).*

Williams CB 1930.
The migration of butterflies. Edinburgh & London: Oliver & Boyd.

9. Videos

Anon. 1996. Diversity in the rainforest. Peru; 50 min. NTSC (VHS) format. Faringdon, UK: E. W. Classey.; for address see Appendix L, Dealers. *($24.95 +$7).*

Audubon Society 1995a.
Audubon Society's butterflies for beginners. *Live footage in natural habitat, including immatures, of 33 wide-spread North American species. (VHS, $20).*

—1995b. Audubon Society's butterfly gardening. *Shows growing conditions and cultivation of 25 of the best North American nectaring plants and the butterfies that favor them. ($25).*

Brower LP 1995. Strategy for survival: Behavioral ecology of the monarch butterfly. *Videocassette of 1977 prize-winning film. Available from Lincoln P. Brower, Sweetbriar College, Sweet Briar, VA 24595. ($35).*

Callaway Gardens Education Department, Pine Mountain, GA 31822.
Discover butterflies. *Video and workbook, elementary school level. ($25).*

Ebner J 1992. Butterflies of Wisconsin. E/D Productions, P.O.Box 556, Okauchee, WI 53069. *Field-recorded accounts of 49 Wisconsin species. ($40).*

Maza R de la Jr 1995.
Mexican butterflies. (Eng. or Span.), CD-ROM. Editiones en Technicología Avanzada, Attn. John Stark, Camino Real a Xochimilco No. 60 Tepepan, Xochimilco, México D.F. 16020. *($50 US).*

Walton RK 1996a. Common butterflies of the Northeast. (30 min. VHS); address below.

— 1996b. Skippers of the Northeast. (48 min. VHS). Richard K Walton, 7 Concord Greene No. 8, Concord MA 01742. *($19.95 + $3 each, or $35 postpaid for pair; + 5% tax within MA).*

Appendix K

Organization Lists

1. Butterfly Houses and Insect Zoos

This list covers the United States and Canada and is alphabetical by province or state.

Canada

Butterfly World, Coombs, near Parksville, Vancouver Island, BC V0R 1M0, Canada. Tel. 604-248-7026.
Okanagon Butterfly World, 1190 Stevens Rd., Kelowna, BC V1Z 1G1, Canada. Tel. 604-769-4408.
Victoria Butterfly Gardens, PO Box 190, Brentwood Bay, BC V8M 1R3. Tel. 604-652-3822.
Metro Toronto Zoo, 361 A Old Finch Ave., Scarborough, ONT M1B 5K7.
Niagara Parks Butterfly Conservatory, 7400 Portage Rd. South, Box 150, Niagara Falls, ONT L2E 6X8.
Montreal Botanical Garden and Insectarium, 4101 Sherbrooke St. East, Montreal, QUE H1X 2B2. Tel. 514-872-1400.

USA

Marine World Africa USA, Butterfly World, Vallejo, CA 94589. Tel. 707-644-4000 x 270.
Natural History Museum of Los Angeles Co., Insect Zoo, 900 Exposition Blvd., Los Angeles, CA 90007-4057.
San Diego Wild Animal Park, The Hidden Jungle, 15500 San Pasqual Valley Rd., Escondido, CA 92027. Tel. 619-234-6541.
Butterfly Pavilion, 6252 West 104th Ave., Westminster, CO 80020. Tel. 303-469-5441.
Butterflies in Flight, 6151 Everett St., Naples, FL 33940. Tel. 941-793-2359.
Butterfly World, Tradewinds Park South, 3600 West Sample Road, Coconut Creek, FL 33073. Tel. 305-977-4400.
Wings of Wonder, Cypress Gardens, SR 540, Cypress Gardens, FL 33884. Tel. 800-282-2123.
Day Butterfly Center, Callaway Gardens, Pine Mountain, GA 31822. Tel. 706-663-2281.
Audubon Zoological Garden, PO Box 4327, New Orleans, LA 70178. Tel. 504-861-2537.
The Butterfly Place, 120 Tyngsboro Road, Westford, MA 01886. Tel. 508-392-0955.
Butterfly-Hummingbird Garden, Detroit Zoological Park, 8450 West Ten Mile Road, PO Box 39, Royal Oak, MI 48068. Tel. 810-398-0903.
Mackinac Island Butterfly House, 1308 McGulphin St., Mackinac Is. MI 49757. Tel. 906-847-3972.
St. Louis Zoological Park, St. Louis, MO 63110. Tel. 314-781-0900.
Butterfly Zone, The Bronx Zoo, 2300 Southern Blvd., Bronx, NY 10460. Tel. 718-367-1010.
Cincinnati Insect Zoo Insectarium, 3400 Vine St., Cincinnati, OH 45220. Tel. 513-281-4701 x 8348.
Cleveland Metroparks Zoo, Tropical Butterfly Garden, 3900 Brookside Park Drive, Cleveland, OH 44109. Tel. 216-661-6500.
Cockrell Butterfly Center, Houston Museum of Natural Science, 1 Herman Circle Drive, Houston, TX 77030. Tel. 713-639-4629.
Mercer Arboretum & Botanic Garden, 22306 Aldine-Westfield, Humble, TX 77308. Tel. 713-443-8731. Small exhibit, emphasizing immatures.
Moody Gardens, Rainforest Pyramid, 1 Hope Blvd., Galveston, TX 77554. Tel. 800-582-4673.
Utah's Hogle Zoo, Butterfly World, 2600 E. Sunnyside Ave., Salt Lake City, UT 84108. Tel. 801-582-1631.

2. Organizations for Lepidopterists

The organizations listed here alphabetically are open to anyone interested in Lepidoptera, or in the case of an "entomological" organization, in any aspect of entomology.

Amateur Entomologists' Society, The Hawthornes, Frating Road, Great Bromley, Colchester, Essex CO7 7JN, England.

American Entomological Society, 1900 Race Street, Philadelphia, PA 19013. Publishes Entomological News *and* Memoirs of the American Entomological Society.

Association for Tropical Lepidoptera, c/o Florida State Collection of Arthropods, P. O. Box 141210, Gainesville, FL 32614. Publishes Tropical Lepidoptera, Holarctic Lepidoptera, *and a newsletter.*

Cambridge Entomological Club, 16 Divinty Ave., Cambridge, MA 02138. Publishes Psyche. *Monthly meetings during the academic year.*

Connecticut Entomological Society, Dept. of Biology, Yale Univ., P.O. Box 6666, New Haven, CT 06520.

Entomological Society of America, 9301 Annapolis Road, Suite 300, Lanham, MD 20706. Puts out numerous publications including American Entomologist. *Membership: professional entomologists. "ENT NET" volunteers available for public presentations.*

Florida Entomological Society, P.O. Box 7326, Winter Haven, FL 33883-7326. Professionals and amateurs, incl. lepidopterists. Caribbean area covered; occasional meetings. Publishes Florida Entomologist.

4-H Clubs: Listed in many local phone books (look up under "Four" in alphabetical business listing); or contact your state land grant university for 4-H or Cooperative Extension Program.

Hawaiian Entomological Society, Dept. of Entomology, 3050 Maile Way, Univ. of Hawaii, Honolulu, HI 96822.

High Country Lepidopterists, c/o Paul A. Opler, 3354 Valley Oak Drive, Loveland, CO 80538-8921. Group meets annually.

Idaho Entomological Group, c/o Smith Museum of Natural History, Albertson College of Idaho, Caldwell, ID 83605. Meetings, field trips, newsletter.

Idalia Society of Mid-American Lepidopterists, 4637 West 69th Terrace, Prairie Village, KS 66208-2547. Publishes Idalia, *a newsletter.*

Insect Migration Association, Univ. of Toronto, Scarborough Campus, Scarborough, ONT M1C 1A4, Canada. Conducts monarch tagging program.

Journey North, 125 North First St., Minneapolis, MN 55401. Tel. 612-339-6959. Monarch migration surveillance.

Kansas Entomological Society, Snow Entomological Museum, University of Kansas, Lawrence, KS 66045.

Kentucky Lepidopterists, c/o Dept. of Biology, Univ. of Louisville, Louisville, KY 40292-0001. Publishes Kentucky Lepidopterist.

Lepidoptera Research Foundation, c/o Santa Barbara Museum of Natural History, 2559 Puesta Del Sol Road, Santa Barbara, CA 93105. Publishes Journal of Research on the Lepidoptera *and a newsletter. (Not a "group-type" organization.)*

Lepidopterists' Society, Ernest H. Williams, Secretary, Dept. of Biology, 198 College Hill Road, Hamilton College, Clinton, NY 13323 USA. Publishes News, Journal *and* Memoirs. *(Default address: Natural History Museum of Los Angeles County, 900 Exposition Blvd., Los Angeles, CA 90007-4057). Pacific Slope Section: All Lepid. Soc. members with ZIP codes 80000 or higher are automatic members, as are also all members from western Canadian provinces and western Mexican states. Meets any year when Lep. Soc. annual meeting is in the East.*

Lorquin Entomological Society, c/o Insect Zoo Director, Natural History Museum of Los Angeles County, 900 Exposition Blvd., Los Angeles, CA 90007-4057. Aimed at study and conservation of arthropods through education; eight meetings per year; newsletter.

Maryland Entomological Society, c/o Dept. of Biol. Sciences, University of Maryland, Baltimore, 5401 Wilkens Ave., Baltimore, MD 21228-5348. Formal and field meetings; education and habitat preservation.

Michigan Entomological Society, c/o Dept. of Entomology, Michigan State Univ., East Lansing, MI 48824. Publishes newsletter, Entomology Notes, *and* Great Lakes Entomologist. *Meetings and field trips.*

Mississippi Entomological Society, Extension Entomology, P.O. Box 5426, Mississippi State, MS 39762.

Monarch Butterfly Sanctuary Foundation, 2078 Skillman Ave., Roseville, MN 55113. Overwintering-grounds conservation; local economics; resource lists.

Monarch Migration Association of North America, 7 Concord Greene #8, Concord, MA 01742. Newsletter, census taking, tagging.

Monarch Program, P. O. Box 178671, San Diego, CA 92177. Tel. 619-944-7113. Tagging program, newsletter, butterfly vivarium.

Monarch Watch, Dept. of Biology, Univ. of Kansas, Haworth Hall, Lawrence, KS 66045. Biological information, educational programs, bibliography.

New York City Butterfly Club, 28 West 9th Road, Broad Channel, NY 11693. Publishes a newsletter, The Mulberry Wing. *Study and conservation of Lepidoptera.*

New York Entomological Society, Dept. of Entomology, American Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192.

North American Butterfly Association (NABA), 4 Delaware Road, Morristown, NJ 07960. Publishes a newsletter,

Anglewing; *a serial*, American Butterflies; *and the* Fourth of July Butterfly Count Report.

Northwest Lepidopterists' Association, Dept. of Biology, Western Oregon State College, Monmouth, OR 97361. A loose group for gathering regional data, open to all; annual workshop meeting.

Ohio Lepidopterists, 1241 Kildale Square North, Columbus, OH 43229. Publishes a newsletter, The Ohio Lepidopterist.

Oregon Entomological Society, c/o Dept. of Entomology, Oregon State Univ., Cordley Hall 2046, Corvallis, OR 97331. Amateurs and professionals, includes lepidopterists; meeting, field trip.

Pacific Coast Entomological Society, c/o California Academy of Sciences, Golden Gate Park, San Francisco, CA 94118-4599.

Societas Europaea Lepidopterologica, c/o Treasurer, Volpinistrasse 72, D-80638, Munich, Germany (or c/o Dept. Biology, Univ. of Louisville, Louisville, KY 40292-0001, to process your dues in US $ instead of DM). Publishes a newsletter and a journal.

Sociedad Mexicana de Lepidopterologia, A.C., Elena 182 Col. Nativitas 03500 Mexico, D.F.; or c/o Luis G. Lopez del Paso, Concepcion Beistegui 1819, Narvarte, Mexico D.F.

Sonoran Arthropod Studies, Inc. (SASI), P. O. Box 5624, Tucson, AZ 85703. Publishes Backyard Bugwatching.

South Carolina Entomological Society, P.O. Box 582, Clemson, SC 29633-0582.

Southern Lepidopterists' Society, 5421 NW 69th Lane, Gainesville, FL 32653. Publishes a newsletter, Southern Lepidopterists' News.

Southwestern Entomological Society, Department of Entomology, Texas A & M Univ., College Station, TX 77843.

Tennessee Entomological Society, Dept. of Entomology and Plant Pathology, Univ. of Tennessee, Knoxville, TN 37901.

Toronto Entomologists' Association, 34 Seaton Drive, Aurora, ONT L4G 2K1, Canada. Study of Lepidoptera of Ontario. Meetings, field trip, annual summary.

Utah Lepidopterists Society, 7136 South 2200 West, West Jordan, UT 84084. Study of Lepidoptera, of Utah in particular. Publishes a bulletin; meetings, field trips.

Washington State Entomological Society, Dept. of Entomology, Washington State Univ., Pullman, WA 99164-6432.

West Virginia Entomological Society, W. Va. Dept. of Agriculture, 1900 Kanawha Blvd. East, Charleston, WV 25305-0191. Meetings, newsletter.

Wisconsin Entomological Society, Dept. of Entomology, Univ. of Wisconsin, 237 Russell Labs, 1630 Linden Drive, Madison, WI 53706. Tel. 608-262-6510. Newsletter, meetings; open.

Xerces Society, 4828 SE Hawthorne Blvd., Portland, OR 97215-3252. Devoted to conservation of invertebrates, various publications including the serial Wings.

Young Entomologists' Society, Inc., 1915 Peggy Place, Lansing, MI 48910. Various publications and educational materials.

Appendix L

Dealers and Suppliers

Abbreviations:

B	Books
C	Cabinets
E	Equipment (durable goods)
ED	Educational materials
K	Chemicals
L	Livestock, life-cycle kits
P	Insect pins
PH	Pheromones
S	Supplies (consumable goods)
SP	Specimens, papered

Amazon Adventures, 2458 Shore Blvd. West, Columbus, OH 43232. Tel. 614-864-9089. Rain forest excursions.

American Biological Supply Co., 2405 NW 66th Court, Gainesville, FL 32606-1633. Tel. 904-377-3299. B, C, E, K, P, S.

American Science & Surplus, 3605 Howard Street, Skokie, IL 60076. Tel. 708-982-0870. Odds and ends of useful equipment: plastic and glass vials, plastic droppers, watch glasses, corks.

Antiquariaat Junk, Van Eeghenstraat 129, 1071 GA Amsterdam, Netherlands. B, esp. rare; good response to want lists.

Antiquariat Goecke & Evers, Inh. Erich Bauer, Sportplatzweg 5, D(W)-7538 Keltern-Weiler, Germany. B, esp. European publications.

Apollo Books, Kirkeby Sand 19, DK-5771 Stenstrup, Denmark. Fax: 45 62 26 37 80. Entomological books.

Asher Rare Books, P. O. Box 258, NL-1970 AG IJmuiden, Netherlands. Antiquarian books, including Lepidoptera.

BioQuip Products, Inc., 17803 LaSalle Avenue, Gardena, CA 90248. Tel. 301-324-0620. B, C, E, P, S, SP.

Brill, E.J., 24 Hudson Street, Kinderhook, NY 12106. Specialty books.

Butterfly Farm, S.A., Apartado 323-6150 Santa Ana, Hacienda Los Reyes, La Guacima de Alajuela, Costa Rica. Ph/fax 011-506-438-0115. L, SP.

Butterfly World, Tradewinds Park, 3600 W. Sample Road, Coconut Creek, FL 33073. Tel. 305-563-7925. "Livestock: H. charitonius *only, minimum order $200 + shipping."*

C.P.M. Inc., Dallas, TX 75238-1337. Fine artists' brushes for genitalia preparations.

Carolina Biological Supply Co., 2700 York Road, Burlington, NC 27215. Tel. 800-334-5551. B, C, E, ED, K, L, P, S.

Century Photo Products and Accessories, 205 S. Puente St., Brea, CA 92821. Tel. 800-767-0777. Polypropylene filing pages for prints and negatives.

Classey, E. W., Ltd., P. O. Box 93, Faringdon, Oxon, SN7 7DR, England. Tel. 011 44 1367 244700, 24-hr. message service. B, including want lists; ED.

Combined Scientific Supplies, P.O. Box 1446, Fort Davis, TX 79734. Tel. 915-426-3851. SP.

Connecticut Valley Biological Supply Co., P. O. Box 326, Southampton, MA 01073. Tel. 800-628-7748. B, E, ED, K, L, P, S.

Deco Enterprise, P. O. Box 155, Taiping, Porak, Malaysia. SP.

Delta Education, Box M, Nashua NH 03061-6012. Tel. 800-442-5444. ED, L.

Edmund Scientific Co., Dept. 14D6, C915 Edscorp Bldg., Barrington, NJ 08007. Tel. 609-547-8880. Miscellaneous equipment items not found elsewhere.

Entomological Reprint Specialists, P.O. Box 77224, Dockweiler Station, Los Angeles, CA 90007-0224. Distributes The Moths of America North of Mexico *series, and other specialty publications.*

Fine Science Tools, Inc., 323-B Vintage Park Drive, Foster City, CA 94404. Tel. 800-523-2109. Forceps; 30-, 31-, and 33-gauge hypodermic needles; free catalog.

Great Lakes IPM, 10220 Church Road NE, Vestaburg, MI 48891. Tel. 517-268-5693 or -268-5911. PH and pheromone traps.

Hart, Len, 51 Benton Road, South Shields, Tyne & Wear NE34 9UB, England. Tel. day 191 5369599; eve. 191 4220494. S; $5 for list.

Holbrook Travel, 3540 NW 13th St., Gainesville, FL 32609. Tel. 352-377-7111. Lepidoptera excursions.

Holland Photo, 1221 S. Lamar, Austin, TX 78704. Tel. 512-442-4274. Professional grade photo processing.

Ianni Butterfly Enterprises, P.O. Box 81171, Cleveland, OH 44181. Tel. 216-888-2310 or -888-9763. Licensed importer-exporter, worldwide specimens with data, including certification as necessary. E, P, SP.

Insect Lore Products, P.O. Box 1435, Shafter, CA 93263. Tel. 800-LIVE BUG. Bombyx mori *silkworm eggs. B, ED, L, grade-school oriented.*

Jordan Paper Box Co., 5045 West Lake St., Chicago, IL 60644. Tel. 312-287-5362. Riker mounts and various storage and exhibit boxes.

Kahn, Bruce (NJ—prefers phone contact). Tel. 908-889-1759. Broker for all chemicals, in small retail lots; referred by Fisher Scientific, tel. 800-766-7000.

Keystone Universal Corp., Box 3241, 18400 Rialto, Melvindale, MI 48122. Tel. 313-388-0063. Retailer for ammonium carbonate.

Kirk Enterprises, 4370 E. US Highway 20, Angola, IN 46730. Tel. 219-665-3670. Kirk close-up photo bracket.

Koehn, LeRoy C., 6085 Wedgewood Village Circle, Lake Worth, FL 33463-7371. Tel. 561-966-1655. Bait traps and light traps.

Lane Science Equipment Corp., 225 West 34th Street, Suite 1412, New York, NY 10122. C.

Mason Box Co., 521 Mt. Hope Street, Attleboro Falls, MA 02763. Tel. 800-225-2708. Boxes suitable for storing transparencies.

MCM Electronics, 650 Congress Park Drive, Centerville, OH 45459-4072. Tel. 800-543-4330 "Dual helping hands" holding jig for photographing larvae.

Merle's Monarchs, 1030 Neil Creek Road, Ashland, OR 97520. Monarch livestock.

Montes Azules S.A. de C.V., attn. John Stark, Camino Real a Xochimilco #60, 16020 Mexico D.F., Mexico. Tel. 52-5-420-5959, Chiapas specimens with permits, no CITES or U.S. endangered.

Nasco, 901 Janesville Avenue, Fort Atkinson, WI 53538, and Nasco West, 1524 Princeton Avenue, Modesto, CA 95352. Tel. 800-558-9595. B, C, E, ED, K, L, P, S

National Allergy Supply, P. O. Box 1658, Duluth, GA 30136.

Natural History Booksellers, 121 Hickory Hill Road, Kensington, CT 06037. Tel. 203-225-5353. Buys and sells scholarly natural history books, and periodicals.

Nature Discoveries, 389 Rock Beach Road, Rochester, NY 14617. Tel. 716-544-8198 or -865-4580. ED, L, SP.

Nebraska Scientific, 3823 Leavenworth St., Omaha, NE 68105. Tel. 402-346-7214. B, E, K, L, P, S.

NegaFile Systems, Inc., P.O. Box 78, Furlong, PA 18925. Tel. 215-348-2356. Slide file drawers for transparencies and all kinds and sizes of filing. Retail sales direct.

NoIR Medical Technologies, P. O. Box 159, South Lyon, MI 48178. Tel. 800-521-9746. Ultraviolet protective goggles. Will sell retail if you (1) request catalog, then (2) send in order and payment,

along with note from physician such as: "Please provide one Model ___ UV Shield for protection against avocational exposure to UV."

Osram Sylvania, Inc., National Customer Support Center, P. O. Box 275, Westfield, IN 46074. Tel. 800-LIGHTBULB for information about mercury vapor and fluorescent lights.

Papillon Distributors, Inc., The Butterfly Place, P. O. Box 1541, 120 Tyngsboro Rd., Westford, MA 01886-4995. Tel. 508-392-0955. B, L.

Poly Sciences, 400 Valley Road, Warrenton, PA 18976-2590. Tel. 800-523-2575. Stains for genitalia preparations.

Print File, Inc., P. O. Box 607638, Orlando, FL 32860-7638. Archival preservers for prints, slides, and negatives.

Schoolmasters Science, 745 State Circle, Box 1941, Ann Arbor, MI 48106. Tel. 800-521-2832. B, E, ED, S.

Serrano, Miguel, Tropical Butterflies, 6823 Rosemary Drive, Tampa, FL 33625. Tel. 813-963-6074. Papered tropical specimens, Florida livestock.

Sigma Chemical Co., P. O. Box 14508, St. Louis, MO 63178-9916. Tel. 800-325-3010. Usually ships only to laboratories, but will sometimes accept credit cards. Stain for genitalia preparations.

Southern Biological Supply Co., P. O. Box 68, McKenzie, TN 38201. Tel. 800-748-8735. B, E, K, P, S.

Staples, John, Breeder of Lepidoptera, 389 Rock Beach Road, Rochester, NY 14617. L, SP.

Thomas Scientific, P. O. Box 99, Swedesboro, NJ 08085-6099. Tel. 609-467-2000. K, S.

Thorne, Ken, P. O. Box 684, Lambeth, Ontario N6P 1R2, CANADA. Tel. 519-652-6696. Worldwide papered and pinned specimens; CITES documentation.

Top Drawer Entomological Storage Equipment Co., 6524 Stoneman Dr., North Highlands, CA 95660. Tel. 916-344-1626. Cornell & California Academy drawers, glass-topped Schmitt boxes.

Transworld Butterfly Co., Apartado 6951, 1000L, San Jose, Costa Rica. Tel. 506-228-4768. SP, many reared.

Trécé Inc., 1143 Madison Lane, Salinas, CA 93912. Tel. 408-758-0205. PH and pheromone traps.

University Products, Inc., 517 Main Street, P. O. Box 101, Holyoke, MA 01041-0101. Tel. 800-628-1912. C, E, for storing slides and photographs.

UVP Inc., 2066 W. 11th St., Upland, CA 91786. Tel. 800-452-6788. UV-blocking eye-wear.

VWR Scientific, P. O. Box 13645, Philadelphia, PA 19101-9711. K, S.

Ward's Natural Sciences Establishment, 5100 W. Henrietta Road, P. O. Box 92912, Rochester, NY 14692, and 11850 E. Florence Avenue, Santa Fe Springs, CA 90670. Tel. 800-962-2660. "Send $15 for biology catalog." B, C, E, ED, L, P.

Wedge Entomological Research Foundation, 85253 Ridgetop Drive, Eugene, OR 97405-9535. Publishes and distributes The Moths of America North of Mexico *series.*

Weldon & Wesley, Ltd., Lytton Lodge, Codicote, Hitchin, Herts SG4 8TE, England. Tel. (01438) 820370. Books, current or old, book searches.

Worcester Glassine Envelope Co., 7720 Wisconsin Ave., Bethesda, MD 20814. Tel. 800-426-5723. Credit card phone orders only, $40 minimum, lots of 1000; 3000 of smallest size exceeds minimum; 3-4 week lag time.

Worldwide Butterflies, Over Compton, Sherborne, Dorset, DT9 4QN, England. Tel. (44) 1935 74608. SP.

Young Entomologists' Society, Inc., 1915 Peggy Place, Lansing, MI 48910-2553. Tel. 517-887-0499. B, E, ED including school programs and resource rentals, L, P.

Appendix M

Project Suggestions

(Contributors include Susan S. Borkin, Michael M. Collins, Julian P. Donahue, Ronald W. Hodges, John Holoyda, and Floyd W. Preston.)

A number of project suggestions, provided by various reviewers as well as the author, are included here. Some are narrow in scope with regard to time or subject. Others may involve repeated observation over an extended period, over a broad area, or both.

Some of these projects are individual endeavors, while others may require the cooperation of several people working in similar fashion toward the same goals. Physical constraints and the availability of time can help you decide whether to "go it alone" or seek collaboration. But whatever your choice, try to include your kids!

Don't be dismayed if you are well along into a project and then learn, as you read more, that others have already recorded what you are observing. This is not time lost. You have been sharpening your skills and increasing the depth of your knowledge, and the process proves to be more valuable than the finished product. Hopefully, you will also have enjoyed what you were doing.

Some of the projects are of relatively short duration. Some could be addressed by a single observer over a period of 5–10 years; some would better be undertaken by coordinated groups. Knowledgeable members of the Lepidopterists' Society can be consulted and should take a leading role in organizing protocols for these group efforts. It is important that methods be consistent and data comparable among different observers and areas.

Projects benefitting from an organized approach are marked with a pyramid: **Δ.**

The following suggestions are not arranged in order of increasing complexity or potential scientific value. All have the potential for producing useful scientific information while you engage in a fascinating avocation.

1. Field Observations, Biology

- Study shelters constructed by larvae, recording the process of construction; the details of finished structures; how the larva uses the structure; speculate on benefits derived. Take note of "housekeeping" details, and when the structure is abandoned, whether it is replaced or not.
- Observe and record all aspects of lepidopteral natural history, especially habitats and larval foodplants. Do preference trials with various foodplants regarding oviposition. Evaluate comparative growth of larvae on various accepted foodplants. Note those rejected.
- Observe ovipositing females to determine placement of eggs in the field, and in captivity. Record associated behavior. Use this information to determine the feasibility of searching actively for eggs already laid in the field. If you find eggs, try to rear them to determine whether you found the species you were looking for, or something different.
- Make observations on fine points of flower choices by butterflies—by species, and in terms of what else is simultaneously available in the area. Note color preferences, and whether they can be determined to be based on color and not on other parameters, such as UV reflectance, or preferred nectar.
- Record the feeding and nonfeeding behaviors of particular species of larvae. Do they select certain parts of the plant, or certain leaflets, and avoid others? Are they edge feeders, surface feeders, borers? Do they retire between meals? Where to? Do they feed only in daytime,

night time, any time? Have they any specific reaction to sunshine? Does air temperature play a role? What differences occur in later instars?

- Mark and release reared individuals from a holding cage in a butterfly garden to verify establishment of residence. Record the marking methods used (see Newman ref. in Chapter 5, or in Appendix J under Gardening).
- Study field mating behavior within and across genera and families, so that this information can be available for taxonomic decisions.
- Similarly study nectaring behavior.
- Record field observations on predation by all classes of predators.
- Make a habit documenting and preserving parasitoids.
- Make a detailed study of sesiid pheromone trap placement and reactions of incoming males, time of peak responsiveness, and any interactions or behavioral differences between different species attracted to the same pheromone trap.
- See Appendix D on *Papaipema.* Search out life histories of this genus. There is still much to be learned, particularly about western species.
- Make more complete observations, for a particular horticultural "hardiness zone," about which Lepidoptera species are truly resident, as opposed to being species that "breed north" with regularity each year. Observe whether the stages going into winter remain viable. Δ
- Study the effects of visible decoys on perching or patrolling males: color patches, spread specimens, color photos in various enlargements. Does motion alter the effectiveness of the decoy? Is UV reflectance from the decoy an asset?
- Use tethered reared female saturniids to confirm the presence of the species in an area where its continued existence is in doubt. Negative results should be recorded, but are not absolute proof of absence; siting or timing may adversely affect your success.
- Try hybridization experiments (as with saturniids), and look for parallels in the wild.
- Study wild behavior, and see whether it can be manipulated experimentally.
- If you capture an aberrant, try to get ova to rear through several generations (inbred) to see if the aberration will repeat or become accentuated. Attempt to cage-mate any reared aberrant, for the same purpose. Retaining specimen quality of the aberrant is less important than discovering its genetic potential.
- Be alert to differences in behavior or life cycle in different populations of what are assumed (not necessarily correctly) to be the same species.
- Participate in a program for tagging migrating monarchs. Δ
- Make lists of incomplete or absent life histories for rearers to work on. Emphasize those which would be especially useful for taxonomic or ecological studies.

2. *Distribution, Flight Periods*

- Compile better information to correlate the first spring or summer flight of a species with the current local plant-growth stage, to prepare phenological calendars for species in your area (Heitzman 1963). Nectar sources for first emergences, and larval foodplant growth status are useful indicators.
- Keep record charts, by species, for a given area, to document the pattern of flight over a period of years.
- Make detailed micromaps of distribution and temporal occurrence of butterflies and moths; identify ecological niches and habitat preferences, as a means of defining conservation areas.
- Make regional lists of butterflies, following the recommendations of Clench (1979) or of McMahon (1991). The title of the latter article refers to urban areas, but the text extends to associated terrain types in parks, reservations, etc. Δ
- Make baseline faunal surveys of significant habitats that are, or might be, proposed for

development (destruction) or protection; continue monitoring areas already under protection. Δ

- Long-term population monitoring is especially needed for species of concern, where management or lack thereof may be a factor in the sustained survival of populations. Local professionals can define species at risk, so that methods referred to earlier in this section can be applied. Δ
- Replicate the project of the Ohio Lepidopterists to discern and document fauna over a defined area. Δ
- Perform distribution studies to learn whether "rare" edge species of state concern are occasional overflow or wanderers from a population thriving on the other side of a political boundary (and hence not endangered), or a species actually rare and potentially endangered because of narrow habitat requirements.

3. Interaction with the Public

- Assist in preparing synoptic reference collections for study or exhibit at local parks, preserves, etc.
- Serve as a guest speaker or as an aide in schools: teachers are eager for such help.
- Establish a relationship with a researcher and contribute expertise, time, money, etc.

4. Moonlighting

- Study the effects of moonlight on moth activity: can objective observations reinforce, refute, or even explain the "conventional wisdom" by which we govern our mothing activities?
- How much moonlight inhibits visitation to artificial light—how far into the lunar cycle? Keep in mind that the waning moon is shining in the later part of the night, when fewer observations are usually being made.
- If moths are not coming to your lights, why not? What are they doing instead?
- Compare the effects of moonlight on moths, regarding light trap visitation, feeding at bait and bait traps, and nectaring at flowers.
- Does moonlight affect female moth calling, or male response? For each study session record observations on weather, temperature, atmospheric pressure gradients, latitude where study is done, etc., so that the data will be available for retrospective evaluation.

5. Study of Man-made Devices

- Evaluate commercial butterfly-feeders as adjuncts to butterfly gardening: what types of feeders attract, and what species of butterflies respond? In addition to butterflies (or moths), what insects, birds, or mammals visit them? How well do they compete with natural nectar sources? Is placement significant in relation to nearby accepted nectar sources? Test paired feeders, one "clean," and one baited. Experiment with different baits. Try comparing with a "neutral" feeding station, such as a drab flowerpot saucer. Is attraction visual, olfactory (chemical) or a combination? If UV reflectance can be added to the feeder (perhaps with plastic film sun-shields), does it have an effect?
- Evaluate artificial "mud puddles" in a similar fashion. Try paired puddles, keeping one entirely free of additives (such as urine, bouillon, etc.).
- Study "hibernation boxes" (this requires that you live in an area where one or more species naturally hibernate). Clarify the particulars of man-made hibernation housing. Is there a design utilized by butterflies? Are compartments or crevices preferred? Is cedar, cypress or other aromatic wood repellant? Does proximity to an aging woodpile or outbuilding make a difference? Is roosting gregarious or single? Will more than one individual of a species use

a single compartment? Will more than one species use the same structure? What is the best orientation and exposure on tree trunk, building, or pole, regarding sun or compass, wind, and height above ground? Should the hibernation box be positioned low enough so that it can become snow-covered? How early in the autumn do they start to be used? Up to what nighttime temperatures do they continue to be used in spring?

REFERENCES

Clench HK 1979. How to make regional lists of butterflies: Some thoughts. J Lepid Soc 33:216–232

Heitzman R 1963. Record charts for the collector of Lepidoptera. J Lepid Soc 17:44–46.

McMahon J 1991. Studying butterfly populations in urban areas. Michigan Entomol Soc, Entomol Note No. 23.

Appendix N

Careers Involving Lepidoptera

(The material in this appendix is largely the work of Julian P. Donahue)

Even if a "career" involving Lepidoptera is not your goal, the information below may give you thoughts about things you can do to share your knowledge and enthusiasm with the public, either as a volunteer or for a bit of compensation to help support your hobby.

Full-time jobs that pay real money to work with Lepidoptera are scarce and highly coveted, with a very slow turnover rate (after all, who wants to give up a job that pays you to do your hobby?).

In the quest for the perfect job, keep an open mind. Remember that in the Grand Scheme of Things the major function of butterflies and moths is NOT to serve as objects of beauty, wonder, and desire for humans; their major function is to be bird food (not to mention food for fish and multitudinous invertebrates, and to serve as pollinators and nutrient recyclers). You would still be a lepidopterist if you were identifying caterpillars from the stomachs of birds as part of an avian ecology study, or sleuthing the diets of grizzly bears by searching in their droppings for the undigested clues to the moths on which they had gorged themselves.

Regardless of whether you think there might be a job involving Lepidoptera in your future, broaden your horizon, interest and enjoyment by grasping any opportunity to learn more about botany and ecology as well. The value of these adjuncts can hardly be overemphasized.

1. Research

Research positions, mostly requiring a Ph.D. degree, include:

- Systematics and taxonomy (mostly in public and university museums, and state and federal government laboratories, often requiring the identification of insect pests).
- Research, usually combined with teaching and/or part-time curating, in colleges and universities, where Lepidoptera may be the subject of investigations into such diverse topics as physiology, biochemistry, foodplant preferences, evolution, genetics, behavior, etc.

  In a university, one could be a professor of physiology who uses moths as research tools; or one could be a professor of entomology who does systematic research, manages the insect teaching and research collection, and teaches courses in entomology.

2. Teaching

Teaching, from the earliest preschool on up, is greatly enlivened by knowledge of Lepidoptera, whether you are a classroom teacher or a science teacher.

College and university teaching usually goes hand-in-hand with research, as already noted.

3. Conservation

Conservation positions involving Lepidoptera in whole or in part are becoming increasingly common. Human overpopulation has resulted in habitat fragmentation and destruction, necessitating the creation and protection of urban, suburban, and wilderness parks and parcels to protect species and habitats threatened with destruction. Although plants, birds, and mammals are still the most frequently cited reasons for protecting a habitat, threatened and endangered Lepidoptera (especially butterflies) are gaining increasing prominence, for several

reasons (and in some cases, as in that of the El Segundo blue, a butterfly is the prime reason to protect a habitat).

The increased appreciation of Lepidoptera in the environment is the result of a number of simultaneous developments:

- The booming popularity of butterfly gardening since the mid-1970s;
- The large number of species placed on the threatened or endangered list;
- The availability of field guides to facilitate identification;
- The emergence of butterfly watching as a hobby, with several books and even an association devoted to this activity;
- And the wider availability of excellent butterfly and moth photographs, not only because of technical improvements in photographic equipment, but because more and more people have discovered that photographing a butterfly is a far more challenging task than simply catching it.

Full-time positions in the conservation field include preserve management and restoration, state and federal endangered species programs, and state nongame species and natural heritage programs. Some universities have teaching programs in conservation biology.

There are occasional research opportunities with conservation organizations.

4. Economic Entomology

Economic entomology is the practical or applied side of entomology that deals with problems caused by Lepidoptera (as opposed to pure or noneconomic entomology, in which the research has no known immediate practical benefit). There are numerous pest species of Lepidoptera, such as fall webworms, bagworms, gypsy moths, armyworms, cutworms, corn earworms, etc. Jobs in this field include consulting entomologist, extension entomologist (most land-grant universities have them), biochemist and toxicologist (in universities and in private industry), as well as integrated pest management specialists.

5. Entrepreneurism

Entrepreneurism and self-employment offer a wide range of independent job possibilities, some yet to be imagined.

- *The rising popularity of insect zoos has increased the demand for consistent and reliable sources of livestock from butterfly farms for display (usually shipped as live pupae for release in a butterfly house).*
- *Contract collectors collect, broker, or import specimens for research, display, biological supply houses, and decorative purposes, despite the need to comply with increasingly complex regulations.*
- *A number of independent butterfly videos have been made, and more are surely in production.*
- *Superb photographs from free-lance photographers appear in a wide variety of publications; sales are limited only by the marketing ability and imagination of the photographer.*
- *The market for computer-based products is still in its infancy. Entrepreneurs will develop and market a variety of Lepidoptera-based items, from checklist, identification, and inventory programs to mapping programs employing the global positioning system, high-resolution graphics—programs and multimedia applications the nature of which are beyond our present imagination.*

6. Outdoor Education and Ecotourism

Outdoor Education may offer full-time or part-time employment opportunities. Positions are often found in the larger school districts, many colleges and universities, and at the state level (check the department of fish and wildlife, conservation, or other name used in your state). Knowledge of Lepidoptera alone may not be sufficient in an education job, where a strong knowledge of plants, birds, mammals, and ecology will also be important. It is surprising, however, how ignorant of things entomological many outdoor educators are; a good general knowledge of entomology never fails to impress teachers and, especially, kids.

Ecotourism provides a few individuals part-time income, or at least free travel and expenses, while teaching special courses in butterfly biology and identification in the laboratory and/or the field in connection with nature centers and the like. Others escort field trips all over the world, with observing (such as field trips to view overwintering monarch butterflies in California or Mexico) or collecting Lepidoptera the primary purpose of the trip.

Appendix O

New Moon Tables

This material was condensed from a far more detailed table (Brahde 1977) that gives the date, hour and minute for each new and full moon from 601 B.C. to 2700 A.D. Such detail is useful for calculating the moment of high tide or of past or future lunar events, but the moth collector needs only the date—the best collecting seems to occur in the two weeks "straddling" the date of the new moon.

Table 1 lists new moon dates through 2067. Table 2 lists retrospective dates. The lunar month is 29 days, 12 hours, 44 minutes.

Table 1. New Moon Dates, Prospective

To use this table select the row for the year and the column for the month. Where these intersect is the new moon date. About 35% of years have a month with two new moons, hence such entries as "1+31" for July 2000. About every nineteen years February has no new moon, as in 2014 (and 1995).

	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	
1999	17	16	17	16	15	13	13	11	9	9	8	7	1999
2000	6	5	6	4	4	2	1+31	29	27	27	25	25	2000
01	24	23	25	23	23	21	20	19	17	16	15	14	01
02	13	12	14	12	12	10	10	8	7	6	4	4	02
03	2	1	3	1	1+31	29	29	27	26	25	23	23	03
04	21	20	20	19	19	17	17	16	16	14	12	12	04
05	10	8	10	8	8	6	6	5	3	3	2	1+31	05
06	29	28	29	27	27	25	25	23	22	22	20	20	06
07	19	17	19	17	16	15	14	12	11	11	9	9	07
08	8	7	7	6	5	3	3	1+30	29	28	27	27	08
09	26	25	26	25	24	22	22	20	18	18	16	18	09
2010	15	14	15	14	14	12	11	10	8	7	6	5	2010
11	4	3	4	3	3	1	1+30	29	27	26	25	24	11
12	23	21	22	21	20	19	19	17	16	15	13	13	12
13	11	10	11	10	10	8	8	6	5	5	3	3	13
14	1+30	—	1+30	29	28	27	26	25	24	23	22	22	14
15	20	18	20	18	18	16	16	14	13	13	11	11	15
16	10	8	9	7	6	5	4	2	1	1+30	29	29	16
17	28	26	28	26	25	24	23	21	20	19	18	18	17
18	17	15	17	16	15	13	13	11	9	9	7	7	18
19	6	4	6	5	4	3	2	1+30	28	28	26	26	19
2020	24	23	24	23	22	21	20	19	17	16	15	14	2020
21	13	11	13	12	11	10	10	8	7	6	4	4	21
22	2	1	2	1+30	30	29	28	27	25	25	23	23	22
23	21	20	21	20	19	18	17	16	15	14	13	12	23
24	11	9	10	8	8	6	5	4	3	2	1	1+30	24
2025	29	28	29	27	27	25	24	23	21	21	20	20	2025
	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	

Table 1, continued.

	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	
2026	18	17	19	17	16	15	14	12	11	10	9	9	2026
27	7	6	8	6	6	4	4	2+31	30	29	28	27	27
28	26	25	26	24	24	22	22	20	18	18	16	16	28
29	14	13	15	13	13	12	11	10	8	7	6	5	29
2030	4	2	4	2	2	1+30	30	28	27	26	25	24	2030
31	23	21	23	21	21	19	19	18	16	16	14	14	31
32	12	11	11	10	9	8	7	6	4	4	3	2	32
33	1+30	—	1+30	29	28	26	26	24	23	23	22	21	33
34	20	18	20	18	18	16	15	14	12	12	11	10	34
35	9	8	9	8	7	6	5	3	2	1+31	29	29	35
36	28	27	27	26	25	24	23	21	20	19	18	17	36
37	16	15	16	15	15	13	13	11	9	9	7	6	37
38	5	4	5	4	4	3	2	1+30	28	28	26	26	38
39	24	23	24	23	23	21	20	19	18	17	16	15	39
2040	14	12	13	11	11	9	9	8	6	6	4	4	2040
41	2	1	2	1	30	29	28	28	26	25	23	23	41
42	21	20	21	20	19	17	17	15	14	14	12	12	42
43	11	9	11	9	9	7	6	5	3	3	1	1+31	43
44	30	28	29	27	27	25	24	23	21	20	19	19	44
45	18	16	18	17	16	15	14	12	11	10	8	8	45
46	7	5	7	6	6	4	4	2+31	30	29	27	27	46
47	26	24	26	25	24	23	22	21	19	19	17	16	47
48	15	14	14	13	12	11	11	9	8	7	6	5	48
49	4	2	4	2	2+31	30	29	28	27	26	25	24	49
2050	23	21	23	21	20	19	18	17	16	15	14	14	2050
51	12	11	12	11	10	8	8	6	5	4	3	3	51
52	2+31	—	1+30	29	28	26	16	24	22	22	21	21	52
53	19	18	20	18	18	16	15	14	12	11	10	10	53
54	8	7	9	8	7	6	5	3	2	1+30	29	28	54
55	27	26	28	26	26	25	24	22	21	20	18	18	55
56	16	15	16	14	14	13	12	11	9	9	7	6	56
57	5	3	5	4	3	2	1+31	30	28	28	26	26	57
58	24	22	24	22	22	21	20	19	17	17	16	15	58
59	14	12	14	12	11	10	9	8	6	6	5	5	59
2060	3	2	2	1+30	29	28	27	26	24	24	23	22	2060
61	21	20	21	20	19	17	17	15	13	13	12	11	61
62	10	9	11	9	9	7	6	5	3	2	1+30	30	62
63	29	28	30	28	28	26	25	24	22	21	20	19	63
64	18	17	18	16	16	14	14	12	11	10	8	8	64
65	6	5	7	5	5	4	3	2+31	30	29	27	27	65
66	25	24	25	24	24	22	22	21	19	19	17	17	66
2067	15	13	15	13	13	11	11	10	8	8	7	6	2067
	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	

Table 2. New Moon Dates, Retrospective.

	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	
1998	28	26	28	26	25	24	23	22	20	20	19	18	1998
97	9	7	9	7	6	5	4	3	1	1+31	30	29	97
96	20	18	19	17	17	16	15	14	12	12	11	10	96
95	1+30	—	1+31	29	29	28	27	26	24	24	22	22	95
94	11	10	12	11	10	9	8	7	5	5	3	2	94
93	22	21	23	21	21	20	19	17	16	15	13	13	93
92	4	3	4	3	2	1+30	29	28	26	25	24	24	92
91	15	14	16	14	14	12	11	10	8	7	6	6	91
1990	26	25	26	25	24	22	22	20	19	18	17	17	1990
89	7	6	7	6	5	3	3	1+31	29	29	28	28	89
88	19	17	18	16	15	14	13	12	11	10	9	9	88
87	29	28	29	28	27	26	25	24	23	22	21	20	87
86	10	9	10	9	8	7	7	5	4	3	2	1+31	86
85	21	19	21	20	19	18	17	16	14	14	12	12	85
84	3	1	2	1	1+30	29	28	26	25	24	22	22	84
83	14	13	14	13	12	11	10	8	7	6	4	4	83
82	25	23	25	23	23	21	20	19	17	17	15	15	82
81	6	4	6	4	4	2	1+31	29	28	27	26	26	81
1980	17	16	16	15	14	12	12	10	9	9	7	7	1980
79	28	26	28	26	26	24	24	22	21	21	19	19	79
78	9	7	9	7	7	5	5	4	2	2+31	30	29	78
77	19	18	19	18	18	16	16	14	13	12	11	10	77
76	1+31	29	30	29	29	27	27	25	23	23	21	21	76
75	12	11	12	11	11	9	9	7	5	5	3	3	75
74	23	22	23	22	21	20	19	17	16	15	14	13	74
73	4	3	5	3	2	1+30	29	28	26	26	24	24	73
72	16	15	15	13	13	11	10	9	7	7	6	5	72
71	26	25	26	25	24	22	22	20	19	19	18	17	71
1970	7	6	7	6	5	4	3	2+31	30	30	28	28	1970
69	18	16	18	16	16	14	14	13	11	11	9	9	67
68	29	28	28	27	27	25	25	23	22	21	20	19	68
67	10	9	11	9	9	8	7	6	4	3	2	1+31	67
66	21	20	22	20	20	18	18	16	14	14	12	12	66
65	2	1	3	2	1+30	29	28	26	25	24	23	22	65
64	14	13	14	12	11	10	9	7	6	5	4	4	64
1963	25	24	25	23	23	21	20	19	17	17	16	16	1963
	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	

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Glossary

aedeagus	The male intromittent organ; the penis.
ambient	Surrounding.
androconial	Refers to groups of scales in the male, which bear chemicals vital to courtship.
appressed	Pressed close to, or lying flat against something.
bursa	A sack or saclike bodily cavity.
cline	Gradual change in a character or feature across the distributional range of a species or population, usually correlated with an environmental or a geographic transition.
curating	Arranging and organizing (vernacular, but used routinely in entomology).
detritus	Accumulated material, debris.
diapause	Period of arrested development.
eclose	To emerge, usually as an adult from a pupa.
exotic	From another part of the world; foreign.
exuviae	The cast skin of an arthropod.
fecula	Excrement or droppings.
Fibonacci series	A series of numbers in which each member is the sum of the two preceding numbers. For example, a series beginning 0, 1... continues as 1, 2, 3, 5, 8, 13, 21, and so forth. Natural patterns, such as the spiral growth of leaves on trees, seed arrangement on composite flower heads, or the shell of the chambered nautilus exemplify the Fibonacci series.
foodplant	A plant that a larva habitually feeds on.
frass	Plant fragments made by a boring insect, usually mixed with excrement.
gynandromorph	Abnormal individual bearing structural characteristics of both sexes.
hatch	To emerge, usually as a larva from an egg.
homologous	Similar in structure and evolutionary origin, but not necessarily in function, as the flippers of a seal and the human hand.
host plant	Plant on which an insect lives.
insolation	Solar radiation striking the earth.
instar	An insect between successive molts, the first instar being the insect between hatching and the first molt.
juxta	Sclerotized plate, often shield-shaped, ventrad of aedeagus, which it helps to support.
larva	Caterpillar. Stage of feeding and growth between egg and pupa; plural, larvae; "ex larva," from a larva.
osmeterium	Fleshy, tubular, eversible, usually Y-shaped scent gland at the anterior end of certain caterpillars (esp. swallowtails).
ovum	Egg; plural, ova; "ex ovo," from an egg.
parasitoid	Animal that feeds on or in another living animal for a relatively long time, consuming all or most of its tissues, and eventually killing it (a *parasite* usually does not kill its host).
photoperiod	Ratio of light to darkness in the daily cycle.
polyphenism	(Seasonal). Forms of a species differing in appearance depending on the season.
porrect	Extending forward horizontally.
prepared specimen	Here, a specimen readied and labeled for permanent storage.
pupa	Nonfeeding stage between larva and adult; plural, pupae; "ex pupa," from a pupa.
sexing	Ascertaining sex of specimens.
stadium	Period between molts in a developing insect.
stage	Level, degree, or period of time in the course of a process, especially a step in development, as larval stage.
systematics	Taxonomy.
systematist	Taxonomist.
taxon	Taxonomic category or group, such as phylum, class, order, family, genus, or species; plural, taxa.

taxonomy	Classification of objects in an ordered system that indicates natural derivations and relationships.
tegula	Small scaled structure overlying the base of the front wing.
tegumen	Roof- or hoodlike structure of male genitalia giving rise to uncus and various other processes, and with which valvae are dorsally articulated.
tufts	Short clusters of elongated scales.
uncus	Mid-dorsal structure arising from posterior margin of tegumen, usually strongly sclerotized, sometimes bearing projections, spines or bristles; often an important male genital character.
valva	One of the paired, presumably clasping organs of male genitalia; plural, valvae.
vinculum	Flat, U-shaped ring with which valvae are basally articulated.
vestiture	Scales covering surface of body or wings.

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