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August 13, 2007

New Pest Response Guidelines
False Codling Moth
Thaumatotibia leucotreta

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New Pest Response Guidelines: False Codling Moth Thaumatotibia leucotreta was prepared by Jeffrey Stibick, USDA–APHIS–PPQ–Emergency and Domestic Programs and edited by Patricia S. Michalak, USDA–APHIS–PPQ–Manuals Unit.

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R. L. Dunkle *May 3, 2007*

Richard Dunkle, Deputy Administrator **Date**
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False Codling Moth

Credits

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Industry, State Regulatory Officers, Universities, and Governmental Agencies as credited in: September 1983. *Action Plan: False Codling Moth Cryptophlebia leucotreta (Meyrick)*. USDA–APHIS–PPQ–Emergency Programs.

Graphics

Cover

Adult moth—Georg Goergen/IITA Insect Museum, Cotonou, Benin

Citrus, Corn, Cotton, Macadamia, Peach—USDA–Agricultural Research Service

False Codling Moth

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False Codling Moth

Chapter 1

Introduction

Contents

Purpose

Use *New Pest Response Guidelines: False Codling Moth* as a guide when designing a program to detect, monitor, control, contain, or eradicate an infestation of false codling moth (*Thaumatotibia* [= *Cryptophlebia*] *leucotreta* [Meyrick]).

United States Department of Agriculture (USDA)–Animal and Plant Health Inspection Service (APHIS)–Plant Protection and Quarantine (PPQ) developed this guide through discussion, consultation, or agreement with staff at USDA–Agricultural Research Service (ARS), universities, industries, and state departments of agriculture.

If the pest is detected in the United States, USDA–APHIS–PPQ personnel will produce a site-specific action plan based on this report. Personnel in state agriculture departments and others concerned with developing local survey or control programs for this pest will find the *Guidelines* useful.

The *Guidelines* will be updated as new information becomes available. Specific emergency programs should be based on information available at the time of the incident.

Pest Status

False codling moth is a significant import threat to the United States for the following reasons:

- False codling moth is a pest of economic importance to many crops in its native habitat including avocado, citrus, corn, cotton, macadamia, peach and plum.
- False codling moth could easily become established in the southern and southwestern United States.
- Establishment of false codling moth in the United States could result in economic losses of billions of dollars.
- False codling moth has been frequently intercepted at United States ports of entry in both cargo and passenger luggage.
- False codling moth larvae are difficult to detect once they enter the fruit.
- False codling moth larvae eggs are laid singly and are difficult to detect.

This pest has not been found within the United States.

Detection of a single larva in fruit destined for export can result in rejection of an entire consignment (Ministry of Agriculture and Forestry 2004).

Disclaimers and Document Comprehension

This document is not intended to be complete and exhaustive. It provides a foundation, based upon the literature available, to assist further work. Some key articles were not available at the time of writing, and not all specialists and members of the research community were consulted for their advice. Conduct your own literature search. Search the World Wide Web frequently, since material is updated periodically.

Commercial Suppliers

References to commercial suppliers or products should not be construed as an endorsement of the company or product by USDA.

Contacts

Success of an emergency program depends on the cooperation, assistance, and understanding of other involved groups. The appropriate liaison and information officers should distribute news of program progress and developments to interested groups, including:

- Other federal, state, county, and municipal agricultural officials
- Grower groups (such as specific commodity or industry groups)
- Commercial interests
- Academic entities with agricultural interests
- Land-grant universities with Cooperative Extension Services
- State and local law enforcement officials
- Public health agencies
- Foreign agricultural interests
- National, state and local news media, and
- Consumers

Initiating an Emergency Pest Response Program

An emergency pest response program or incident response consists of detection and delimitation, and may be followed by programs in regulation, containment, eradication and/or control.

If a newly detected exotic or imminent pest threat lacks a current *New Pest Response Guidelines* for reference, the New Pest Advisory Group (NPAG) evaluates the pest. After assessing the risk to U.S. plant health and consulting with experts and regulatory personnel, NPAG makes a recommendation to PPQ management for a course of action.

Follow this sequence in any order when initiating an emergency pest response program:

1. New or reintroduced pest is discovered and reported.
2. Pest is examined and pre-identified by a regional or area identifier (**See Identification on page 3-1**).
3. Pest identity is confirmed by national taxonomic authority (**See Identification on page 3-1**).
4. *New Pest Response Guidelines* are consulted or NPAG is assembled to evaluate the pest.
5. Depending on the urgency, official notifications are made to the National Plant Board, cooperators, or trading partners.

6. Delimiting survey is conducted at site of detection (**See Survey on page 4-1**).
7. Incident Assessment Team can be sent to evaluate the site.
8. Recommendation is made, based on the assessment of surveys, other data, and recommendations of the Incident Assessment Team or an NPAG, as follows (**See Control on page 6-1**):
   - ❖ Take no action.
   - ❖ Regulate the pest.
   - ❖ Contain the pest.
   - ❖ Suppress the pest.
   - ❖ Eradicate the pest.
9. State departments of agriculture are consulted.
10. If appropriate, a control strategy is selected.
11. PPQ Deputy Administrator authorizes a response.
12. Command post is selected and the Incident Command System is implemented.
13. Further detection surveys are conducted (**See Survey on page 4-1**).
14. Field identification procedures are standardized (**See Identification on page 3-1**).
15. Data reporting is standardized.
16. Environmental assessments are completed as necessary.
17. Treatment is applied for required pest generational time (**See Control on page 6-1**).
18. Environmental monitoring is conducted if appropriate.
19. Pest monitoring surveys are conducted to evaluate program success (**See Survey on page 4-1** and **Control on page 6-1**).
20. Programs are designed for eradication, containment or long-term control of the pest (**See Control on page 6-1**).

Program Safety

Safety of the public and program personnel has priority in preprogram planning and training, and throughout operations. Safety officers and supervisors must enforce on-the-job safety procedures.

Support For Program Decision Making

USDA–APHIS–PPQ–Center for Plant Health, Science and Technology (CPHST) provides technical support to emergency pest response program directors concerning risk assessments, survey methods, control strategies, regulatory treatments, and other aspects of pest response programs.

2

False Codling Moth

Chapter 2

Pest Information

Contents

Introduction

Use the *Pest Information* chapter learn more about the classification, history, host range, and biology, of the false codling moth.

Classification

Use **Table 2-1** as an aid to the identification of false codling moth.

TABLE 2-1 Classification of False Codling Moth

Phylum	Arthropoda
Class	Insecta
Order	Lepidoptera
Family	Tortricidae
Tribe	Grapholitini
Species	*Thaumatotibia leucotreta*
Synonym	*Cryptophlebia leucotreta*
Common name	False codling moth
Easily confused with the following species	*Cryptophlebia peltastica* (Litchi moth), *Cydia pomonella* (Linnaeus) (Codling moth in U.S.A.), *Cydia toreuta, Ecdytolopha punctidiscana* (Todd Gilligan, pers. comm.), *Mussidia nigrevenella* (in West Africa) (Moyal & Tran 1989; Silvie 1990; CPC 2004), *Thaumatotibia batrachopa* (Macadamia nut borer in Africa)

History

The false codling moth is native to Africa (Newton 1998). The pest has a wide range of wild and cultivated host plants. It has infested cotton in most equatorial areas; citrus in Southern Africa; and recently macadamia nuts in Malawi (**Figure 2-1 on page 2-2**) (Newton 1998).

False codling moth has occasionally been found in Europe, where it was imported with produce from Africa (Bradley et al. 1979; Karvonen 1983). Border inspections have intercepted false codling moth in Denmark, Finland, Netherlands and United Kingdom; the countries have remained free of the pest.

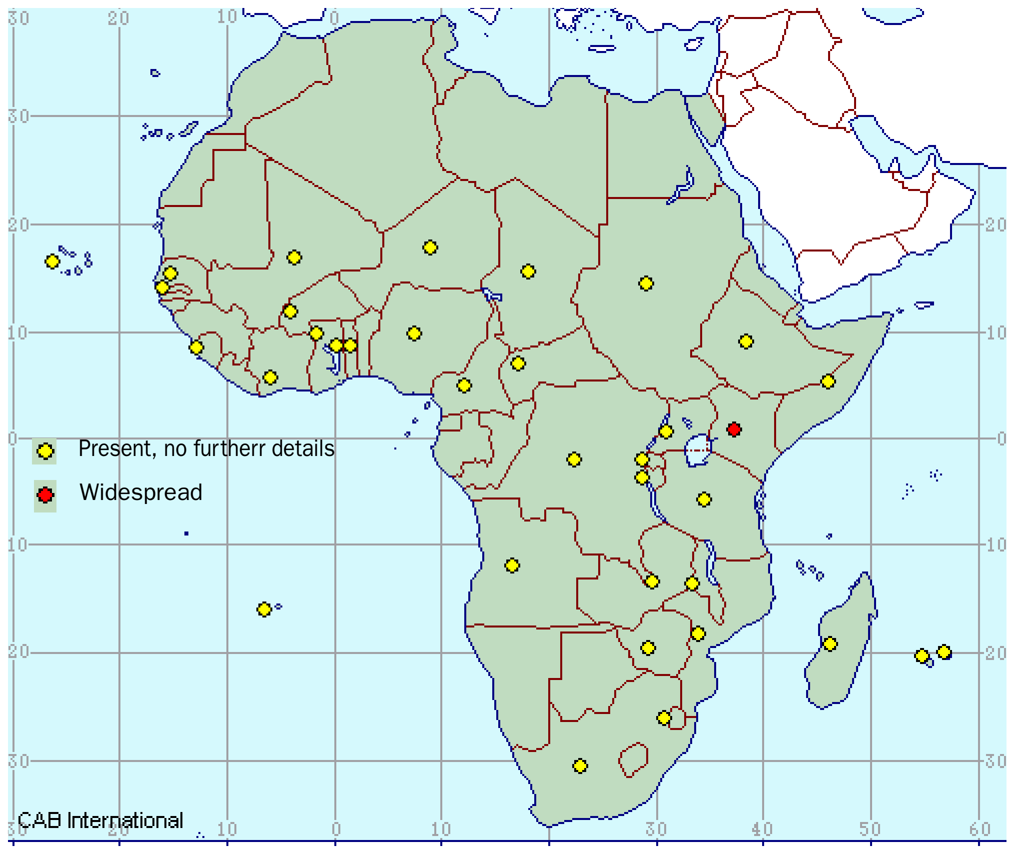


FIGURE 2-1 Geographic Distribution of False Codling Moth. Bullets indicate the presence of this pest. False codling moth is established in the following African countries: Angola, Benin, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Congo Democratic Republic, Côte d'Ivoire, Eritrea, Ethiopia, Gambia, Ghana, Kenya, Madagascar, Malawi, Mali, Mauritius, Mozambique, Niger, Nigeria, Rwanda, Réunion, Saint Helena, Senegal, Sierra Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Uganda, Zambia, and Zimbabwe [Image courtesy of CPC (2006)].

Documentation by PPQ

PPQ first recognized the importance of false codling moth as a potential threat in 1960 (USDA 1960).

In 1982, PPQ–Emergency Programs assigned their highest priority to this pest. At the same time, Area Identifiers Odermatt, Bauman, Blackburn, Ford, and Ead (1982) reported that false codling moth was likely to be detected.

PPQ reviewed the status of this pest in an *Action Plan* (USDA 1983), and quickly revised it (USDA 1984). At the same time, Whittle (1984) revised the earlier report (USDA 1960).

Since 1984, reports of false codling moth in the United States were limited to documents produced by Cooperative Agricultural Pest Survey (CAPS).

Ecological Range

False codling moth has established populations in geographic areas with climates equivalent to USDA climatic zones 7b through 10a (**Figure 2-2**) (Venette et al. 2003).

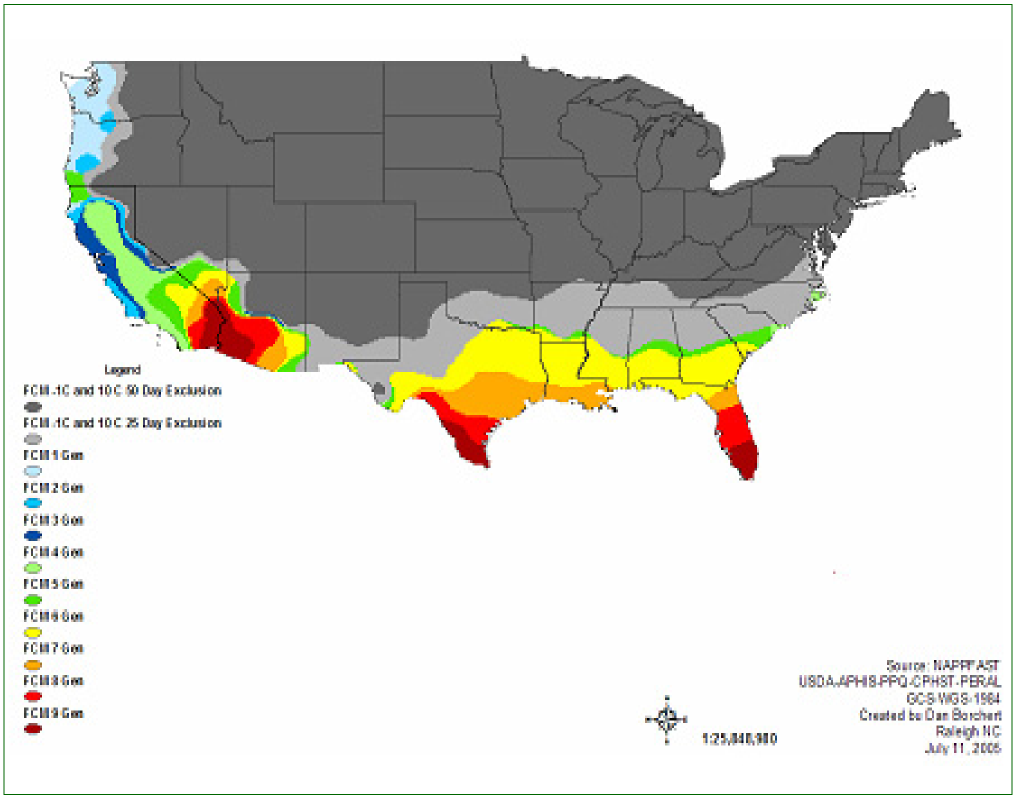


FIGURE 2-2 Estimated Potential Number of Generations of False Codling Moth. Base developmental temperature is 12°C (53.6°F). Upper developmental temperature is 40°C (104°F). Estimation of generation time is 450 degree days. Two potential exclusion layers exist of 25 and 50 or more days where minimum daily temperature is below -1°C (30.2°F) and average daily temperature is below 10°C (50°F) [Image courtesy of Borchert (2005].

Damage to Host Plants

False codling moth is a pest of economic importance to many crops throughout sub-Saharan Africa, South Africa and the islands of the Atlantic and Indian Oceans. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop. Important host crops include avocado (*Persea americana*), citrus (*Citrus* spp.), corn (*Zea mays*), cotton (*Gossypium* spp.), macadamia (*Macadamia* spp.), and peach and plum (*Prunus* spp.).

Avocado

Moths lay eggs superficially on the fruit of avocado. Larvae hatch and develop, and can enter through the skin. Larvae are unable to develop in avocado fruit. However, their entrance creates lesions that lessen the marketability of fruit (Figure 2-3). Lesions develop into a raised crater on the fruit surface, with an inconspicuous hole in the center where the larva has entered (Du Toit et al. 1979). Granular excreta can also be seen. Of the lepidopteran pests that damage avocado fruits, false codling moth is the most important (Erichsen & Schoeman 1992).

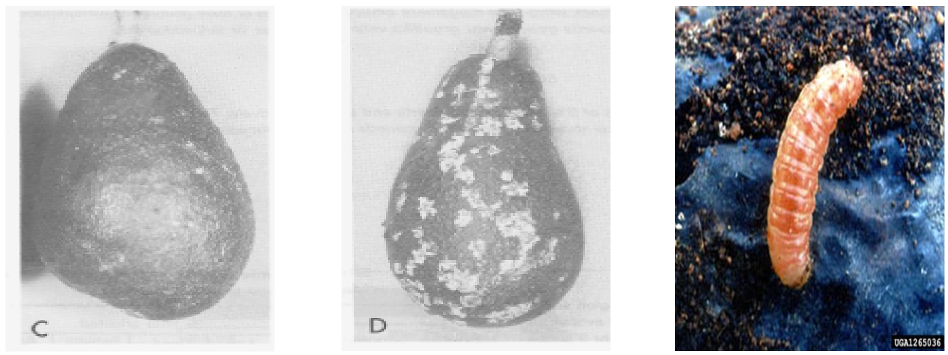


FIGURE 2-3 Left and Middle: Damage to Avocado Fruit Caused by False Codling Moth [Images courtesy of Du Toit et al. 1979]. Right: Larva of False Codling Moth in Avocado Fruit [Image courtesy of Tertia Grové, Institute for Tropical and Subtropical Crops, http://www.forestryimages.org].

Citrus

All stages of citrus fruit are vulnerable to attack (Figure 2-4 on page 2-6). False codling moth larvae are capable of developing in hard green fruit before control measures can be started. Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers.

Yield losses due to this pest have been as great as 20% (USDA 1984a).

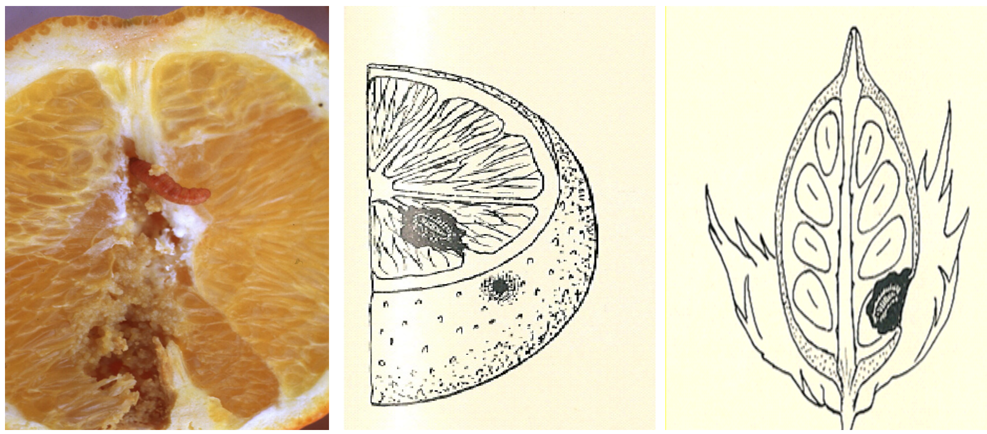


FIGURE 2-4 Larvae of False Codling Moth Within Orange Fruit [left image courtesy of Sean Moore, CRI; middle image courtesy of Hill (1983)]and Cotton Boll [right image courtesy of Hill (1983)]

Corn

Larvae damage corn by entering the ear from the husk through the silk channel.

Cotton

In Ugandan cotton, false codling moth caused 20% loss of early sown varieties and 42—90% loss of late varieties. Larval penetration of cotton bolls facilitates entry of other microorganisms that can rot and destroy the boll (Byraruhanga 1977; Couilloud 1994) (Figure 2-4). According to Erichsen & Schoeman (1992), the cultivars Edranol, Hass and Pinkerton were the most susceptible to attack by false codling moth.

Macadamia

False codling moth has caused significant yield losses of 30% or more to macadamia crops in Israel and South Africa. Larvae damage the nuts by feeding on the developing kernel after they pierce the husk and shell. Nuts reaching 14—19 mm diameter size are at the most risk because nutrient content is the greatest; concurrently, false codling moth reaches the adult stage by this point and is able to oviposit on the nuts (La Croix and Thindwa 1986; Wysoki 1986).

Stone Fruits

All stages of stone fruits are vulnerable to attack. False codling moth larvae are capable of developing in hard green fruit before control measures can be started. Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers.

Infestation by false codling moth has resulted in up to 28% loss of late peach crops. Larvae damage stone fruits as they burrow into the fruit at the stem end and begin to feed around the stone. Infestations can be identified by the brown spots and dark brown frass. Peaches become susceptible to damage about six weeks before harvest.

Detecting infested peaches can be difficult if the fruit is still firm and abscission has not occurred; consequently, the danger of selling potentially infested fruit poses a serious threat to the peach industry (Blomefield 1978; Daiber 1975; USDA 1984a).

Life Cycle

The life cycle of the false codling moth includes egg, larval, pupal and adult stages (**Figure 2-5**). Diapause is absent (CPC 2004).

The complete life cycle ranges from 30 days (under best conditions) to 174 days (under poor conditions). Two to 10 generations can succeed annually. As many as five generations per year can succeed on oranges in South Africa (Venette et al. 2003). The number of generations is influenced by several factors, including temperature, food availability and quality, photoperiod, humidity, latitude and the effect of predators and diseases. With an uninterrupted supply of plant hosts, false codling moth remains active throughout the year.

Females live longer than males. The ratio of males to females is 1:2 (Couilloud 1994; Daiber 1980).

Moisture has a considerable effect on control of false codling moth. Gunn (1921) reported that infestations were less severe where heavy rainfall was recorded.

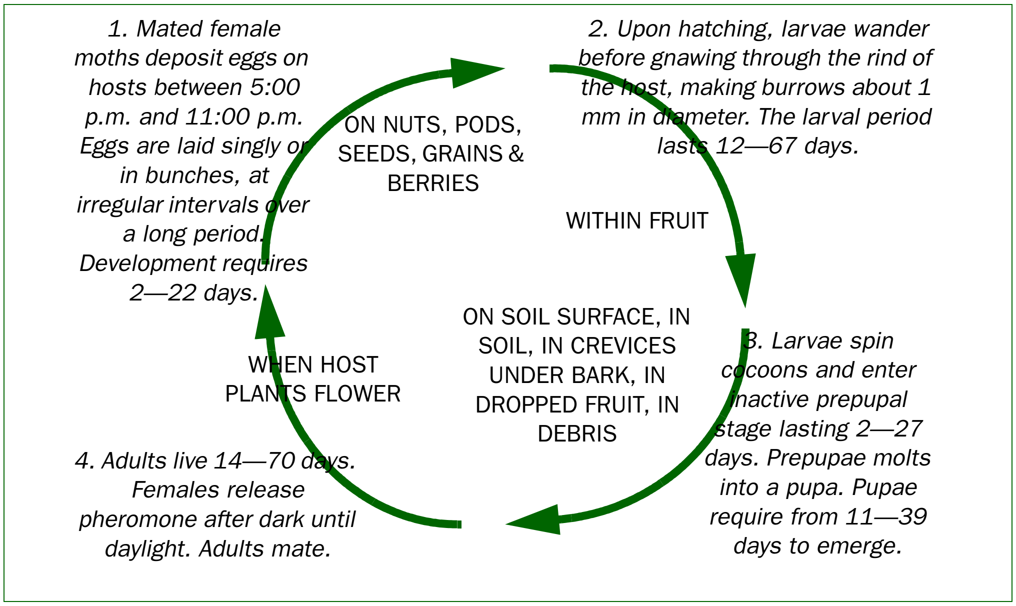


FIGURE 2-5 Life Cycle of False Codling Moth

Egg Stage

Laying

Mated female moths fly at night, depositing eggs on suitable hosts between 5:00 p.m. and 11:00 p.m. Eggs are laid singly or in bunches. Females lay eggs at random in depressions of the rind of host fruit; on smooth, non-pubescent surfaces; on fallen fruit; or on foliage. Females lay eggs at irregular intervals over a long period.

At an optimum temperature of 25°C (77°F), females can lay three to eight eggs per fruit (nuts, pods, seeds, grain heads, and berries) and up to 800 over her life span. If there are numerous females, many eggs can accumulate on the fruit. However, only a few can survive due to lack of food and cannibalism.

On peaches, eggs are almost always laid on the upper surface of peach leaves. On cotton, green bolls are preferred (Blomefield 1978; Couilloud 1994; Daiber 1975; Newton 1988a 1989a; Newton and Crause 1990; Ochou 1993).

Development

Egg development requires 2—22 days depending on temperature. Eggs are extremely sensitive to cold temperatures and extended periods of low humidity. Temperatures below freezing over a 2—3 day period can kill eggs (Blomefield 1978; Daiber 1979).

Larval Stage

Feeding

Upon hatching, larvae wander before gnawing through the rind of the host, making burrows about 1 mm in diameter. The entrance is conspicuous due to the presence of frass and discoloration of the surrounding rind.

If the host has a hard rind, such as an acorn, entrance is made at the base or attachment to the cup where softer tissue exists. When the host has a soft rind, such as citrus or peach, the larvae will burrow into the rind almost anywhere. Larvae prefer the navel end, or an injured area or cut in the rind. In some rinds, such as avocado, the entrance is marked by formation of a raised crater (Blomefield 1978; Couilloud 1994; Daiber1979, 1989; La Croix 1986; Newton and Crause 1990) (**Figure 2-3 on page 2-5**).

In cotton, the young larvae feed almost entirely inside the boll wall. Older larvae penetrate the inner septum and feed on developing seeds and lint, and can complete development on seeds in open bolls. Such feeding damages the boll, providing an entrance for pathogens. Under humid conditions, pathogens enhance decay.

Development

The larval period lasts 12—33 days in warm weather and 35—67 days in cool weather; there are five instars (**Figure 2-6 on page 2-9**). Younger larvae feed near the surface; older larvae bore toward the center. Generally, only 1—3 larvae per fruit survive.

Temperature and poor food quality can slow down the rate of larval development. By the time the larva is ready to leave the fruit, the fruit might have dropped.

Mature larvae exit the fruit, then drop to the ground on silken threads.

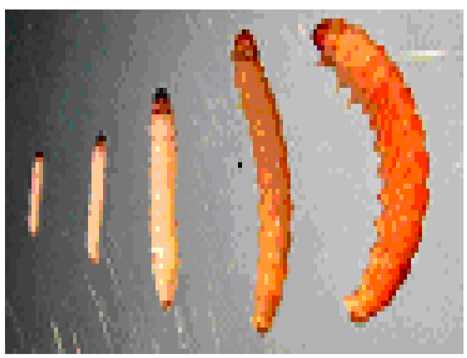


FIGURE 2-6 Larvae of False Codling Moth
[Image courtesy of S. Bloem and J. Hofmeyr]

Pupal Stage

Prepupae

Larvae spin a cocoon and enter an inactive prepupal stage lasting 2—27 days. If newly formed cocoons are covered with sand, the prepupae can leave the cocoons to form a new cocoon at the soil surface. The prepupae molts into a pupa.

Pupae

Pupae are cream colored and soft, maturing to a hardened dark brown. The male pupa requires a longer development period than the female pupa. Females require from 11—39 days to emerge, depending on temperature, and the entire cocoon stage is 13—60 days. Males can require from 13—47 days to emerge, depending on temperature.

Pupation occurs on the soil surface, in soil, in crevices under bark, in dropped fruit or in debris. Pupae emerge slightly from the cocoon before adult emergence takes place. The empty pupal skin usually remains attached to the cocoon. Pupae are sensitive to cold temperatures and to heavy rainfall. Older pupae are more resistant to cold (Daiber 1989; Myburgh and Bass 1969).

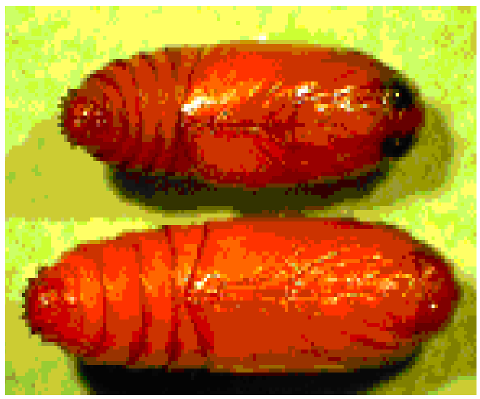


FIGURE 2-7 Pupae of False Codling Moth [Image courtesy of S. Bloem and J. Hofmeyr]

Adult Emergence

The small, inconspicuous moths fly only at night and spend days resting on shaded portions of the host. Males live 14—57 days; females survive 16—70 days. Dispersal normally is limited to several hundred meters. Numbers are generally controlled by temperature and the availability of host material (APHIS 1983).

Moth activity increases with the onset of host flowering. Females call males through pheromone release starting several hours after dark, peaking 5 hours later, and dropping off rapidly thereafter until daylight (Bestmann et al. 1988). Adults can mate several times per day (Blomefield 1978; Couilloud 1994; Newton 1989b).

FIGURE 2-8 Adult False Codling Moth [Image courtesy of Crop Protection Compendium (2004)]

Predicting Insect Development

Generation or degree day models are poor predictors of developmental time and adult emergence of false codling moth (Jim E. Carpenter, pers. comm. 2005). To produce pest models using weather, climate and soil data, use the graphical user interface available from NAPPFAST (http://www.nappfast.org/index.htm). Or, use the degree-day utility available from University of California, Integrated Pest Management Programs (http://www.ipm.ucdavis.edu/WEATHER).

See Resources on page A-1 for more sources of information on predicting insect development, and on collecting local temperature data.

3

False Codling Moth

Chapter 3

Identification

Contents

Introduction

Use *Chapter 3 Identification* as a guide to identification of the false codling moth. Accurate identification of the pest is pivotal to assessing its potential risk, developing a survey strategy, and determining the level and manner of control.

Authorities

Qualified state, county, or cooperating university personnel can screen and perform tentative identification of suspected false codling moth specimens. Before survey and control activities are initiated in the United States, a specialist recognized by PPQ—National Identification Services (NIS) must verify the first detection of the false codling moth. For information regarding personnel specializing in this pest, contact NIS at the following address:

National Identification Services
USDA–APHIS–PPQ
4700 River Road, Unit 133, 4A20
Riverdale, MD 20737
Office: 301.734.5312
Fax: 301.734.3621
Email: joseph.f.cavey@aphis.usda.gov or
paul.a.courneya@aphis.usda.gov

Pre-identification

Use this section as a guide to pre-identification of false codling moth.

Thaumatotibia spp.

Moths in the genus *Thaumatotibia* share the following characters:

- Size small to medium
- Color gray
- Forewings broad with a blackish triangular pretornal patch

Thaumatotibia spp. resemble *Cryptophlebia* spp. with the following **exceptions**:

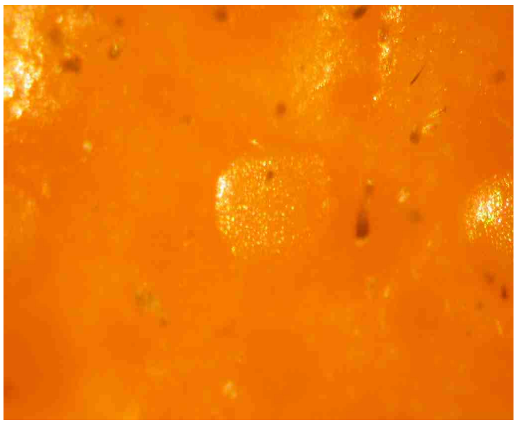
- T8 in male with a broadly sclerotized plate with convex posterior margin and laterally produced into curved points
- Long filiform scale tufts from shallow membranous pockets on each side of T8 **absent**
- Male and female genitalia are diagnostic (Komai 1999)

Thaumatotibia leucotreta

Eggs

Use the following characters to identify eggs of *Thaumatotibia leucotreta* (**Figure 3-1**):

- Length 1 mm
- Color translucent white
- Shape flat and oval
- Surface with shiny reticulate sculpture (APHIS 1983; Newton 1998)



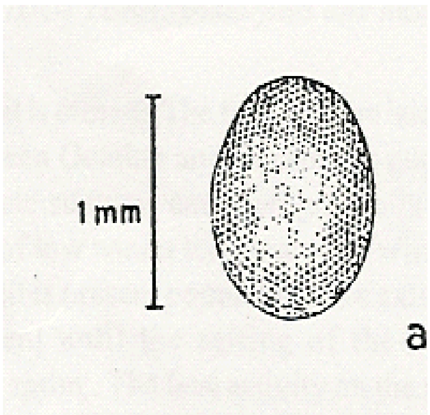


FIGURE 3-1 Eggs of False Codling Moth [Images courtesy of H. Hoffmeyr]

First and Subsequent Larval Instars

Use the following characters to identify first instar larvae of *Thaumatotibia leucotreta*:

- Length 1—1.3 mm
- Body color creamy white with minute black spots, each with a short hair
- Head color brownish black

Subsequent instars take on a characteristic pinkish-red color which is less intense on the underside.

Fifth Instar Larvae

Use the following characters to identify fifth (final) instar larvae of *Thaumatotibia leucotreta*:

- Length 12—20 mm
- Body color diffuse overall pink tending to orange yellow on the sides, top and legs
- Head color light maroon
- Pronotum color yellowish brown
- Pronotum jutting out in front, to both sides, and to the rear
- Anal comb with 2—7 teeth (APHIS 1983)

Pupae

Use the following characters to identify pupae of *Thaumatotibia leucotreta*:

- Length 7 mm
- Color yellow to dark brown
- Segments with transverse row spines
- Males smaller than females
- Males ventral side of ninth abdominal segment with two knobs side by side in the center
- Females lack knobs
- Encased in cocoon with soil and leaf fragments (APHIS 1983)

Adults

Use the following characters to identify adults of *Thaumatotibia leucotreta* (**Figure 3-2 on page 3-4**):

- Body length 6—9 mm
- Body width 2.5 mm
- Wing span 16—20 mm
- Color grayish brown to dark brown or black
- Forewings broad, elongate with black triangular patch
- Forewings fringed with hairs
- Hindwings lighter grayish brown, darker towards outer margins
- Thorax with double posterior crest
- Males with the following characters:
    - Genital tuft large, pale gray
    - Hind legs with dense brush of grayish white hairs
    - Inner side of hind tibia with tufts of modified scales
    - Hind wings with deep semicircular pocket (APHIS 1983)

FIGURE 3-2 Male Adult of False Codling Moth [Image courtesy of CPC (2004)]

Similar Species

In the United States, codling moth (*Cydia pomonella* [Linnaeus]) is easily confused with the false codling moth (Bradley et al. 1979).

In West Africa, false codling moth is often found in conjunction with the pyralid moth (*Mussidia nigrevenella* [Moyal & Tran 1989; Silvie 1990]). Other similar species in West Africa are the macadamia nut borer (*Thaumatotibia batrachopa*) and the litchi moth (*Cryptophlebia peltastica*). Distinguish the male litchi moth from similar species by a subtriangular or Y-shaped T8 with a pair of tufts of filiform scales from membranous pockets on each side (Newton 1998). **See Taxonomy on page D-1** for additional information.

Collection, Rearing and Preparation of Specimens

Labeling

Label samples immediately after collection with the following information:

- Location
- Time of sampling
- Method used to obtain the sample
- Name of sampler
- Temperature and habitat

Rearing

If possible, collect eggs, larvae and pupae with sufficient host material for rearing. Adult specimens that were raised from eggs or larvae are easier to identify, compared with specimens collected from sticky traps. Consult with identifiers for instructions on artificially rearing insects.

Important	**Facilities must meet the security standards for rearing of quarantined insects.**

Eggs

Carefully collect fruits, nuts or leaves with eggs and place them in a rearing cage with soil placed on the bottom. Maintain humidity at approximately 50%.

Preserving

Eggs and Pupae

Preserve eggs and pupae in vials of ethyl alcohol.

Larvae

Drop larvae in boiling water for several seconds, then preserve in ethyl alcohol.

Adults

Reared Adults—Do not place moths in alcohol. Pin moths to a block of foam but do not spread the wings. Spreading delays shipment and makes specimens fragile. Save the pupal and larval cast skins with the moth in a dry vial.

Captured Adults on Sticky Traps—Cut out the portion of the trap with the moth and pin it in a foam bottom box. Use enough pins to prevent the moth from shaking loose in the mail.

Shipping

Double-box and ship the specimens with at least 2 inches of padding around the sample.

Address	**Include PPQ Form 391 (See PPQ 391 Specimens For Determination on page B-1) marked "Urgent". See the Manual for Agricultural Clearance http://www.aphis.usda.gov/import_export/plants/ppq_manuals.shtml) for instructions on completing the form.**

Mail samples to the following address:

Leader, Taxonomic Services Unit
USDA–ARS–BA–PSI
Building 046, Room 101A, BARC-EAST
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False Codling Moth

Chapter 4

Survey

Contents

Introduction

Use the *Survey Procedures* chapter when conducting a pest survey for false codling moth.

The purpose of a survey is to determine the extent and means of pest dispersal. Surveys are also used to assess the effectiveness of treatments. Use detection and delimiting surveys to detect and delimit false codling moth. Use a monitoring survey to measure the effectiveness of applied treatments on the pest population. Use **Table 4-1** and the following sections to learn more about survey procedures.

TABLE 4-1 Decision Table of Surveying Methods for False Codling Moth

If you:	Then use this method:
Are unsure that the pest is present	**Detection survey**—Use traps baited with pheromone and/or visual inspection and/or soil sampling to capture specimens. Install traps; inspect plants, fallen fruit, nuts, bolls, etc.; or sample soil at suspect locations. Consult with a false codling moth specialist to confirm your identification.
Know the pest is present and need to define its geographic location	**Delimiting survey**—Use traps baited with pheromone and visual inspection at specific locations and densities to capture specimens according to the plan outlined below. Consult with a false codling moth specialist to confirm your identification.
Have applied a control procedure and need to measure its effectiveness	**Monitoring survey**—Use traps baited with pheromone and/or visual inspection and/or soil sampling to capture specimens. Install traps; inspect plants, fallen fruit, nuts, bolls, etc.; or sample soil at suspect locations. Consult with a false codling moth specialist to confirm your identification.

Before Starting

When using sticky traps to catch small moths, ensure that proper taxonomic support and funds are present. False codling moths are difficult to work with because they are so small. Managers considering a survey need to take this into account before a large project is approved. Large projects require statewide or regional surveying. In particular, the following questions should be addressed:

Who will screen the samples?—Tiny insects are easy to miss. Screeners must be qualified to recognize the target.

Who will prepare the samples?—Making slides of Lepidoptera genitalia or wings is difficult. If specimen quality is poor, timely results are impossible.

Does the specialist have time to examine samples?—For a statewide survey, this could mean many extra samples to add to the work plan.

If local staff lacks expertise, are funds available for training?—In most cases, training is the key to solving the problems above.

See Identification on page 3-1 for guidance on screening procedures or contact the following specialist

Address	Dr. Steven C. Passoa National Lepidoptera Specialist USDA–APHIS–Plant Protection and Quarantine The Ohio State University Museum of Biodiversity 1315 Kinnear Road Columbus, OHIO 43212 Telephone: 614 688 4471 Fax: 614 688 4487

Detection Survey

Use a detection survey to determine if the false codling moth exists in an area. Conduct a detection survey by using a combination of the following survey tools:

- Traps baited with pheromone
- Visual inspection of plants, fallen fruit, nuts, bolls, etc.
- Soil sampling

Search for the false codling moth wherever hosts are growing. See Hosts on page C-1 for a list of plants to check. In your survey, consider human and natural means of dispersal.

Positive results indicate that false codling moth is present. Based strictly on a detection survey, it is invalid to claim that a pest does not exist in an area if the trapping results are negative. Negative results are valuable clues to pest movement, particularly when considered with positive trapping results from similar areas.

Once the moth has been detected in an area, conduct a traceback investigation to determine its source. **See Traceback Investigation on page 4-5** for more information.

Monitoring Survey

Use a monitoring survey to evaluate the effectiveness of an action taken to contain, suppress or eradicate the pest. Use the same survey tools as detection and delimiting surveys.

Delimiting Survey

Once the false codling moth has been detected, use a delimiting survey to gather sufficient information about the pest population to facilitate containment, suppression or eradication. Sticky traps baited with a pheromone are the most efficient tools to use for delimiting false codling moth. If necessary, supplement trapping with visual inspection.

Use the delimiting survey decision table (Table 4-2) and the trapping scheme (Figure 4-1 on page 4-5) as guides when conducting a delimiting survey.

TABLE 4-2 Decision Table for Delimiting False Codling Moth

If you find:	In an area that is:	Then take this action:	And supplement with:
One or more adults	Apparently in the original infestation site or core	Set 36 traps baited with pheromone per square mile at or near the original infestation site.	Visual inspection
One or more (any stage)	Within a 1 square mile area	Set 36 traps baited with pheromone per square mile in 9 square miles around the original area.	Visual inspection of 100 hosts per square mile in the 9 square mile area
	Within a 6 square mile area	Set 36 traps baited with pheromone per square mile in 25 miles around the original area.	Visual inspection of 100 hosts per square mile in the 25 square mile area
One or more egg masses or larvae	All locations	→	Collect host material (fruits, nuts, berries, heads of grain, and seeds) within 216 yards of the specimens and rear to the adult stage for identification AND collect soil samples within 200 yards of any larval or egg detection and at any spot where dropped, especially prematurely dropped fruit, occur; examine soil for larvae, cocoons and pupae.

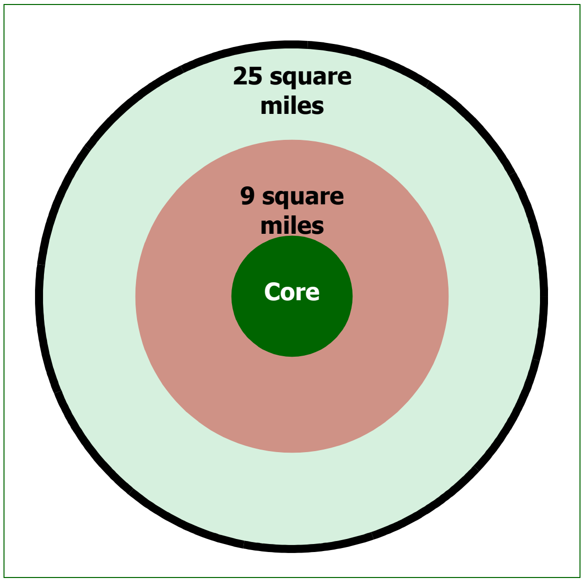

FIGURE 4-1 Trapping Scheme for False Codling Moth. Begin by setting 36 traps per square mile in the core area where false codling moth was detected.

Traceback Investigation

Use a traceback investigation to locate the source of an introduction after false codling moth has been detected. Traceback investigations are necessary for determining if an isolated detection is spurious—for example, the moth was conveyed into an area by air currents—or if it is evidence of an established population. Typically, if a single false codling moth is found in an area far removed from a port of entry or host plant, it was likely transported to the site. The same is true for isolated detections during cool seasons. Remember that false codling moth is inactive at air temperatures lower than 10°C (50°F).

Use wind field maps to plot the possible path of the false codling moth. Calculate the estimated day and time of arrival (based on the circumstances at the site and likely air mass movements) and work backward in time and space to construct a logical path.

Site circumstances that provide clues to the estimated time of arrival include the following types of detections:

- Associated with the arrival of a weather system
- Adults with no evidence of larval feeding
- Inland locations away from obvious ports of entry
- Ending abruptly outside a given area
- New generation or stage in the life cycle
- Sudden outbreaks or increases in numbers not associated with local breeding populations

Once the path of the moth is plotted, carry out surveys along the path until the likely introduction site is located. Likely origins include port environs, areas where over-wintering is possible, or agricultural areas where hosts are abundant. Allowing for the imprecision of this method, surveys add weight to conjecture about the origin of an introduction.

Computer generated atmospheric trajectory analyses are available to help identify potential sources of infestation and to trace the probable movement of plant pests with air masses. **See Predicting Insect Development on page A-6** for more information on this topic,

Visual Inspection of Plants

Examine host fruits, nuts, berries, heads of grain, corn ears, and seed from the core and buffer areas. Look for plants showing signs of poor growth or rot; holes in fruit, nuts or bolls; adults hidden in foliage; and crawling larvae. False codling moth larvae feed internally, so that on most hosts there are few external symptoms.

Check crop fields, fencerows, ditch banks, roadsides or other habitats for suitable hosts. Areas with damaged or poorly growing plants should receive priority in the survey.

Use **Figure 4-2 on page 4-7** as a guide when examining plants within fields. Follow a similar sampling pattern for each field surveyed. Collect samples at least 75 feet from the edge of five different locations. At each sample location within the field, inspect at least 10 plants from three adjoining rows (or at equally spaced intervals).

Avocado

Look for lesions on the skin of fruit. Larvae enter through the skin but are unable to develop in avocado fruit. However, the damage caused by false codling moth develops into a raised crater on the fruit surface with an inconspicuous hole in the center where the larva has entered (Du Toit et al. 1979). Granular excreta can also be seen. **See Avocado on page 4-7** for more information.

Citrus

On oranges, look for a brown patch on the skin, usually with evidence of a hole bored in the center, sometimes with a dark brown frass exuding from the hole (CPC 2004). Oranges or other citrus can also drop fruit prematurely. **See Citrus on page 4-7** for more information.

Cotton

In cotton, larvae feed on well formed bolls. In the United States, this stage of plant development corresponds with June and July (Angelini and Labonne 1970). Look for signs of damage to the boll as well as premature or unusual boll drop. **See Cotton on page 2-6** for more information.

Macadamia

False codling moth larvae burrow into the green husk of macadamia fruit and occasionally through the nut shell to feed on the kernel (van den Berg 1995). Examine leaves and the surface of the nuts for signs of eggs and larvae. **See Macadamia on page 2-6** for more information.

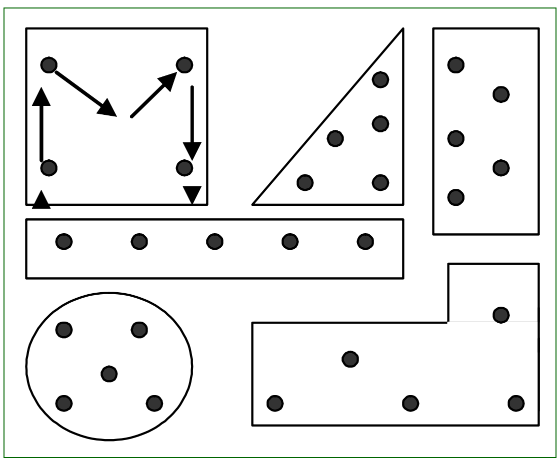

FIGURE 4-2 Sampling Pattern for Visual Inspection of Plants. Bullets indicate random locations for inspecting plants in various shaped fields.

Soil Sampling

Collect soil samples within 200 yards of any larval or egg detection and at any spot where dropped—especially prematurely dropped—fruit occur. Soil samples should consist of loose surface soil and any debris. Examine soil for larvae, cocoons and pupae. **See Identification on page 3-1** for more information on identifying false codling moth.

Trapping

Use yellow delta traps baited with a pheromone lure to catch false codling moth. Bait traps with a mixture of (Z)-8-Dodecen-ol acetate and (E)-8-Dodecen-ol acetate at a ratio of 50:50. Service traps weekly. **See Resources on page A-1** for information on trapping supplies.

Initiate sampling at the site of the detection. Set out traps at the site of the detection and in each square mile in the first and second buffer areas in a standard grid array. In tree crops, suspend traps from the tree limbs at a height of 4 ½ feet (1.5 m). In row crops, place traps on stakes at the same height as the crop. Replace bait at midseason or every 8 weeks, whichever is shorter. In wild hosts, follow the same guidelines for placement of traps.

Host Collecting and Holding

If you find eggs or larvae at any location, collect host material (fruits, nuts, berries, heads of grain, and seeds) within 216 yards of the specimens and rear to the adult stage for identification. **See Rearing on page 3-5** for more information.

Orientation of Survey Personnel

Experienced personnel should train their replacements. Adequate training on survey techniques and procedures will likely require three working days.

Survey Records

Survey records and data recording formats should be standardized. Maintain survey records, noting the areas surveyed, sites trapped, dates, locations, and hosts. All survey data must be forwarded to the Coordinator, State Cooperative Agriculture Pest Survey (CAPS), for entry into the National Agricultural Pest Survey Information System (NAPIS).

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False Codling Moth

Chapter 5

Regulatory Procedures

Contents

Introduction

Use *Chapter 5 Regulatory Procedures* to learn about the rules that must be followed by regulatory personnel when conducting pest survey and control programs.

Instructions to Officers

Officers must follow instructions for regulatory treatments or other procedures when authorizing the movement of regulated articles. Understanding the instructions and procedures is essential when explaining procedures to persons interested in moving articles affected by the quarantine and regulations. Only authorized treatments can be used in accordance with labeling restrictions.

Find instructions for regulatory treatments in the PPQ *Treatment Manual*. If a regulatory treatment does not appear in the manual, the proposed treatment will be reviewed and tested by CPHST treatment specialists.

Issuing an Emergency Action Notification

An Emergency Action Notification (EAN) can be issued pending positive identification or further instruction from the USDA–APHIS–PPQ Deputy Administrator.

If necessary, the Deputy Administrator will issue a letter directing PPQ field offices to initiate a specific emergency action under the Plant Protection Act of 2000 until emergency regulations can be published in the *Federal Register*.

Emergency Quarantine Action

The Plant Protection Act of 2000 provides for authority for emergency quarantine action. This provision is for interstate regulatory action only. Intrastate regulatory action is provided under state authority. However, if the Secretary of Agriculture determines that an extraordinary emergency exists and that the measures taken by the state are inadequate, USDA can take intrastate regulatory action provided that the governor of the state has been consulted and a notice has been published in the *Federal Register*. If intrastate action cannot or will not be taken by a State, PPQ might find it necessary to quarantine an entire State.

Access to Private Property

PPQ works in conjunction with state departments of agriculture to conduct surveys, enforce regulations, and take control actions. PPQ employees must have permission of the property owner before accessing private property. If an extraordinary emergency is declared or if a warrant is obtained, PPQ can enter private property without owner permission. PPQ prefers to work with the state to facilitate access when permission is denied; however, each state government has varying authorities regarding accessing private property. A General Memorandum of Understanding (MOU) exists between PPQ and each state. PPQ officers must have permission of the owner before accessing private property. For clarification, check with your state plant health director (SPHD) or state plant regulatory official (SPRO) in the affected state.

Regulated Articles

Regulated articles include the following:

- Any above ground part listed as a host that exists in the regulated area (**See Hosts on page C-1**)
- Plant parts liable to carry the pest in trade or transport, including the following:
  - Fruits (including pods)
  - Flowers, inflorescences, cones, calyxes (CPC 2004)
- Nursery stock with fruit, berries, and other fruiting structures
- Debris and leaves from infested fields, groves and gardens from within regulated areas
- Any other products, articles, or means of conveyance, of any character whatsoever, when it is determined by an inspector that they present a hazard of spread of the false codling moth and the person in possession thereof has been so notified

Quarantine Actions

Regulatory action will be required under the following conditions:

- More than one moth is found in an area less than 6 square miles within one estimated life cycle
- One mated female, or a larva, or a pupa are detected
- A single moth is detected that is determined to be associated with a current eradication project

Regulated Establishments

Field personnel will attempt to detect the pest within the regulated area at all establishments where regulated articles are sold, grown, handled, moved or processed. The following types of establishments might be included:

- Airports
- Landfill sites
- Processing plants
- Produce markets
- Flea markets
- Nurseries
- Horticultural shops
- Flower shops
- Farms
- Home gardens
- Any other establishment that handles regulated articles

Surveys can be set up at establishments deemed to be at risk by project personnel. Survey personnel should install and service pheromone traps at regulated establishments. Service traps weekly if catches of insects are high, or every two weeks if trap catches are low.

Use of Pesticides

The PPQ *Treatment Manual* and this document identify the authorized pesticides, and describe the methods and rates of application, and any special application instructions (**See Control on page 6-1**). Concurrence by PPQ is necessary before using any other pesticide or procedure for regulatory purposes.

Approved Regulatory Treatments

Approved regulatory treatments appropriate for this pest are determined by program management or a technical advisory committee in conjunction with the CPHST. Check the PPQ *Treatment Manual* for current recommendations.

Treatment options include the following:

- Sanitation
- Destruction of wild and cultivated hosts
- Application of recommended insecticides (aerial or ground)
- Application of other cultural controls
- Application of biological controls

Principal Activities

Regulatory activity depends, among many other factors, on the degree of the infestation. For example, it might not be necessary to safeguard vegetable stands throughout the regulated area if they are engaged in local retail activity only and the infestation is limited.

Principle activities for conducting a regulatory program to contain false codling moth include the following:

- Advise regulated industry(ies) of required treatment procedures.
- Supervise, monitor, and certify commodity treatments of commercial lots of regulated articles.
- Make regulatory visits to the following:
    - Security and airline personnel
    - Vegetable stands
    - Flower stands
    - Local growers, packers, and processing plants
    - Farmer's associations, produce markets, and flea markets
    - Commercial haulers of regulated articles
    - Public transportation
    - Post offices
- Visit canneries and other processing establishments.
- Monitor the movement of waste material to and from landfills to ensure adequate disposal of regulated articles.
- Monitor the movement of regulated articles through major airports and other transportation centers.
- Observe major highways and quarantine boundaries for movement of host materials.

Removing Quarantines

Project managers identify and remove areas from quarantine requirements after the false codling moth is declared eradicated. Eradication is assumed when sufficient time, equal to three false codling moth life cycles, has passed since the last specimen recovery. At minimum, one life cycle must elapse after control activities have ceased. APHIS will publish a Notice of Quarantine Revocation in the *Federal Register* when areas are removed from quarantine requirements.

Orientation of Regulatory Personnel

Initially, program personnel will be limited to those already trained or experienced. Experienced individuals train their replacements. A training period of three working days should be sufficient for the orderly transfer of these functions.

Regulatory Records

Maintain standardized regulatory records and database(s) in sufficient detail to carry out an effective, efficient, and responsible regulatory program.

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False Codling Moth

Chapter 6

Control

Contents

Introduction

Chapter 6 Control provides information about methods used to eradicate, manage, or prevent an infestation of false codling moth. Pest control includes those measures taken to eradicate or manage a pest population. Consider all strategies before beginning a program.

False codling moth is difficult to eradicate, especially if it becomes established. The pest is most effectively managed or prevented with sanitation and host destruction. Sanitation, host destruction, and other methods of control are discussed within this chapter.

TABLE 6-1 Decision Table to Eradicate or Manage Khapra Beetle

If you want to:	Then go to:
Eradicate the pest	**Eradication on page 6-4**
Contain or suppress the pest	**Management on page 6-6**

Laws Pertaining to Pesticide Use

The Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA) authorizes the Environmental Protection Agency (EPA) to regulate pesticides. All persons using and applying pesticides should understand the laws pertaining to pesticide use and application. The following are provisions of FIFRA that are most pertinent to emergency pest control programs:

- Restricted use pesticides must be applied by a certified applicator.
- Use of any pesticide inconsistent with the label is prohibited.
- Violations can result in heavy fines or imprisonment.

States can register pesticides on a limited basis for local needs according to the following Sections:

- **Section 18**—EPA administrators can exempt federal or state agencies from FIFRA if it is determined that emergency conditions exist that require such exemptions.
- **Section 24**—A state can provide registration for additional uses of federally registered pesticides formulated for distribution and use within that state to meet special local needs in accordance with the purposes of this act.

For additional information concerning exemptions, see the Emergency Programs Manual, Section 14. Contact Environmental Services staff to assure that any pesticide being considered as part of an eradication program conforms to pesticide use requirements. Obtain all required environmental documentation before beginning.

Efficacy

Use a monitoring survey (**Monitoring Survey on page 4-3**) to evaluate the effectiveness of an action taken to eradicate, contain, or suppress the pest. Use the same survey tools as used for detection and delimiting surveys.

The application and use of insecticides and other controlled substances should be assessed through the use of an appropriate monitoring program. The monitoring program will include at least the evaluationn of dye cards to monitor the following elements of aerial application:

- Droplet size
- Droplet distribution
- Identification of wind drift components
- Verification of spray block boundaries
- Identification of skips

Find instructions to evaluate efficacy of spray applications in the PPQ Treatment Manual.

Environmental Monitoring

Environmental monitoring is an important consideration in all programs. Contact PPQ Environmental Monitoring staff to learn if environmental monitoring is required. Environmental Services staff can evaluate environmental impact by monitoring the following:

- Air, to detect residual airborne pesticides
- Water, to detect insecticide levels resulting from direct application, leaching, and runoff
- Soil, to determine insecticide levels and residues
- Foliage, to detect residues

- Non-target organisms before, during and after applications and post treatments, to determine impact of pesticides

Treatment Duration

Continue eradication measures for at least two life cycles of the false codling moth. Monitor the success of the program for at least one life cycle after the termination of eradication measures.

Orientation of Personnel

Experienced personnel will train their replacements. A training period of three days should be sufficient for training.

Records

Program personnel must maintain records and maps noting the locations of all detections, the number and type of treatments, and the materials and formulations used in each treated area.

Treatment Options

Consider the treatment options described within this chapter when taking action to eradicate, contain, or suppress false codling moth. A review of the current literature has yielded many promising tools. Some tools were successfully used outside the United States; others were included in the *Guidelines* based on observations that they may prove to be successful in the United States after further investigation.

Prescribe treatments after considering the following:

- Results of detection and delimiting surveys (**See Survey on page 4-1** for information on surveys.)
- Local conditions
- Pest development and emergence
- Current status of the pest
- Current status of all control options

Treatment can include any of the following options, used singly or in combination:

Sanitation and host destruction—False codling moth is most effectively managed with sanitation (Newton 1998) and host destruction. **See Sanitation on page 6-6** and **Destruction of Wild and Cultivated Hosts on page 6-7**.

Application of other cultural controls—In conjunction with insecticides or other controls, cultural controls can be useful in both eradication and management programs. **See Cultural Controls on page 6-6**.

Application of insecticides—Insecticides can be effective tools in eradication programs if application is properly timed to coincide with egg laying and hatching. See Insecticides on page 6-8.

Application of synthetic pheromones—Mate-finding and reproduction are chemically-mediated processes that can be manipulated to eradicate or manage false codling moth. See Synthetic Pheromones on page 6-10.

Application of sterile insect technique—If false codling moth is detected early, or the infestation occurs in a limited area, sterile insect technique can be an effective means of eradicating false codling moth. See Sterile Insect Technique on page 6-12.

Release of biological control organisms— Biological control organisms can be effective in suppression or management and, if used properly, can aid in efforts to eradicate an infestation. See Biological Control on page 6-11.

Eradication

Eradication is the application of phytosanitary measures to eliminate a pest from an area. Eradication is the first priority to consider when a new pest has been introduced. Plan to eradicate the false codling moth if the following conditions exist:

- Pest population is confined to a small area.
- Infestations are found at only a few locations.
- Detection occurs soon after the introduction.
- Pest population density is low.

Treatment Area

Once a decision has been made to eradicate the false codling moth, use the decision table (Table 6-2) to define the treatment area.

TABLE 6-2 Decision Table for Eradication Treatment Area of False Codling Moth

If this number is detected:	In an area of this size:	Then treatment will commence and extend this distance beyond the detection:
1 to 5 larvae, pupae or gravid females OR 2 to 5 males or virgin females	Less than 6 square miles	200 yards
6 or more of any stage	Greater than 6 square miles	2 1/2 miles

Application of Insecticides

See Insecticides on page 6-8 for information on selection of insecticides.

Approved Spray Treatments

Aerial Spray

Aerial application of an insecticide should be initiated immediately. Apply aerial sprays at the prescribed intervals over a period equal to two life cycles. The number of applications will vary depending on the day degree accumulations in the infested area. The area to be sprayed will extend a minimum of 1 1/2 miles (2.4 km) beyond any known infestation. After an estimated two generations of negative trapping, discontinue spray operations. Weather conditions may dictate changes in spray schedule.

Ground Spray

Initiate ground application of insecticide immediately. All host plants which provide for reproduction of the false codling moth on the infested property, adjacent property, and within 216 yards (200 meters) of the known infestation will be sprayed at the prescribed intervals. On properties which cannot be aerially sprayed or where inclement weather precludes the use of aerial treatments, ground treatments may be used to maintain a viable insecticide application at the detection location and within 216 yards (200 meters) surrounding it. Ground spraying may be discontinued after an estimated two generations of negative trapping or after the iniation of aerial treatment.

The decision to apply insecticides will be based on the best weather information available. In the event rain washes an application from the foliage, plans will be implemented to reapply the insecticide. Retreatment should **not** be considered if weather reports indicate a 50-percent or greater chance of precipitation in the 48-hour period following washoff.

The objective is to maintain a viable spray in the eradication zone and prevent environmental contamination. Any treatment or retreatment recommendations will be considered based on environmental monitoring.

Supplements to Spray Treatments

Use the following methods to supplement aerial and ground spraying (Table 6-3).

TABLE 6-3 Supplemental Methods of Control for False Codling Moth

Where:	Then use this treatment:	And supplement with:
Properties with larvae in fruit	Ground or aerial spray	Strip all preferred host fruit within 216 yards (200 m) of the larval site if practicable.
Properties with confirmed larval, pupal, and/or egg infestations and the environs within 216 yards (200 m) surrounding it	Approved soil treatments applied within the drip line of all host plants; at prescribed intervals.	
Hosts are present within the core and buffer areas	→	Sanitation in orchards, nurseries, farms, gardens, and other establishments
Area of infestation is very limited	→	Destruction of host fruits or crops (cotton, corn field crops) by disking or plowing. All host material must be left completely buried by the disking or plowing process.

Management

Management includes steps taken to contain or suppress a pest population. Containment is the application of phytosanitary measures in and around an infested area to prevent spread of a pest. Suppression is the application of phytosanitary measures in an infested area to reduce pest populations. False codling moth is most effectively managed with the cultural controls described below.

Cultural Controls

In conjunction with insecticides or other tools, cultural controls can be useful in both eradication and management programs.

Important Cultural controls can be subject to obtaining environmental documentation under the National Environmental Policy Act (NEPA) and the Endangered Species Act (ESA). Check with the program manager to make sure such documentation is in order.

Sanitation

Carry out sanitation at weekly intervals in nurseries, farms, gardens, and other establishments where hosts are present within the core and buffer areas. Depending on the circumstances and equipment available, use the following techniques before fruit are the size of marbles:

- Remove infested fruit from cultivated and wild host trees.
- Remove all fallen fruit (corn ears, nuts, pods, bolls, etc.) from the area, including fruit from nearby wild hosts, especially acorns.
- Remove all out of season fruit.

Destruction of Wild and Cultivated Hosts

Use the following methods to destroy all host material completely:

Burning, Disking or Plowing—Collect, pile and burn host material if local ordinances permit. Disk under residue or bury in an approved landfill. Bury host material under two to three feet of soil; firmly pack the soil on top to prevent escape (Gunn 1921).

Soil Disturbance—Cultivation of the soil, especially in the spring or early summer, is effective. Cultivation destroys large numbers of false codling moth pupae in the soil that have left the fallen fruit. Give particular attention to the soil under the drip line of perennial crops such as trees. If hosts are annuals or biennials, cultivate the entire field (Gunn 1921).

Feed to Animals—Some host materials can be fed to animals. Exercise caution when considering this disposal option.

Control Weeds—Trim, cultivate, or apply herbicides to grasses and weeds growing along roadsides, in fields, or in row crops. False codling moth can reproduce on a wide variety of hosts. This pest can attack new, unreported hosts. Therefore, keeping these areas clean can help to suppress this pest.

Inspect and Clean Vehicles—Inspect vehicles, trucks, wagons, and other vehicles used in host fields or used to transport host material, to avoid accidental movement of host material with eggs or larvae.

Miscellaneous

Rotate Crops—Crop rotation can be a feasible method of control in seasonal or biennial crops. Be sure to rotate host crops with non-host crops.

Flood Fields—If a field can be flooded for at least two days, this technique can be an effective means to eradicate false codling moth. Pupae, which remain on or near the soil surface, will suffocate when soil is flooded.

Plant Trap Crops—Planting economically unimportant crops can divert false codling moth away from a valuable cash crop. In East Africa, sorghum and cissus show promise as trap crops against false codling moth in cotton (Hill 1975). Corn failed to be an effective trap crop in cotton in Uganda (Newton 1998).

Insecticides

Important The insecticides described below may **not** currently be registered in the United States for use against false codling moth. Please review **Laws Pertaining to Pesticide Use on page 6-1** for more information, and read the product label to learn more. Some of the following are guidelines developed to control false codling moth in Africa. The insecticides will likely be effective in the United States, if they are registered. For updates on the status of any pesticide, contact the US–EPA or the registrant or distributor of each product.

False codling moth is difficult to eradicate or mangage with insecticides for the following reasons:

- Since larvae spend most of their lives inside the host, they are inaccessible to non-systemic insecticides.
- The pest has developed resistance to commonly used systemic and non-systemic insecticides (Reed 1974).

In oak forests, pesticides can be ineffective due to survival of larvae and pupae in fallen acorns on the ground (Jim E. Carpenter, USDA–ARS–Crop Protection and Management Research, pers. comm.). **See Sanitation on page 6-6** to manage false codling moth in this environment.

Apply insecticides immediately upon detection of false codling moth. Apply insecticides in the late afternoon, evening or at night to coincide with the nocturnal habits of adults and larvae. After an estimated two generations of negative trapping and survey, applications can be discontinued.

Choose from the following types of insecticides:

- Biological insecticides
- Insect growth regulators
- Other pesticides

Biological Insecticides

***Bacillus thuringiensis kurstaki* (BTk)**

Use BTk in ecologically sensitive areas, urban areas, or in an area where chemical insecticides should be alternated or discontinued. Apply as a full-coverage spray when larvae are present. Repeat at 10—14 day intervals while larvae are active. Effectiveness of aerial delivery is enhanced if done by helicopter, since the downdraft turns the leaf surfaces for better exposure. Mugo et al. (2001) found that BTk was more effective than dimethoate, when both were applied in Kenya to corn to control false codling moth.

Beauveria bassiana

Beauveria bassiana has been found among pupae in leaf litter in South Africa. However, attempts to culture and apply it were unsuccessful (Newton 1998).

***Cryptophlebia leucotreta* granulovirus (CIGV)**

Efficacy of this virus to control false codling moth remains under study (Moore 2003; Mlanjeni 2005).

Naturalyte (Success®)

Naturalyte is composed of the fermentation products of *Saccharopolyspora spinosa*, a naturally occurring soil organism (**See Success© (Spinosyn) on page A-2**). Efficacy of this product to control false codling moth has **not** been studied.

Insect Growth Regulators

Teflubenzuron

CME 134 (teflubenzuron) effectively controls false codling moth in citrus (Newton 1989). However, it is currently **not** registered for use in the United States.

Triflumuron

Triflumuron effectively controls false codling moth in citrus (Newton 1989). However, it is currently **not** registered for use in the United States.

Tebufenozide

Tebufenozide mimics the action of the natural insect hormone that induces the molting and metamorphosis process in insects. Mimic©2LV is currently registered for use in the United States. Confirm® is **not** registered for use against false codling moth.

Other Pesticides

Sweet and Hollings (1983) recommended the following pesticides for control of false codling moth in the Republic of South Africa:

- Azinphos-methyl Guthion® Solupak 50%
- Methomyl
- Cypermethrin 200 EC

USDA–APHIS–PPQ (1983) recommended application of the following insecticides to eradicate the false codling moth in the United States:

- Azinphos-methyl (Gusathion®, Guthion®)
- Diazinon
- Fenvalerate
- Permethrin

Check with the registrant of each pesticide to determine whether it can be applied in the United States. Since 1983, fenvalerate was voluntarily cancelled by the registrant. Azinphos-methyl, diazinon, and permethrin are currently registered by the US–EPA for use in the United States (US–EPA, 2007a-d).

Synthetic Pheromones

Mate-finding and reproduction are chemically-mediated processes that can be manipulated to eradicate or manage false codling moth.

Mating Disruption

Releasing large concentrations of commercially available synthetic pheromone [(E)-7-dodecenyl acetate, (E)-8-dodecenyl acetate and (Z)-9-dodecenyl acetate] in the infested area can interfere with the ability of the male to find the female, thereby disrupting reproductive success.

Important The synthetic pheromones described below might not currently be registered in the United States for use against false codling moth. Please review **Laws Pertaining to Pesticide Use on page 6-1** for more information, and read the product label. The following are guidelines developed by researchers to control false codling moth in Africa. The synthetic pheromones will likely be effective in the United States, if they are registered. For updates on their status, contact the registrants or distributors of each product.

Exosect System®

Exosect System is a mating disruption system available from Exosect Limited in the United Kingdom. Exosect System uses a pheromone attractant to lure the male false codling moth. When the male contacts the dispenser, a powdered pheromone adheres to its antenna. The pheromone overwhelms the receptive sensors so the male is unable to detect a virgin female. Exosect uses minute amounts of pheromone (often less than the concentration produced by the moths) thereby leaving pollinators, parasites and predators unaffected. This product is under development. **See Resources on page A-1** for more information.

ARBICO® Product

ARBICO Organics has suggested that its false codling moth pheromone product might be used to disrupt mating of false codling moth, if applied in large concentrations. **See Resources on page A-1** for more information.

7-Vinydecyl acetate

Burger et al. (1990) reported that 7-vinydecyl acetate is a strong pheromonal inhibitor for false codling moth that might be useful for mating disruption.

Last Call FCM®

Last Call FCM is a combination of sex pheromones with permethrin, contained within a special applicator. The product is applied by hand using a precalibrated dispenser to deliver 50-microliter drops. Male moths are attracted by the pheromone. The moth dies on contact with the pheromone-pyrethrin combination, effectively denying most female moths a chance to mate. **See Resources on page A-1** for more information.

Biological Control

Biological control is a promising tool for eradication or management of a pest. However, the subject is beyond the scope of this document.

Planning a Biological Control Program

Biological control of false codling moth requires further investigation to be effective. The following guidelines should be used when planning a biological control program:

1. Identify an effective biological control agent with the potential to control the target pest or pests. Use **Table 6-4** as a guide. *Trichogrammatoidea cryptophlebiae* is a promising biological control organism since it is both effective and commercially available. Newton (198l and 1989) found that inundative releases of *T. cryptophlebiae* throughout the season were the best strategy in view of the long-term suppression of damage. *T. cryptophlebiae* is an egg parasitoid of close relatives of the false codling moth.
2. Locate a reliable source of the biological control agent(s).
3. Determine the following:
   - ❖ Release rates
   - ❖ Synchronization with the host
   - ❖ Temperature and other environmental requirements
   - ❖ Appropriate host plants
4. Coordinate the introduction of biological control agents with other strategies in a pest management program. For example, identify pesticides and cultural practices that are compatible.

See Suppliers of Beneficial Organisms on page A-1 for a list of resources.

TABLE 6-4 Biological Control Organisms Active Against False Codling Moth

Mechanism	Species	Family	Reference
Unknown	*Actia cuthbertsoni*	Tachinidae	CIBC 1984
Larval parasitoid	*Agathis bishopi*	Braconidae	Newton 1998; CIBC 1984
Unknown	*Agathis (Bassus)* spp.	Braconidae	CIBC 1984
Larval parasitoid	*Agathis leucotretae*	Braconidae	Newton 1998; CIBC 1984
Predator of pupae	Ants	Formicidae	Newton 1998
Pupal parasitoid	*Apophus leucotretae*	Ichneumonidae	CPC 2004; Newton 1998; CIBC 1984
Unknown	*Apanteles typhon*	Braconidae	CIBC 1984
Unknown	*Apanteles* spp.	Braconidae	CIBC 1984
Egg and larval parasitoid	*Chelonus curvimaculatus*	Braconidae	CPC 2004; Newton 1998; CIBC 1984
Unknown	*Chelonus* spp.	Braconidae	CIBC 1984
Unknown	*Elasmus johnstoni*	Elasmidae	CIBC 1984
Larval and pupal parasitoid	*Mintha* spp.	Tachinidae	Newton 1998
Predator	*Orius* spp.	Anthocoridae	Newton 1998
Larval parasitoid	*Oxycoryphe edax*	Chalcidae	Newton 1998
Unknown	*Phanerotoma curvicarinata*	Braconidae	CIBC 1984
Predator	*Rhynocoris albopunctatus*	Reduviidae	Newton 1998
Pupal predator	Shrew	Soricidae	Newton 1998
Egg parasitoid	*Trichogrammatoidea cryptophlebiae*	Trichogrammatidae	Newton 1988, 1989, 1998; CIBC 1984
Egg parasitoid	*Trichogrammatoidea spp.*	Trichogrammatidae	CPC 2004

Sterile Insect Technique

The use of sterile insect technique (SIT) for control of false codling moth promises to be effective. However, it remains under investigation (Bloem et al. 2003). A supply of sterilized false codling moths is currently **un**available. Artificial diets are available [see Theron (1947), Schwartz (1972) and Carpenter et al. (2003) for more information], but overflooding ratios have not been worked out.

SIT has been an effective tool in other eradication and suppression programs. Pest control personnel at USDA–APHIS have used SIT to control the following pests:

- Mediterranean fruit fly (*Ceratitis capitata*)
- Screwworm fly (*Cochliomyia hominivorax*)
- Pink bollworm (*Pectinophora gossypiella*)

SIT employs radiation to sterilize large numbers of male insects. When released, the sterilized males compete with the viable males. This technique is most effective in species with females that mate only once. After mating with a sterile male, the female will usually lay sterile eggs, thereby reducing the reproductive success of the pest.

Many factors determine if a particular species is a good candidate for SIT, including the following:

- Competitiveness after irradiation
- Ability to be reared in large numbers
- Inherited sterility
- Development of a pheromone for monitoring

The release of completely sterile females and partially sterile males (able to produce sterile F1 progeny when mating with feral females) might be more effective for against false codling moth (Bloem United States Department of Agriculture (USDA)–Animal and Plant Health Inspection Service (APHIS)–Plant Protection and Quarantine (PPQ) developed this guide through discussion, consultation, or agreement with staff at USDA–Agricultural Research Service (ARS), universities, industries, and state departments of agriculture.

2003).

Consult with USDA–ARS–Crop Protection and Management Research scientist James E. Carpenter in Tifton, Georgia, for information on possible off shore supplies of sterilized false codling moths for a SIT program.

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False Codling Moth

Chapter 7

Environmental Compliance

Contents

Introduction

Use *Chapter 7 Environmental Compliance* as a guide to environmental regulations pertinent to the false codling moth.

A key element in designing a program or an emergency response is consultation with Environmental Services (ES), a unit of APHIS' Policy and Program Development staff. ES performs the following functions:

- Prepares environmental documentation such as environmental impact statements and environmental assessments to aid in making decisions
- Provides consultation on the topic of endangered species
- Coordinates pesticide registration and approvals for APHIS programs, ensuring that registrations and approvals meet the needs of programs and conform to pesticide use requirements

In addition, PPQ's Environmental Compliance team assists ES in the development of required documentation and implements any environmental monitoring that may be required of program activities. Refer to **Resources on page A-1** for additional information.

Disclaimer

All uses of pesticides must be registered or approved by appropriate federal, state, and/or tribal agencies before they can be applied. The information provided on pesticide labels may not reflect all of the actual information, including precautions and instructions for use, which you are required to follow in your specific state or locality. It is the responsibility of persons intending to use a pesticide to abide by the label, including labeling that has been approved for the particular state or locality in which the chemical is to be used, and to comply with all federal, state, tribal, and local laws and regulations relating to the use of the pesticide. Staff within APHIS programs are responsible for their compliance with applicable environmental regulations, which often include measures above and beyond those listed on pesticide labels.

National Environmental Policy Act

The National Environmental Policy Act (NEPA) requires that federal agencies document the potential adverse effects of their actions. The process often requires public input. The exact nature of the documentation and public involvement is dictated by the potential for adverse effects and the significance of those effects.

It is likely that most pest control responses will include actions that need up to 30 days of public comment prior to initiation. Therefore, it is imperative to involve Environmental Services and Environmental Compliance early in the planning process. Doing so assures public involvement and a quick response.

Depending on the proposed program, NEPA requirements will be met with a categorical exclusion, environmental assessment, or environmental impact statement. Some programs can prepare their own NEPA documentation. Contact Environmental Services or Environmental Compliance if you are unsure which document should be prepared, or if you have little experience writing such documents. Refer to **Resources on page A-1** for contact information.

Endangered Species Act

The Endangered Species Act (ESA) requires that all federal actions, including emergency responses, do not harm federally protected threatened or endangered species. Before an action can begin, it must be determined if protected species are in the project area. If such species are present, measures must be put in place to protect them from potential adverse effects of the action. Such work requires coordination with the U.S. Fish and Wildlife Service and/or the National Marine Fisheries Service.

Several methods are available to ensure compliance with ESA, but the exact one chosen is dictated by the nature of the emergency, proposed response, and location. As soon as possible in the early stages of the response, contact staff at Environmental Services or Environmental Compliance, who can provide the necessary guidance and support in conducting the necessary analyses and developing the required documentation. Refer to **Resources on page A-1** for contact information.

Federal Insecticide, Fungicide, and Rodenticide Act

The Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA) requires that chemicals used for control have approved labels and that all label requirements are followed. These requirements can include applicable uses, maximum application rates, handling instructions, and personal protective equipment. If no label is available for the emergency in question (i.e., the pest of concern is not listed as one for which the chemical may be used), it is possible to obtain a new label or a label exemption. If a label change is needed or no label can be located for your program needs, immediately contact Environmental Services, who can assist in label changes and emergency use exemptions. Refer to **Resources on page A-1** for contact information.

Other Laws

The National Environmental Policy Act, Endangered Species Act, and the Federal Insecticide, Fungicide, and Rodenticide Act, are of critical importance to all pest control programs, but other laws may apply depending on program locations and activities. These include the Migratory Bird Treaty Act, the Coastal Zone Management Act, and the Bald and Golden Eagle Protection Act. By including Environmental Services and Environmental Compliance early in program planning, guidance can be provided on meeting the requirements of these and other laws that may apply. Refer to **Resources on page A-1** for contact information.

Environmental Monitoring

Environmental monitoring of APHIS pest control activities may be required as part of compliance with the above laws, as requested by program managers, or as suggested to address concerns with controversial activities. This is especially true for less benign chemical controls and aerial application of chemicals.

Monitoring may be conducted with regards to worker exposure, quality assurance and control, off-site deposition, or program efficacy. Different tools and techniques are used depending on the monitoring goals, program chemicals, and control techniques. Environmental monitoring is coordinated by Environmental Compliance (EC). Staff from EC will work with the program manager to develop an environmental monitoring plan, conduct training to implement the plan, provide day-to-day guidance on monitoring, and provide an interpretive report of monitoring activities.

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False Codling Moth

Chapter 8

Pathways

Contents

Introduction

The *Pathways* chapter provides information on the interceptions of false codling moth at U.S. Ports.

Source	Information in this chapter was compiled from the following source, unless noted otherwise: Touhey, P. 2005. *Trogoderma granarium*. Personal Report to J. Stibick on Interceptions in the PIN 309 Reports: 4 pp.

Inspectors intercepted false codling moth 174 times at United States ports of entry during the period 1985—2003. The pest was detected on 22 host plants originating in 15 African countries. Hosts included *Capsicum* spp., *Solanum* spp., *Citrus* spp., *Zea mays* (seeds) and *Cola* spp. (seeds). About 41% of the interceptions were on peppers (*Capsicum* spp.). The infested items were intercepted en route to 39 States, including Texas and California, where the pest might easily establish itself (Venette et al. 2003).

Travel

Since 1985, 94% of United States interceptions of false codling moth were from international airline passenger baggage.

Natural

Natural spread of false codling moth to the United States is unlikely.

Commerce

Since 1985, six percent of United States interceptions of false codling moth were from cargo or stores (8.8% were from stores, 0.6% were from general cargo and 0.6% were from permit cargo). One maritime interception was reported (from stores). In July, 2002, the pest was intercepted in Memphis, Tennessee, from cargo on *Capsicum* spp. from Africa. In August, 2004, the pest was intercepted in Baltimore on *Capsicum* spp. from Ghana. The pest was intercepted on grapes from South Africa in 2001 (once), 2002 (twice) and 2005 (once) (Netshifhefhe 2005).

Countries of Origin

During the period 1985—2003, interceptions of false codling moth originated from 15 African countries on 22 host plants. Most interceptions were from Nigeria and Ghana.

Destinations

During the period 1985—2003, approximately one third or 57 of the interceptions were found at 10 ports in areas where false codling moth could establish itself (**Table 8-1**).

TABLE 8-1 Interceptions of False Codling Moth at United States Ports of Entry During The Period 1985-2003

Port Of Entry	Number Of Interceptions
Houston, Texas	17
Los Angeles, California	16
Dallas, Texas	6
Atlanta, Georgia	5
Miami, Florida	4
San Francisco, California	3
St. Louis, Missouri	2
Memphis, Tennessee	1
Galveston, Texas	1
Orlando, Florida	1
Total	174

Risk of Establishment

False codling moth is a significant import threat to the United States for the following reasons:

- False codling moth is a pest of economic importance to many crops in its native habitat including avocado, citrus, corn, cotton, macadamia, and peach and plum.
- False codling moth could easily become established in the southern and southwestern United States.
- Establishment of false codling moth in the United States could result in economic losses of billions of dollars.

- False codling moth has been frequently intercepted at United States ports of entry in both cargo and passenger luggage.
- False codling moth larvae are difficult to detect once they enter the fruit.
- False codling moth eggs are laid singly and are difficult to detect.

False Codling Moth

Glossary

aerial treatment. Application of insecticide to a treatment area by aircraft.

APHIS. Animal and Plant Health Inspection Service

array. Arrangement of traps within one square mile.

array sequence. Arrangement of traps (arrays) from the core area outward to the perimeter or buffer areas.

block. Unit (e.g., 1 square mile area) of a detection survey in which all survey activities are conducted.

buffer area. Survey area that is:

- Beyond the core block
- 1—2 miles from the perimeter of a regulated area, or
- 50 miles from the core of a regulated area (in an extended survey)

calling. Emission of sex pheromones by the female moth to attract mates.

cold treatment. Exposure of a host product to cold temperatures lethal to a target pest; can be used alone or with fumigants.

confirmed detection. Positive identification by a recognized expert.

containment. Application of phytosanitary measures in and around an infested area to prevent spread of a pest.

control. Suppression, containment or eradication of a pest population.

core area. Area of 1 square mile surrounding a confirmed detection.

crepuscular organism. Organism active in twilight hours.

day degree. Measure of physiological time using the accumulation of heat units (degrees) above an insect's developmental threshold for a 24-hour period.

delimiting survey. Determination of the extent of an infestation (e.g., distribution, density) in an area where an exotic species has been detected.

delta trap. Five-sided insect trap configured with three lateral sides arranged triangularly and equipped with a lure (i.e., pheromone), a baffled edge, and an adhesive surface inside to capture and secure attracted insects.

detection. Collection of any life stage of an exotic species.

detection survey. Activity conducted in a susceptible area not known to be infested with an exotic species to determine its presence.

developmental threshold. Minimum or maximum temperatures that support physiological development for a species.

diurnal organism. Organism that is active during the day.

eclosion. Insect leaving the egg, or the terminal molt into an adult.

eradication. Application of phytosanitary measures to eliminate a pest from an area.

epicenter. Initial site of an infestation.

evaluation survey. *See also* **Monitoring survey**. Conducting visual or trapping surveys in an area that has been treated with insecticide to evaluate the effectiveness of the treatment.

exotic species. Organism or pest species not native to or historically resident in North America.

fumigation. Application of an approved insecticidal chemical that enters the target pest's tracheal system in volatile form.

generation. Offspring of a parent population that move through the life cycle together.

ground spray. Insecticide application in droplet form, from equipment positioned on the ground or at the vegetation level.

holding. *See also* **host collection.** Collection and retention of infested host material for the purposes of determining characteristics of a pest's use of the material.

host. Species that provides food, shelter or reproductive requirements for another organism.

host collection. *See also* **holding**. Collection and retention of infested host material for the purposes of determining characteristics of a pest's use of the material.

infestation. Collection of the following pests:

- Two or more of an exotic species
- A pupa, larva, or a mated female from an area, or
- The detection of a single adult associated with a current infestation

infested area. Area surrounding a single detection site or a group of sites. The standard designated area of 2.5 miles is used, unless biotic or abiotic factors dictate adjustment of this area.

migratory. Species in which individuals habitually move from place to place usually in search of mates or egg-laying sites.

monitoring survey. ***See also* evaluation survey.** Conducting visual or trapping surveys in an area that has been treated with insecticide to evaluate the effectiveness of the treatment.

natural enemies. Living organisms found in a natural community that kill, weaken, or inhibit the biological potential of a pest species.

nocturnal. Active at night.

non-migratory. Species in which individuals typically do not move far from the area of their birthplace.

parasites. Parasites live on the host (frequently the adult stage) at one or multiple life stages. Parasites sometimes kill but usually merely debilitate the host.

parasitoid. Parasitoids live on the host (often an immature stage) when immature, but are free-living as adults. Parasitoids always kill the host. Like parasites, these organisms are typically host-specific, and some are obligate on certain hosts. They find hosts effectively even when host population numbers are not particularly dense.

pathogen. Agent, usually microbial, that induces illness. Entomopathogens induce illness in insects. These include baculoviruses (primarily those in the genus *Nucleopolyhedrovirus*), nematodes and fungi. Many are species-specific, and cause no collateral infection of other organisms.

phenology. Timing of recurrent biological events.

pheromone. *See* **sex pheromone.**

predator. Free-living organism that consumes substantial numbers of prey. They generally do not prey exclusively on one target species over the course of a season. However, when the population of one species is dense (e.g., when swarming or aggregating to mate; when larvae cluster on hosts) they can be very efficient.

PPQ. Plant Protection and Quarantine.

regulated area. Area that extends at least 2 1/2 miles in any direction from the epicenter of an infestation.

regulated articles. All known or suspected hosts of a confirmed infestation of an exotic species, including soil and any other suspected product or article.

regulatory survey. Trapping or detection program conducted around establishments where regulated articles are sold, handled, processed or moved.

sex pheromone. Chemical substance that is secreted by an insect to attract or to advertise reproductive competence to the opposite sex of the same species.

soil treatment. Application of an approved insecticide to the soil of nursery stock or within the drip line of host plants.

suppression. Application of phytosanitary measures in an infested area to reduce pest populations.

sweep net survey. Survey method in which a mesh net suspended around a hoop is swept through the air or around vegetation to collect insects.

trap survey. Determination of the presence of a pest through the use of randomly or strategically placed devices that capture insects (sometimes aided by an attractant). These traps are maintained and serviced on a schedule dictated by the goal of the survey.

visual survey. Examination of areas for eggs, larvae, pupae, cocoons, or other evidence that a particular insect species is present.

wing trap. Disposable, adhesive-coated capture device used primarily for surveying moths.

urban. Area containing a number of multiple- or single family dwellings.

USDA. United States Department of Agriculture.

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A

False Codling Moth

Appendix A

Resources

Suppliers of Beneficial Organisms

Suppliers of Beneficial Organisms in North America. A free 32-page booklet available online at http://www.cdpr.ca.gov/docs/ipminov/bensuppl.htm from:

California Environmental Protection Agency
Department of Pesticide Regulation
Environmental Monitoring and Pest Management Branch
1020 N. Street, Room 61
Sacramento, CA 95814-5604
Telephone: (916) 324-4100

Trichogramma wasps

BioResources Pty Ltd
Richard Llweellyn
P.O. Box 578
Samford, Queensland 4520
Australia
T: 07 3289 4919
F: 07 3289 4918
Richard@bioresources.com.au
http://www.bioresources.com.au/

Ceder Biocontrol
P.O. Box 356
Citrusdal 7340
South Africa
T: (022) 921-3403
F: (022) 921-3403
ceder@wol.co.za

Suppliers of Insecticides

Bacillus thuringiensis kurstaki

Southern Agricultural Insecticides, Inc.S
Hendersonville, NC
Palmetto, FL
Boone, NC
http://www.southernag.com/

U-Spray, Inc.
4653 Highway 78
Lilburn, GA 30087
Telephone: (770) 985-9388 or 1-800-877-7290
http://www.bugspray.com/

Valent U.S.A., Corporation
P.O. Box 8025
Walnut Creek, CA 94596-8025
Telephone: 1-800-6-VALENT
http://www.valent.com

Success© (Spinosyn)

Dow AgroSciences LLC
9330 Zionsville Road
Indianapolis, IN 46268
Telephone: 317-337-3000
Fax: 317-337-4096
http://www.dowagro.com/homepage/index.htm

Mimic© (Tebufenozide)

Rohm and Hass Company
100 Independence Mall West
Philadelphia, PA 19106-2399
Telephone: (215 592-3000
http://www.rohmhaas.com

Lannate SP (Methomyl)

E.I. du Pont de Nemours and Company
Crop Protection
Wilmington, Delaware 19898
Telephone: 1-888-6-DUPONT
http: www.dupont.com/ag/us

Guthion Solupak (Azinphos-methyl)

Bayer CropScience LP
P.O. box 12014
2 T.W. Alexander Drive
Research Triangle Park
North Carolina 207709
Telephone: 1-866-99BAYER

Pheromone Disruption and Traps

Last Call (Pheromone/Permethrin)

Insect Science SA
Private Bag x4019
Postnet Suite 378
Tzaneen South Africa 0850
Telephone: 015 345 144
http://www.insectscience.co.za/index.htm

John A. Pickett
IACR-Rothamsted, Harpenden, Herts, AL5 2JQ UK
Tel: 01582 763133 x2321 Fax: 01582 762595
john.pickett@bbsrc.ac.uk
http://www.rothamsted.bbsrc.ac.uk/

Pheromone Disruption

Exosect Limited
2 Venture Road
Chilworth Science Park
Southampton SO16 7NP, UK
http://www.exosect.com info@exosect.com
Telephone: 44 (0)23 8076 3838
Fax: 44 (0)23 8076 3828

Advanced Pheromone Technologies, Inc.
P.O. Box 417
Marylhurst, OR 97036-0417
Telephone: 877-244-9610
Fax: 971-327-8407
www.advancedpheromonetech.com

ARBICO Organics
P.O. Box 8910
Tucson, AZ, 85738-0910
Phone: 520-825-9785
Toll Free Phone: 1-800-827-2847
Fax: 520-825-2038
info@arbico.com
http://store.arbico-organics.com/

Association of Natural Biocontrol Producers
10202 Cowan Heights Drive
Santa Ana, CA 92705
Telephone: (714) 544-8295
http://www.anbp.org/

Yellow Delta Trap
ISCA Technologies, Inc.
P.O. Box 5266
Riverside, CA, 92521
Telephone: (909) 686-5008
Fax (815) 346-1722
http://www.iscatech.com/exec/index.htm

Insect Science SA
Private Bag x4019
Postnet Suite 378
Tzaneen South Africa 0850
Telephone: 015 345 144
http://www.insectscience.co.za/index.htm

Great Lakes IPM, Inc.
10220 Church Road
Vestaburg, MI 48891-9746
Telephone: (989) 268-5693 / (989) 268-5911
Toll Free: 1-800-235-0285
Fax:(989) 268-5311
http://www.greatlakesipm.com/

Scentry Biologicals, Inc.
610 Central Avenue
Billings, Montana 59102
(406) 245-3016 (800) 735-5323
Fax (406) 245-2790
Email: scentry@imt.net
http://www.scentry.com/

Trece Incorporated
7569 Highway 28 West
P.O. Box 129
Adair, Oklahoma 74330
Telephone: (918) 785-3061
Fax (918) 785-3063
custserv@trece.com

Insect Science South Africa
Private Bag X4019
Post net Suite 378
TZANEEN
0850
http: www.insectscience.co.za

Entomology Supplies

BioQuip Products, Inc.
2321 Gladwick Street
Rancho Dominguez, CA 90220
Telephone: (310) 667-8800
http://www.bioquip.com/

Wards Natural Science
PO Box 92912
Rochester, NY 14692-9012
Telephone: 800-962-2660
http://www.wardsci.com/

Carolina Biological Supply Co.
2700 York Road
Burlington, NC 27215-3398
Telephone: (800)334-5551
http://www.carolina.com/

Environmental Compliance

Environmental Monitoring, Categorical Exclusions—

USDA–APHIS–PPQ–Emergency and Domestic Programs
Environmental Compliance
4700 River Road, Unit 150
Riverdale, MD 20737
Telephone: 301-734-8247

Environmental Services

FIFRA, ESA, Environmental Assessments—

USDA–APHIS–Policy and Program Development
Environmental Services
4700 River Road, Unit 149
Riverdale, MD 20737
Telephone: 301-734-8565

Additional Resources

Beneficial Organisms

Cornell University—Weedon, C.R., A.M. Shelton, Y. Li, and M.P. Hoffmann. Biological Control: A Guide to Natural Enemies in North America. http://www.nysaes.cornell.edu/ent/biocontrol/

North Carolina State University—Biological Control Virtual Information Center. Center for IPM. North Carolina State University. http://cipm.ncsu.edu/ent/biocontrol/

Predicting Insect Development

University of California Statewide Integrated Pest Management Program 2003 (http://www.ipm.ucdavis.edu/WEATHER)

National Oceanic and Atmospheric Administration
U.S. Department of Commerce

Local Cooperative Extension Service

Private, State, university, or industry sources

B

False Codling Moth

Appendix B

Forms

PPQ 391 Specimens For Determination

This report is authorized by law (7 U.S.C. 147a). While you are not required to respond your cooperation is needed to make an accurate record of plant pest conditions.

See reverse for additional OMB information.

FORM APPROVED
OMB NO. 0579-0010

U.S. DEPARTMENT OF AGRICULTURE
ANIMAL AND PLANT HEALTH INSPECTION SERVICE

SPECIMENS FOR DETERMINATION

Instructions: Type or print information requested. Press hard and print legibly when handwritten. Item 1 - assign number for each collection beginning with year, followed by collector's initials and collector's number. Example (collector, John J. Dingle): 83-JJD-001.
Pest Data Section – Complete Items 14, 15 and 16 or 19 or 20 and 21 as applicable. Complete Items 17 and 18 if a trap was used.

FOR IIBIII USE
LOT NO.
PRIORITY

1. COLLECTION NUMBER
2. DATE — MO | DA | YR
3. SUBMITTING AGENCY — ☐ State Cooperator ☐ PPQ ☐ Other ______

SENDER AND ORIGIN

4. NAME OF SENDER
6. ADDRESS OF SENDER
ZIP

INTERCEPTION SITE

5. TYPE OF PROPERTY *(Farm, Feedmill, Nursery, etc.)*
7. NAME AND ADDRESS OF PROPERTY OR OWNER
COUNTRY/ COUNTY

PURPOSE

8. REASON FOR IDENTIFICATION *("x" ALL Applicable Items)*

A. ☐ Biological Control (Target Pest Name)
B. ☐ Damaging Crops/Plants
C. ☐ Suspected Pest of Regulatory Concern *(Explain in* REMARKS*)*
D. ☐ Stored Product Pest
E. ☐ Livestock, Domestic Animal Pest
F. ☐ Possible Immigrant *(Explain in* REMARKS*)*
G. ☐ Survey *(Explain in* REMARKS*)*
H. ☐ Other *(Explain in* REMARKS*)*

9. IF PROMPT OR URGENT IDENTIFICATION IS REQUESTED, PLEASE PROVIDE A BRIEF EXPLANATION UNDER "REMARKS".

HOST DATA

10. HOST INFORMATION
NAME OF HOST *(Scientific name when possible)*

11. QUANTITY OF HOST
NUMBER OF ACRES/PLANTS
PLANTS AFFECTED *(Insert figure and indicate* ☐ Number ☐ Percent*)*:

12. PLANT DISTRIBUTION
☐ LIMITED
☐ SCATTERED
☐ WIDESPREAD

13. PLANT PARTS AFFECTED
☐ Leaves, Upper Surface ☐ Trunk/Bark ☐ Bulbs, Tubers, Corms ☐ Seeds
☐ Leaves, Lower Surface ☐ Branches ☐ Buds
☐ Petiole ☐ Growing Tips ☐ Flowers
☐ Stem ☐ Roots ☐ Fruits or Nuts

PEST DATA

14. PEST DISTRIBUTION
☐ FEW
☐ COMMON
☐ ABUNDANT
☐ EXTREME

15. ☐ INSECTS ☐ NEMATODES ☐ MOLLUSKS

NUMBER SUBMITTED	LARVAE	PUPAE	ADULTS	CAST SKINS	EGGS	NYMPHS	JUVS.	CYSTS
ALIVE								
DEAD								

16. SAMPLING METHOD
17. TYPE OF TRAP AND LURE
18. TRAP NUMBER

19. PLANT PATHOLOGY – PLANT SYMPTOMS *("X" one and describe symptoms)*
☐ ISOLATED ☐ GENERAL

20. WEED DENSITY
☐ FEW ☐ SPOTTY ☐ GENERAL

21. WEED GROWTH STAGE
☐ SEEDLING ☐ VEGETATIVE ☐ FLOWERING/FRUITING ☐ MATURE

22. REMARKS

23. TENTATIVE DETERMINATION

24. DETERMINATION AND NOTES *(Not for Field Use)*

SIGNATURE
DATE

FOR IIBIII USE
DATE RECEIVED
NO.
LABEL
SORTED
PREPARED
DATE ACCEPTED
RR

PPQ FORM 391 *Previous editions are obsolete.*
(AUG 02)

This is a 6-Part form. Copies must be disseminated as follows:
☐ PART 1 – PPQ ☐ PART 2 – RETURN TO SUBMITTER AFTER IDENTIFICATION ☐ PART 3 – IIBIII OR FINAL IDENTIFIER
☐ PART 4 – INTERMEDIATE IDENTIFIER ☐ PART 5 – INTERMEDIATE IDENTIFIER ☐ PART 6 – RETAINED BY SUBMITTER

FIGURE B-1 Example of PPQ 391 Specimens For Determination [side 1]

OMB Information
According to the Paperwork Reduction Act of 1995, no persons are required to respond to a collection of information unless it displays a valid OMB control number. The valid OMB control number for this information collection is 0579-0010. The time required to complete this information collection is estimated to average .25 hours per response, including the time for reviewing instructions, searching existing data sources, gathering and maintaining the data needed, and completing and reviewing the collection of information.

Instructions
Use PPQ Form 391, Specimens for Determination, for domestic collections (warehouse inspections, local and individual collecting, special survey programs, export certification).

BLOCK	INSTRUCTIONS
1	1. Assign a number for each collection beginning the year, followed by the collector's initials and collector's number **EXAMPLE** In 2001, Brian K. Long collected his first specimen for determination of the year. His first collection number is 01-BLK-001 2. Enter the collection number
2	Enter date
3	Check block to indicate Agency submitting specimens for identification
4	Enter name of sender
5	Enter type of property specimen obtained from (farm, nursery, feedmill, etc.)
6	Enter address
7	Enter name and address of property owner
8A-8L	Check all appropriate blocks
9	Leave Blank
10	Enter scientific name of host, if possible
11	Enter quantity of host and plants affected
12	Check block to indicate distribution of plant
13	Check appropriate blocks to indicate plant parts affected
14	Check block to indicate pest distribution
15	- Check appropriate block to indicate type of specimen - Enter number specimens submitted under appropriate column
16	Enter sampling method
17	Enter type of trap and lure
18	Enter trap number
19	Enter X in block to indicate isolated or general plant symptoms
20	Enter X in appropriate block for weed density
21	Enter X in appropriate block for weed growth stage
22	Provide a brief explanation if Prompt or URGENT identification is requested
23	Enter a tentative determination if you made one
24	Leave blank

Distribution of PPQ Form 391
Distribute PPQ Form 391 as follows:
1. Send Original along with the sample to your Area Identifier.
2. Retain and file a copy for your records.

FIGURE B-2 Example of PPQ 391 Specimens for Determination [side 2]

C

False Codling Moth

Appendix C

Hosts

Contents

Preferred Hosts

Preferred hosts are plants that are known to support the development of false codling moth (**Table C-1**).

TABLE C-1 Preferred Hosts of False Codling Moth [USDA (1983)]

Common name	Scientific Name
Corn	*Zea mays*
Cotton	*Gossypium* spp.
Guava, common	*Sodium guajava*
Macadamia[1]	*Macadamia* spp.
Mandarin orange	*Citrus reticulata*
Oak	*Quercus* spp.
Okra	*Abelmoschus esculentus*
Oranges	*Citrus* spp.
Orange, Temple	*Citrus reticulata x Citrus sinensis*
Peach	*Prunus persica*
Pepper	*Capsicum* spp.
Sorghum	*Sorghum vulgare*
Tangelo	*Citrus paradisi x Citrus reticulata*

1 Source: van den Berg (1995)

Secondary Hosts

Secondary hosts are plants that can support the development of false codling moth; however, the literature has not disclosed all the conditions under which the host-plant relationship occurs (**Table C-2**).

TABLE C-2 Secondary Hosts of False Codling Moth [USDA (1983)]

Common name	Scientific name
Apricot	*Prunus armeniaca*
Avocado	*Persea americana*
Banana	*Musa paradisiaca var. sapientum*
Butterseed	*Butryospermum parkii*
Castor bean	*Ricinus communis*
Cherimoya	*Annona cherimola*
Cherries (All)	*Prunus* spp.
Chrysophyllum magalis-montanum	Chrysophyllum magalis-montanum
Coffee	*Coffea spp.*
Cowpea	*Vigna unguiculata*
Custard apple	*Annona reticulata*
Eggplant	*Solanum melongena*
Elephant grass	*Pennisetum purpureum*
Flowering Maple	*Abutilon* spp.
Grape[1]	*Vitis vinifera*
Grapefruit	*Citrus paradisi*
Ground Cherry	*Physalis* spp.
Hibiscus	*Hibiscus* spp.
Hottentot kafir bean tree	*Schotia speciosa*
Husk tomato	*Physalis ixocarpa*
Jute	*Sida* spp.
Kafir marvolanut	*Sclerocarya caffra*
Kafir plum	*Harpephyllym cattrum*
Large fruited bushwillow	*Combretum zeyheri*
Kapok ceiba	*Ceiba pentandra*
Lemon	*Citrus limon*
Lima bean	*Phaseolus lunatus*
Lime	*Citrus aurantiifolia*
Litchi, Litchee	*Litchi chinensis*
Loquat	*Eriobotrya japonica*
Mallow	*Abutilon* spp.
Mango	*Mangifera indica*
Miraculous berry	*Synsepalum dulciticum*
Oak	*Quercus* spp.
Olives	*Olea europaea*

TABLE C-2 Secondary Hosts of False Codling Moth [USDA (1983)]

Common name	Scientific name
Outeniqua yellowwood	*Podocarpus falcata*
Persimmon	*Diospyros* spp.
Plum	*Prunus* spp.
Pomegranate	*Punica granatum*
Pond Apple	*Annona glabra*
Prune	*Prunus domestica*
Pseudolachnostylis maprounaefolia	*Pseudolachnostylis maprounaefolia*
Red bushwillow	*Combretum apiculatum*
Rose apple	*Syzygium jambos*
Royena pallens	*Royena pallens*
Sidas	*Sida* spp.
Soursop	*Annona muricata*
Spanish bayonet	*Yucca alofolia*
Spanish dagger	*Yucca gloriosa*
Star apple	*Chrysophyllum cainito*
Sugar apple	*Annona squamosa*
Sweetsop	*Annona* spp.
Tomato	*Lyvopersicon esculentum*
Tree tomato	*Cyphomandra betacea*
Triumfetta, Burrbark	*Triumfetta* spp.
Vanguria	*Vangueria infausta*
Walnut	*Juglans* spp.
Ximenia	*Ximenia caffra*
Yucca	*Yucca* spp.
Zizyphus	*Zizyphus mucronata*

1 Source: Opatowski (2005); Netshifhefhe (2005)

D

False Codling Moth

Appendix D

Taxonomy

Contents

Taxonomy of False Codling Moth

The following appendix was reproduced from Venette et al. (2003).

Appendix C. Taxonomy of *Thaumatotibia leucotreta* (Meyrick) and related Tortricidae (prepared by M. DaCosta).

The species name *leucotreta* was removed from the genus *Cryptophlebia* and placed in *Thaumatotibia* by Komai (1999). The genus *Thaumatotibia* Zacher (1915) was not placed in a family when established, and was instead placed as a synonym of *Cryptophlebia* Walsingham (1899) (Nye and Fletcher 1991). Species of *Thaumatotibia* and *Cryptophlebia* are similar to each other externally and the two genera are related to each other. Komai (1999) lists the following characters common to both genera:

1) forewing broad (male broader than female), with a blackish triangular pretornal patch and with an accessory cell of chorda small or absent (chorda coincident with the margin of the discal cell)
2) hindwing with a short discal cell, especially in males
3) eighth tergite (and sometimes preceeding tergites) with a small patch of long scales
4) valva with a patch of very long, curled scales on the outer surface of the cucullus
5) tenth abdominal segment of pupa with a pair of strong spines along anal rise.

Thaumatotibia can be distinguished from *Cryptophlebia* on the basis of the following characters:

1) eighth tergite in male with a broadly sclerotized plate with convex posterior margin and laterally produced into curved points
2) sterigma indicated by an ovate or rectangular sclerite connecting posteriorly with a pair of ovate granulations with modified scales
3) corpus bursa with granular patch at juncture of ductus bursae.

The species *leucotreta* was placed in one of two species groups in *Thaumatotibia* by Komai (1999), the *leucotreta*-group, which is distinguished from the other species group, the *chaomorpha*-group, by (1) the presence of tufts of modified scales on the inner side of the hindtibia (usually), (2) an enlarged inner apical spur and (3) "normal" juxta. According to Bradley et al. (1979), male *T. leucotreta* are distinguished from other species by the specialized hindwing, which is slightly reduced and has a circular pocket of fine hair-like black scales overlaid with broad weakly shining whitish scales in the anal angle, and its heavily tufted hind tibia.

Descriptions of the external morphology as well as the genitalia of *Thaumatotibia leucotreta* (Meyrick) are provided.

<u>Synonyms</u> (from Bradley et al. 1979, Nye and Fletcher 1991, Komai 1999)

At the generic level:
Thaumatotibia Zacher,1915: 529-Heppner, 1980: 34 (as synonym of *Cryptophlebia*).
Type species: *Thaumatotibia roerigii* Zacher, 1915 [=*Argyroploce leucotreta* Meyrick, 1913] by monotypy

- *Argyroploce* Hübner, [1825]

- *Olethreutes* Hübner, 1822, Syst. -alphab. Verz.: 58-67, 69, 72. Type species: *Phalaena arcuella* Clerck, 1759, Icon. Insect. Rariorum 1: pl 10 fig. 8, by subsequent designation by Walsingham, 1895, Trans. Ent. Soc. Lond. 1895: 518.
- *Metriophlebia* Diakonoff, 1969: 89. –Razowski, 1977: 259. –Clarke, 1986: 162 (as synonym of *Cryptophlebia*), syn. n. Type species: *Eucosoma chaomorpha* Meyrick, 1929, by monotypy

At the species level:
leucotreta (Meyrick) 1913. Ann. Transv. Mus. 3: 267-336.

- *roerigii* Zacher, 1915: 529 Beiträge zur Kenntnis der westafrikanischen Planzenschädlinge.-Tropenpflanzer 18: 504-534.

Diagnosis of *Thaumatotibia leucotreta*:
[Description from Komai (1999).] Small to medium-sized, grayish-brown to dark brown/black moths with broad forewings (forewing index: 0.41-0.44mm in males, and 0.38-0.42mm in females) with a blackish triangular pretornal patch. Externally species of *Thaumatotibia* are similar to species of *Cryptophlebia*. Wing venation of *Thaumatotibia* is characterized by a small accessory cell delineated by the chorda from between R2 and R3 (closer to R3) to R4 or from between R1 and R2 (very close to R2) to between R5 and R5, or the absence of accessory cell (the chorda coincident with the margin of the discal cell), and by a short discal cell in the hindwing, especially in the male (0.42-0.43x length of the wing). Eigth tergum in male with a broadly sclerotized plate with convex posterior margin and laterally produced into curved points, with paired patches of long mane-like scales, but without a pair of long filiform scale tufts from shallow membranous pockets on each side of eighth tergum as in *Cryptophlebia*. Male genitalia are characteristic in the large, ovate valva (the outer surface with a patch of very long, curled scales, which is shared with *Cryptophlebia*), in the sacculus often with teeth distally, and in the juxta sometimes producing caudally a pair of denticulate, ovate lobes (the *chaomorpha*-group). The female genitalia is characterized by the sterigma indicated by an ovate or rectangular sclerite, connecting posteriorly with a pair of ovate granulations with modified scales, by the corpus bursae with a ring of granulation at the juncture of the ductus bursae, and sometimes a diverticulum ventrally or laterally.

Description:

Head: [Description from Komai (1999).] As in Figure C1. Frons with very dense, erect and moderately long scales. Antenna filiform, less than 2/3 length of forewing. Labial palps long and wavy; second segment widened distally, but scales appressed and rather short; terminal segment extends forward horizaontally, about 1/3 length of second, slender, with appressed scales, apex blunt.

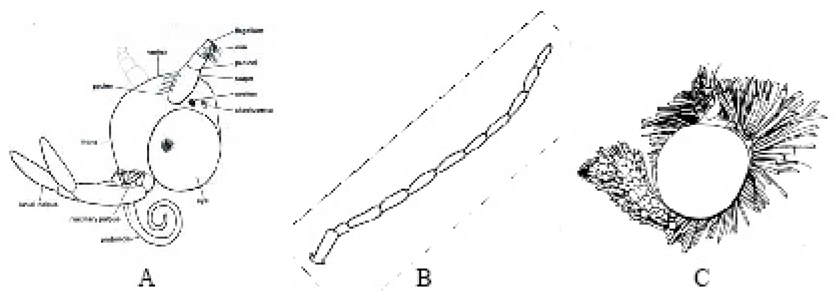


Figure C1. Lateral views of head. A-Ventrolateral view of general moth head [Reproduced from Robinson et al. (1994)]. B-Filiform antenna [Reproduced from Borror et al. (1989)] C-Lateral view head of *Thaumatotibia hemitoma* (Diakonoff)-♂. [Reproduced from Komai (Komai 1999)]

Thorax: [Description from Komai (1999)] Posterior crest present. Hind tibia (as in Fig. C2) with modified scales on inner side, the inner apical spur enlarged with a batch of scales, the bases of which have a layer of secreting cells.

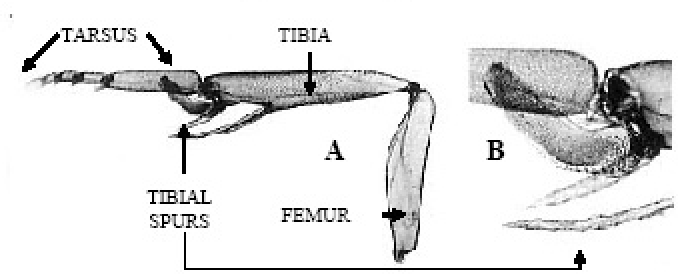
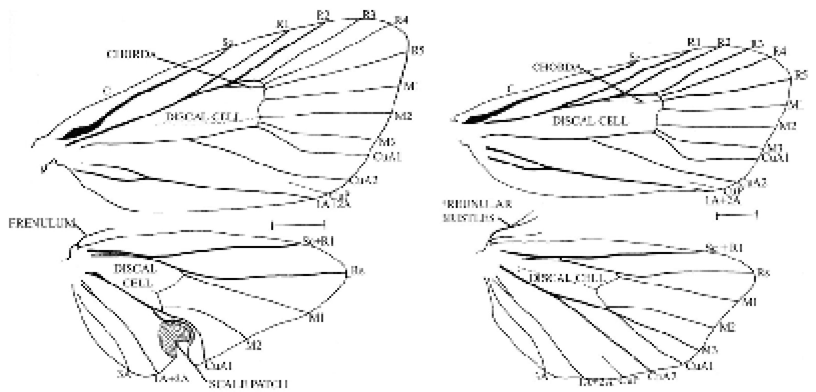


Figure C2. A-Hindtibia-modified scales and apical spur removed. B-Detail of spurs. [Reproduced from Komai (1999)]

Wings: [Description from Bradley et al. (1979).] Forewing pattern a mixture of bluish-gray, brown, black, and rust colored red-brown markings, the most conspicuous is the blackish triangular pre-tornal marking and the crescent-shaped marking above it, and a minute white spot in the discal area.

Venation: As in Fig C3. There is a scent organ on the distal 2/3 of CuA2 on upper side. Its presence is indicated by concavity on wing membrane bounded with thickened ridges bearing the secreting cells [Zagatii and Castel quoted in Komai (1999)].



A B

Figure C3. Venation of *Thaumatotibia leucotreta* (Meyrick), A-male, B-Female. Veins: A-anal; C-Costa, Cu-Cubitus (CuA1-1st anterior cubitus; CuA2-2nd anterior cubitus; CuP-posterior cubitus); D-discal cell; M-Media, R-Radius, Sc-Subcosta. [Reproduced from Komai (1999)]

Abdomen: [Description from Komai (1999)] As in Fig C4. Second sternite with well developed anterolateral processes and sternal apodemes. Male abdominal scent organs: eighth tergite with a broadly sclerotized plate with convex posterior margin and laterally produced into curved points, densely covered with long scales which are easily removable.

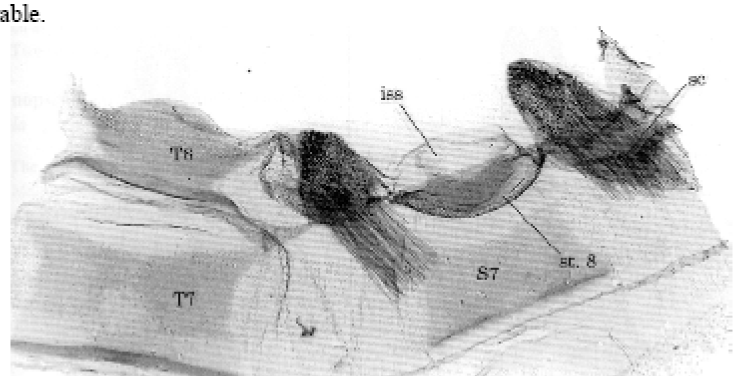


Figure C4. Post abdomen showing: 7th sternite (S7), 8th sternite (st. 8), 7th tergite (T7), 8th tergite (T8), intersegmental ventral sclerite between abdominal segments 8 and 9 (iss), scale-tufts of coremata (sc) [Reproduced from Komai (1999)]

CAPS PRA: *Thaumatotibia leucotreta* 22

Male genitalia: [Description from Komai (1999)] As in Fig. C5. Tegumen a broad band, rounded apically; Aedeagus bulbous basally, narrowed at basal 1/4 to 1/3 and upcurved distally; vesica with series of fine cornuti. Juxta producing caudally a pair of denticuate, "normal" lobes. [Terminology follows Klots (1970).]

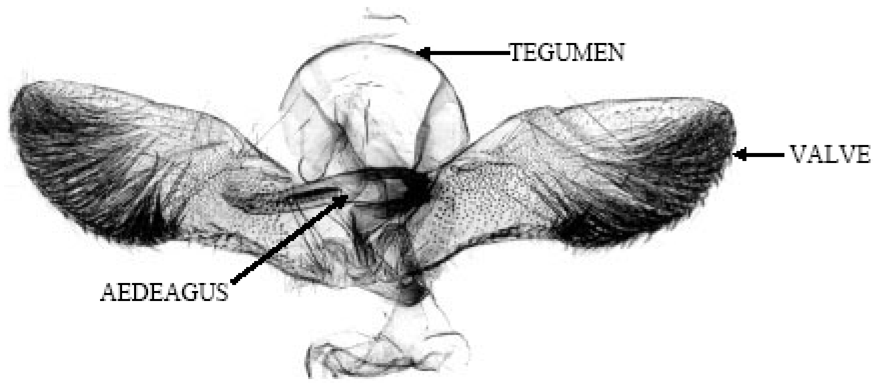


Figure C5. Ventral view male genitalia [Reproduced from Komai (1999)]

Female genitalia: [Description from Komai (1999)] As in Fig. C6. Papillae anales "moderate". Anterior apophyses longer than posterior apophyses. Sterigma an ovate, or rectangular raised sclerite, connecting posteriorly with a pair of ovate granulations with modified scales. Ductus bursa long and narrow, ductus seminalis arising laterally, from posterior 1/4-1/5 of ductus bursa; bulla seminalis present; corpus bursae ovate, with a ring of granulation at juncture of ductus bursa with diverticulum laterally, with two large, curved, blade-shaped signa. Seventh sternite trapezoidal, posterior margin with shallow or deep excavation.

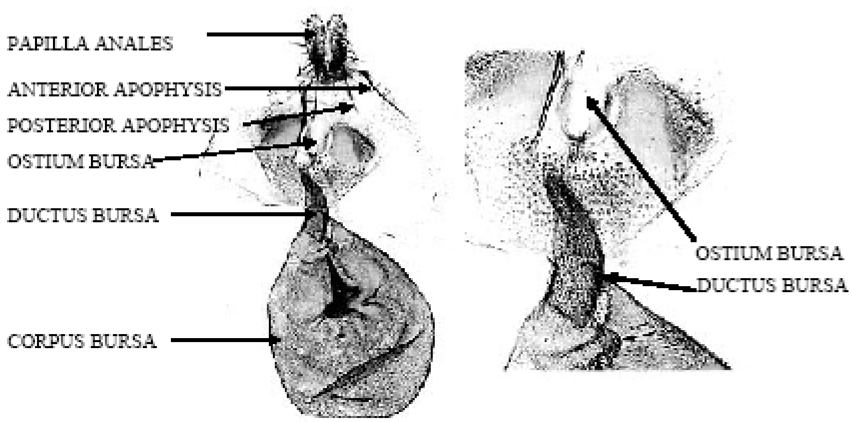


Figure C6. Genitalia *Thaumatotibia leucotreta* (Meyrick). Left-Entire genital apparatus, Right-ostium bursa and posterior part of bursa copulatrix [Reproduced from Komai (1999)]

CAPS PRA: *Thaumatotibia leucotreta* 23

Larva: [Description from Komai (1999).] Body length of mature larva 15mm. Head yellowish-brown. Body orange or pink in final instar. Pinacula large, darker than body color. Spiracle on A8 near the posterior margin. Prolegs with 31-40 crochets arranged in a biordinal circle. Anal fork present. Chaetotaxy As in Fig. C7: SD1 and SD2 on same pinaculum on A9; SV group on A1-A6 trisetose, on A7 and A8 bisetose, A9 unisetose; L group trisetose on A9.

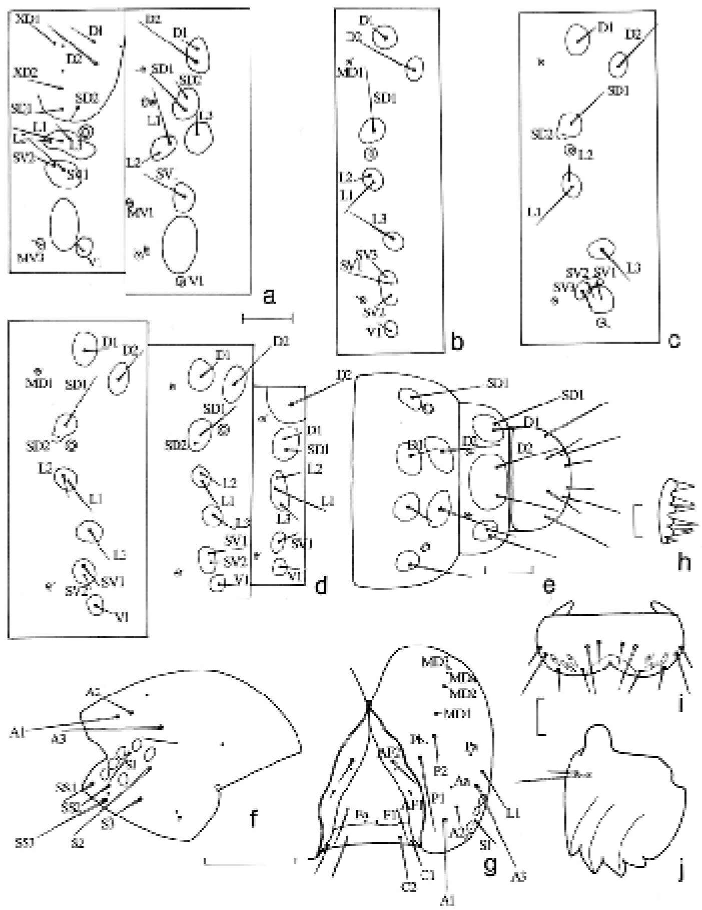


Figure C7. Setal map of *Thaumatotibia leucotreta*; drawing scale a-g: 0.5mm, h-j: 0.1 mm [Reproduced from Komai (1999)]

Pupa**:** [Description from Komai (1999)] As in Fig. C8. Body length 6-10mm. Body pale yellowish-brown. Similar to *Cryptophlebia*. Spiracles transversely ovate. A2-A7 with two rows of dorsal spines; A8-A10 with one row of strong spines, in male A8 with two rows of dorsal spines; A10 with a pair of strong spines along anal rise, without hooked setae except two pairs along anal rise.

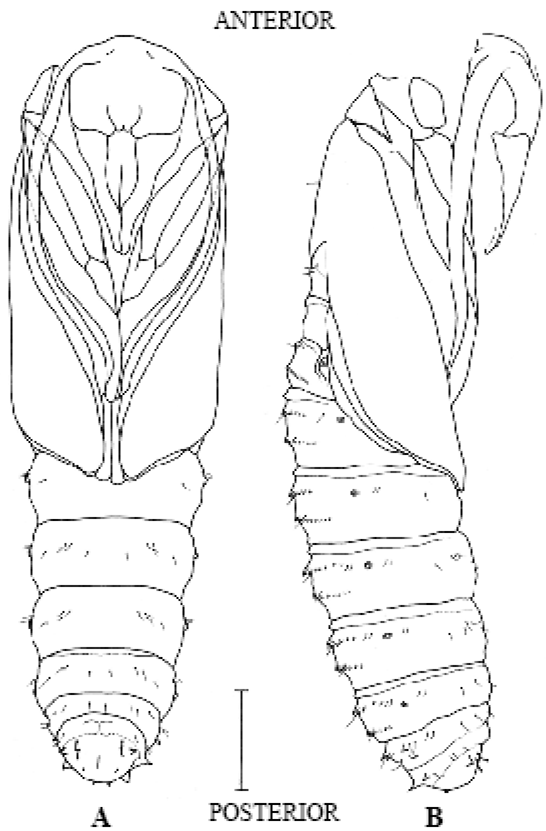


Figure C8. Pupa of *Thaumatotibia sp*. A-Ventral view, B-Lateral view (scale = 1 mm) [Reproduced from Komai (1999)]

<u>Similar species</u>: *Cydia pomonella* (Linnaeus)-the codling moth [occurs in the US]

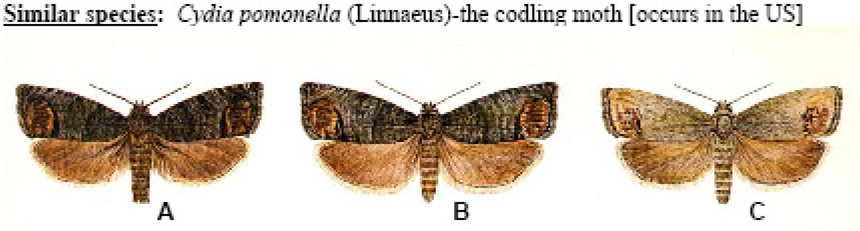


Figure C9. Dorsal views of *Cydia pomonella* (Linnaeus) A-male, B & C-female (to illustrate degree of morphological variation within a sex).
[Reproduced from Bradley et al. (1979)]

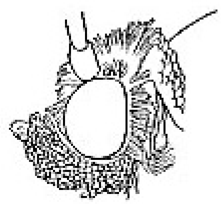


Figure. C10. Lateral view of head of *Cydia pomonella* (Linneaus)
[Reproduced from Bradley et al. (1979)]

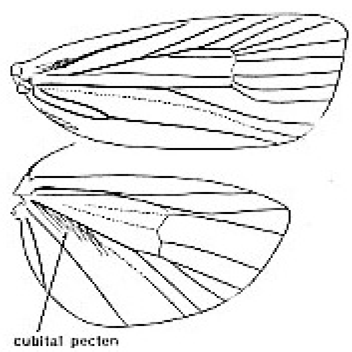


Figure C11. Venation of *Cydia pomonella*
[Reproduced from Bradley et al. (1979)]

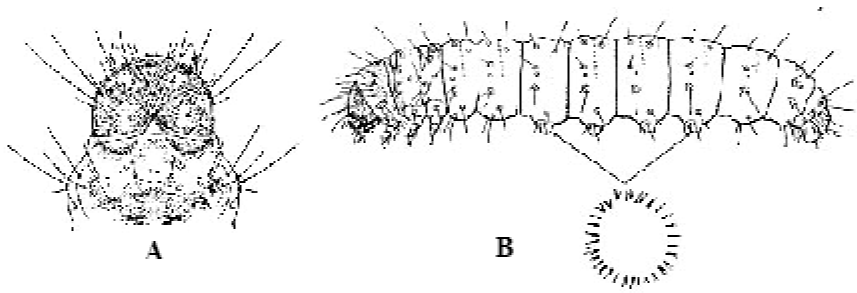


Figure C12. Larva of *Cydia pomonella* (Linneaus). A-Dorsal view of head, B-lateral view of body; pattern of crochets [Reproduced from Bradley et al. (1979)]

Species Similar to False Codling Moth

***Cydia* spp.**

Cydia pomonella (Linnaeus), the codling moth, occurs in the United States and is one of the species most likely to be confused with the false codling moth (Bradley et al. 1979). *Cydia toreuta, C. cupressana, C. injective* and *C. miscitata* are indigenous to the United States and have a similar appearance (**Figure D-3**, **Figure D-4** and **Figure D-5**).

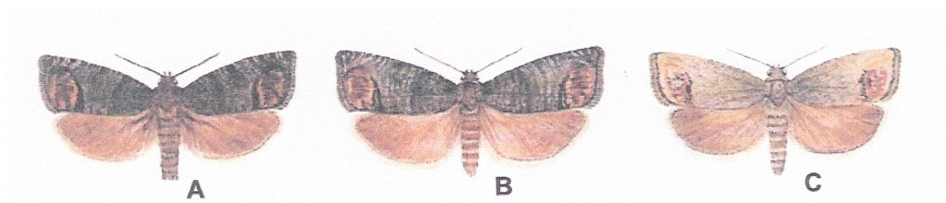


FIGURE D-3 Dorsal Views of *Cydia pomenella;* A-male, B- and C-female [Image courtesy of Bradley et al. (1979)]

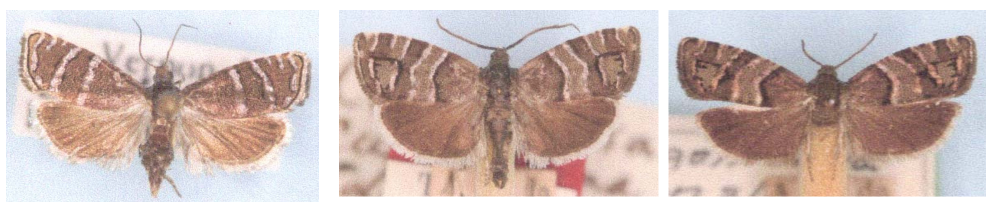


FIGURE D-4 Dorsal Views of (left) *Cydia toreuta*; (middle) *Cydia cupressana* male; (right) *Cydia cupressana* female [Images courtesy of T. Gilligan, Ohio State University]

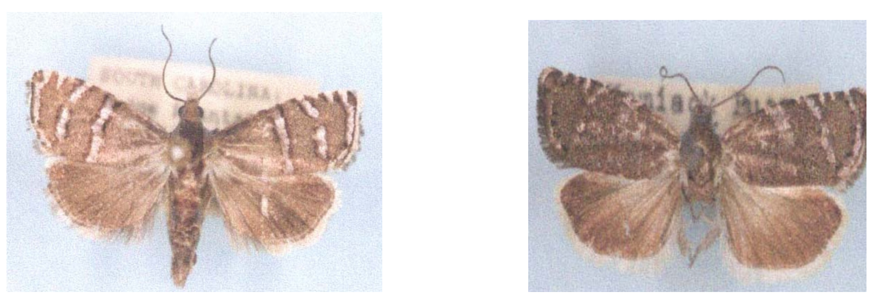


FIGURE D-5 Dorsal views of (left) *Cydia injective* and (right) *Cydia miscitata*

Mussidia nigrevenella

In West Africa, the false codling moth is often found in conjunction with the pyralid moth [*Mussidia nigrevenella* (Moyal & Tran. 1989; Silvie 1990)] (**Figure D-6**).

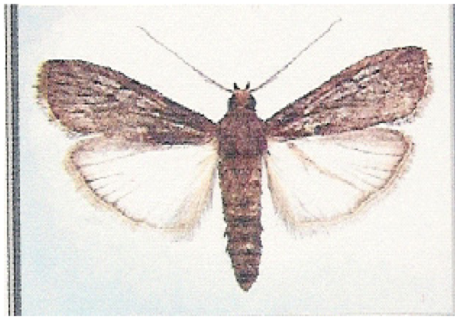


FIGURE D-6 Dorsal View of *Mussidia nigrevenella* [Image courtesy of CPC (2004)]

Cryptophlebia peltastica

The litchi moth (*Cryptophlebia peltastica*) is similar to false codling moth. However, the male can be distinguished by a subtriangular or Y-shaped T8 with a pair of tufts of filliform scales from membranous pockets on each side (Newton 1998) (**Figure D-7**).

FIGURE D-7 Dorsal View of *Cryptophlebia peltastica* [Image courtesy of CPC (2004)]

Thaumatotibia spp.

The macadamia nut borer (*Thaumatotibia batrachopa*) is similar to false codling moth and occurs in Africa. *Thaumatotibia chaomorpha* is similar (**Figure D-8**).

FIGURE D-8 Dorsal View of *Thaumatotibia chaomorpha*
[Image courtesy of T. Gilligan, Ohio State University]

Gymnandrosoma aurantianum

FIGURE D-9 Dorsal View of *Gymnandrosoma aurantianum*
[Image courtesy of T. Gilligan, Ohio State University]

Cryptospasma bipenicilla

FIGURE D-10 Dorsal View of *Cryptospasma bipenicilla* [Image courtesy of T. Gilligan, Ohio State University]

***Ecdytolopha* spp.**

Ecdytolopha punctidiscana are often caught in pheromone traps set for false codling moth (**Figure D-11**).

FIGURE D-11 Dorsal View of (left) *Ecdytolopha mana* and (right) *Ecdytolopha punctidiscana* [Images courtesy of T. Gilligan, Ohio State University]

Tools For Identifying Larvae of Leafrollers

DRAFT
April 2004

TOOLS FOR IDENTIFYING THE LARVAE OF LEAFROLLERS

(LEPIDOPTERA: TORTRICIDAE)

FREQUENTLY INTERCEPTED AT U.S. PORTS OF ENTRY

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Leafrollers (the family Tortricidae) are among the most commonly intercepted microlepidoptera larvae at U.S. ports-of-entry. They frequently infest fruit, nuts, seeds, flowers, leaves, and other parts of vascular plants, and they are encountered in personal baggage as well as in imported commodities. Unfortunately, the most widely available and useful key for APHIS/PPQ port inspectors/identifiers is the portion of Weisman's (1986) Lepidoptera key dedicated to this family. That key includes only 10 species, and the nomenclature is somewhat outdated.

Comprehensive treatments of tortricid larvae were developed by Swatschek (1958) for the European fauna and by MacKay (1959, 1962) for the Nearctic fauna. More recently, Brown (1983) summarized our knowledge of the North American tortricid larvae, providing keys to pest species arranged by host plant groupings (i.e., larvae on conifers, larvae on rosaceous plants, larvae on legumes). Horak and Brown (1991) provided general, brief descriptions of tortricid larvae, focusing on differences at the tribal level. While all of these contributions provide excellent information on the family and are important references, none is particularly useful for identifying the tortricid "fauna" that is encountered by inspectors at our ports.

The purpose of this paper is to provide three dichotomous keys: (1) a much expanded and updated version of the tortricid portion of the Weisman's key; (2) a key to Olethreutinae intercepted most frequently on *Castanea*; and (3) a refined translation of Swatschek's (1958) key to the genera of the tribe Archipini in Europe. The latter group represents the most frequently intercepted Tortricinae at U.S. ports. In addition, a table of the most common plant-tortricid associations encountered at ports is provided.

Before attempting to use the keys and the table, it is important to make sure that you've correctly identified the larva to family. Tortricids share with many microlepidoptera the following: trisetose L group on the prothorax; L1 and L2 on a shared pinaculum on A2-A7; and crochets in a circle. Larvae of tortricids can be distinguished from other microlepidoptera by a combination of the following characters: (1) A8 with SD1 almost always anterad (sometimes slightly dorso- or ventroanterad) of the spiracle; (2) A9 with D2s almost always on a common dorsal pinaculum on A9; (3) A9 with D1 and SD1 present and in two distinct configurations: on the same or different pinacula; and (4) A10 frequently with an anal fork (lost secondarily in many Olethreutinae).

Deviations from the above can be found in the following. (1) Although SD1 is almost always anterad of spiracle on A8, it is anterodorsad in a few genera and almost precisely dorsad in several North American species of *Eucosma* on *Pinus*. (2) The D2s are almost always on a common dorsal pinaculum on A9; however, they are on separate pinacula in several species of *Cydia* in which the larva is extremely pale and the pinacula nearly without pigmentation. (3) In most Olethreutinae D1 and SD1 are on the same pinaculum, and in most Tortricinae they are on separate pinacula. (4) Microlepidoptera larvae with an anal fork are almost always Tortricidae (although the structure is lost in many Olethreutinae). One genus of Gelechiidae (i.e., *Anarsia*) has an anal fork that can be confused with that of a tortricid. This genus has significant gaps in the crochets on the prolegs (including the anal proleg) so that they do not form in a complete circle.

Because larvae are highly variable, many specimens will not key convincingly to a final couplet. In these situations, either follow the "best fit" or return to the previous "convincing" couplet. Some larvae are asymmetrical from side-to-side, particularly in the number of setae. In these situations, the "higher number" is almost always the correct value. That is, on a larva that has a bisetose L-group on the right side of A9 and a trisetose L-group on the left side, consider the trisetose condition as "correct."

Key to Tortricid Larvae

Frequently Intercepted at U.S. Ports-of-entry

1. D1 and SD1 on the same pinaculum on A9 (Fig. 1); anal fork present (Fig. 3) or absent 2

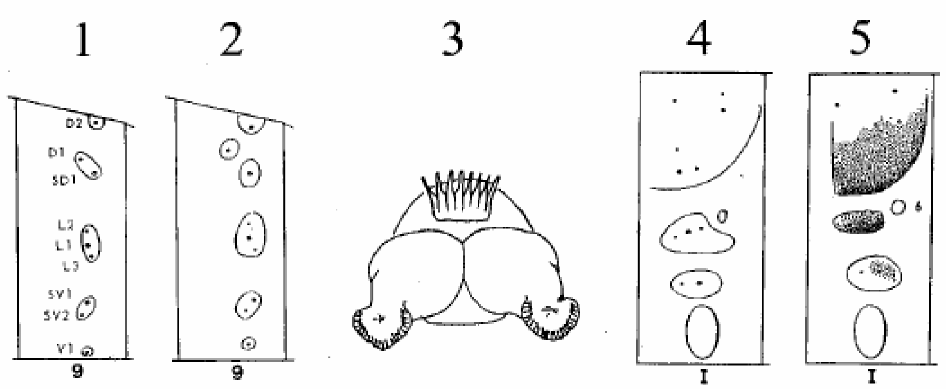
1.' D1 and SD1 on separate pinacula on A9 (Fig. 2); anal fork almost always present . . . . Group 1 ("Tortricinae" Type)

2. L pinaculum on T1 enlarged, extending beneath and beyond (posterad of) spiracle (Fig. 4) . . . . Group 2 ("*Cryptophlebia*" Type)

2.' L pinaculum on T1 simple or variously modified, but not extending beneath spiracle (Fig. 5) 3

3. Anal fork absent . . . . Group 3 ("*Cydia*" type)

3'. Anal fork present . . . . Group 4 ("Olethreutinae" type)



Group 2. *Cryptophlebia* Type

1. Anal fork present 2

1.' Anal fork absent 4

2. L-group on A9 bisetose (both setae on same pinaculum); pinacula usually moderate to large; Vs on A9 much further apart than those on A8 (mostly New World) 3

2.' L-group on A9 usually trisetose (all setae usually on same pinaulum); pinacula moderate; Vs on A9 slightly further apart than those on A8 (mostly Africa) . . . . *Thaumatotibia leucotreta* (Meyrick) complex

3. SV-group usually 2:2:2:2:1; pinacula large; on *Capsicum* (Mexico, Central America) . . . . *Lorita scarificata* (Meyrick)

3.' SV group 3:3:2:2:2(1); pinacula moderate to large; on *Opuntia*, *Pithecellobium*, Asteraceae . . . . many Cochylini

4. At least one (usually more) dorsal pinacula with small incision, notch, or clear spot; Vs on A9 usually 1.5-2.0 times as far apart as Vs on A8 6

4.' Dorsal pinacula usually without small incision, notch, or clear spot; Vs on A9 variable from 1.0-2.0 as far apart as Vs on A8 5

5. SV-group on A1,2,7,8,9 usually 3:3:3:2:2; L-group on A9 trisetose, all setae usually on same pinaculum; abdominal prolegs with 45-48 crochets; on litchi, longan, macadamia, and others (Asia, Hawaii) . . . . *Cryptophlebia ombrodelta* (Lower)

5.' SV-group on A1,2,7,8,9 usually 3:3:2:2(1):1; L-group on A9 uni-, bi- or trisetose; abdominal prolegs with 20-30 crochets, on legumes (South and Central America) *Cydia fabivora* (Meyrick)

6. SV-group usually 3:3:3(2):2:1; L-group on A9 usually trisetose; abdominal prolegs with 50-60

crochets (South and Central America, Caribbean) . . . . *Gymnandrosoma aurantianum* (Lima)

6.' SV-group usually 3:3:3:1:1; L-group on A9 usually bisetose (Asia) . . . . *Cryptophlebia illepida* (Butler)

Group 3. *Cydia* Type

1. D1s, D2s, and SD1s on A9 all on same enlarged (ill-defined) pinaculum (integument conspicuously spiny; crochets on abdominal prolegs 16-19); on *Annona* and *Mammae* (Mexico, Central America, Caribbean) . . . . *Talponia batesi* Heinrich

1.' D2s on pinacula separate from D1 and SD2 (D2 shared pinaculum sometimes very weakly developed) 2

2. All three L setae of A9 usually on same pinaculum 3

2.' L3 absent or on separate pinaculum on A9 5

3. On *Castanea* . . . . go to key of *Castanea*-feeding Olethreutinae

3.' On various other hosts 4

4. South America (Brazil, Argentina, and Chile) on *Aracauria* (body pinkish with medium large, pale brown pinacula; prothoracic shield light brown, anal shield dark brown; L pinaculum of T1 rather large, sometimes notched; Ls on A9 sometimes all on the same pinacula; SV-group 3:3:2(3):1(2):1; crochets 34-37) . . . . *Cydia araucariae* Pastrana

4.' Mexico, on various hosts (including *Carya*, *Prunus*, *Malus*) . . . . *Cydia* sp.

4'' Asia, on *Saccharum officinarum* . . . . *Tetramoera schistaceana* (Snellen)

5. D2s on A9 always on same pinaculum; SV group usually 3:3:2:2:1; distinct pattern on anal and prothoracic shields; 30-35 crochets (cosmopolitan) . . . . . . *Cydia pomonella* (Linnaeus) (in part)