NEWS OF THE LEPIDOPTERISTS' SOCIETY

Volume 58, Number 4 Winter 2016

THE LEPIDOPTERISTS' SOCIETY EST. 1947

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The Lepidopterists' Society is a non-profit educational and scientific organization. The object of the Society, which was formed in May 1947 and formally constituted in December 1950, is "to promote internationally the science of lepidopterology in all its branches; to further the scientifically sound and progressive study of Lepidoptera, to issue periodicals and other publications on Lepidoptera; to facilitate the exchange of specimens and ideas by both the professional worker and the amateur in the field; to compile and distribute information to other organizations and individuals for purposes of education and conservation and appreciation of Lepidoptera; and to secure cooperation in all measures" directed towards these aims. (Article II, Constitution of The Lepidopterists' Society.)

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Mesosemia pacifica, Montezuma Road, departmento de Risaralda, Colombia; top: male, Feb. 27, 2013 (photo by Kim Garwood), bottom: female, May 27, 2014 (photo by David Geale); see article, next page (159)

Digital Collecting:

Montezuma, butterfly heaven in Colombia

Kim Garwood

721 N Bentsen Palm Dr #40, Mission, TX 78572 **kimgrwd@sbcglobal.net**

In the western cordillera of Colombia is a fabulous place for butterflies. My friends and I have been photographing butterflies all over Colombia for the last nine years, and Montezuma, at the border of Parque Nacional Natural Tatamá in the departmento de Risaralda about two hours west of Pereira, has the richest biodiversity that we have found. We're up to 722 species so far, but every trip we keep adding new species. You could stay here for years and keep finding new things.

Colombia has an extremely complicated biogeography, as the Andes split into three chains of mountains in southern Colombia. This gives six different slopes, instead of just east/west slopes as in Ecuador, Peru and Bolivia. In between the three chains are the two major valleys, the Cauca Valley to the west, which is narrower, and the Magdalena Valley to the east, much wider and more developed for agriculture. I would expect there to be locations with higher numbers of species on the east slope of the eastern Cordillera, where you go from cloud forest down to the Amazon lowlands, but I haven't spent as much time there in Colombia as compared to Peru.

The western cordillera has the Choco, the west slope to the Pacific and one of the wettest places in the world, which is a hotbed of endemism. This region has not been studied very well, as historically access has been limited. There are few roads, often very heavy rains, and not much in the way infrastructure and places to stay.

While looking for access to this habitat, my Colombian birding friends found a farmhouse at the end of a dirt road that winds up into the mountains. This has been developed into a comfortable place to stay, and Michelle Tapasco (often called Leo), the woman who owns it, has learned a lot about the care and feeding of international, and Colombian, ecotourists. She's also become a good birder and butterfly photographer. She's mostly vegetarian, and cooks wonderful, healthy meals. The rooms aren't fancy, but they are clean and functional. They have electricity but no internet, and cell phones don't work very well. So you're pretty well off the grid for the duration. But you have power 24/7, so charging batteries and playing with photos in late afternoons/evenings works fine. Michelle's phone is +57 317-684-1034, but she only speaks spanish if you want to call.

My friend David Geale, a Canadian who has been corrupted from leading bird tours five years ago around South America, has become an avid butterfly photographer. He helps organize all my South American photography trips now, plus he sends me all his excellent photos from all his trips. He is a close friend of Michelle, and can easily make reservations for you if you prefer to communicate in english. You can see more of his photos on his site, and find out about his upcoming trips, at **http://mariposabutterflytours.com/**, or at **https://flic.kr/s/aHskpyea2U .**

The farmhouse is at about 1400m, and the dirt road heads up into the mountains from there. You can get up to about 2600m, so the road covers a spectacular range of elevations. You can go back down the dirt road a little ways, and there is a very productive patch of forest about a km below the farm, past some open fields, about the same elevation as the farmhouse. This area appears to be best from April to August, when the sun is further north. The rest of the year it doesn't get sun until after 11am. It's very good for Riodinidae. We've had four species of *Symmachia* here,

Symmachia species: *titiana* (top) and *tricolor* (bottom)
(photos by David Geale)

Thisbe ucubis (left; photo by David Geale), *Eresia levina* (center and right; photos by Kim Garwood)

Argyrogrammana leptographia and *Thisbe ucubis,* as well as a number of Lycaenidae like *Erora aura, Calycopis tamos, Laothus gibberosa, Arzecla taminella, Lathecla latagus,* and *Strymon gabatha.* We also found a very distinctive black and blue crescent there, the endemic *Eresia levina*. Mostly we went up from the farmhouse, where there is great forest all the way to the top, but you would want to spend at least one day at this lower patch. You can get down to the river here too, on a small trail.

Going uphill there is a military base on the top, to protect the communication towers, and the road has been improved quite a bit over the last seven to eight years. The first time I was there, in 2009, we rode horses to the higher elevations, and in some places we rode over landslides. Good thing the horse knew the trail. I just closed my eyes and held on. But now the road has been cleared enough for a large truck to get up to the top to repair the towers, and you can drive up in a 4x4 jeep.

You can spend days walking from the farm, or rent a jeep with a driver and be taken up higher and walk down. Last year my friend Juan visited and drove up in his street car with 4 wheel drive. People in good shape can hike up in several hours, but I rarely get above 1700m on foot. Heading up from the farmhouse where you sleep, you can climb maybe 75-100m in elevation and crest a small hill, then go down to a large bridge across the chasm, about 1350m, maybe two km from the farmhouse. This small hilltop can be good for Riodinidae in the morning. We often see *Sarota, Rhetus dysoni, Euselasia* and others here. It has eastern exposure in the early morning, and if it's sunny, there can be lots of stuff chasing and perching.

Down by the bridge is always a great spot for mudpuddling. We used a lot of bait, mostly rotten fish and male pee. I tell everyone to pee at least once a day at the bridge. The guys often bring a wide mouth bottle so they can collect their pee at night, then have plenty to sprinkle around in the morning. Lots of Nymphalidae fly up and down the canyon, and pee spots on either side of the bridge can be very productive. I usually don't get past there on the first day or two, there's so much to see right there. We have found eight species of *Caligo* so far, and most of them are between the farmhouse and this big bridge, especially if you're out early in the morning.

There is a short, hidden trail that goes down to the water on the far side of the bridge, on your left shortly past the bridge. It can be hard to find, as it gets overgrown. You can slither your way down to the rocky beach, which is a superb spot to put out pee. This is a great spot for big skippers, as they like to fly up and down the stream. We've had a couple of different *Phocides*, both *P. johnsoni* and *P. perillus*, and *Myscelus perissodora* and *Pyrrhopyge papias*, too.

Phocides species: *johnsoni* (top left; photo by Bill Berthet) and *perillus* (bottom left; photo by David Geale); *Pyrrhopyge papias* (right, photos by David Geale)

Also the beautiful *Chalypyge zereda* can be found here. There are lots of *Adelpha* around the bridge. We've had 21 species total at Montezuma, including *lamasi, leuceria, levona, rothschildi, salus, and salmoneus emilia.*

Chalpyge zereda (photos by David Geale)

Adelpha species -- above: *leuceria* (top), *rothschildi* (bottom); right (dorsal and ventral): *lamasi* (top), *salmoneus emilia* (second down), *salus* (second from bottom) and *levona* (bottom). (*lamasi* and *rothschildi* photos by Kim Garwood, the remainder by David Geale)

Anteros species -- top: *allectus* (left, and center with hindwing display), *cruentatus* (right); bottom: *kupris* (left and center), *roratus* (or *chrysoprasta*, right). (photos by David Geale)

Crossing the bridge and going up the other side of the ravine is also excellent. There is another nice Riodinid lek about half way up on this side. We have had several species of *Anteros* here, including *allectus*, *cruentatus*, *kupris*, and *roratus*, or is it *chrysoprasta*? This is one of my favorite genera; they're great to watch bouncing around on the leaves, opening their hindwings to display. They're usually low enough for good photos too. We've also had both *Symmachia titiana* and *tricolor* here (see page 159). David and Martin, one of my Colombian friends, have seen *Prepona werneri* here.

Prepona werneri (photos by Martin Moreno)

Some of the other Nymphalidae genera that show a good variety at Montezuma are *Epiphile*, of which we've seen at least four species: *Epiphile chrysites, epimenes* with the beautiful blue dorsal hindwing, the endemic *E. eriopis*, and *E. neildi*. There's also a nice selection of *Charaxinae*. Three species of *Fountainea* are separated by elevation, so as you go up the mountain the species change. At the farmhouse around 1400m you get the common *Fountainea nessus*, then a bit higher around 1800m you find *F. nobilis pacifica*, then higher yet, around 2400m you get *F. centaurus*. We also found one of my favorites, *Consul panariste*. Plus there are lots of *Memphis*, eight species at least, which can be really difficult to id from a live ventral photo. And on David's last trip

Epiphele species, top to bottom: *chrysitis* (DG), *epimenes* (KG), *eriopus* (KG) and *neildi* (DG) (Photos KG: Kim Garwood; DG: David Geale)

Charaxinae - top three rows are *Fountainea*: *nessus* (top left), *nobilis pacifica* (top right), *centaurus* (above [male and underside] and left [female]) (photos all by Kim Garwood except male *centaurus* by David Geale); bottom: *Consul panariste* (male and underside; photos by David Geale)

Top: *Siderone syntyche;* bottom: *Eunica norica* (Photos by David Geale)

in Jan. 2016 he found the spectacular *Siderone syntyche*, which we had not seen before. Montezuma is also a great spot for the widespread but flighty *Eunica norica*. I've chased this species many times at mid elevation in the Andes, but it's always been shy and not willing to pose for photos. But here we could get them to sit, even for dorsal photos.

On the left as you climb up this side of the ravine there is a small waterfall, where you can get in out of the sun. This is a nice place for lunch. One of the enjoyable aspects of staying at Montezuma is that they bring you a hot lunch in the field. Someone rides up on horseback, finds each person along the trail and drops off their lunch in a nice plastic container, with silverware and napkin, then picks up the empty container on their way back down. Sometimes they ride up on a dirt bike. I haven't had a tasty hot lunch delivered to me in the field at too many other locations. So you can easily spend all day up the trail.

There are four signed areas up the mountain, at different elevations. #1 is the big bridge Rio Claro at 1,350m; #2 is La Clarita, the second bridge at about 1700m; #3 is Cajones at 2,000m; and #4 is Los Choros at 2,400m. Distances are about 2 km to #1, 30 minutes walk; #2 is 5km from the lodge, 1:15 hours walk; #3 is 8km from the lodge, 2:00 hours walk; and #4 is 11km from the lodge, 2:45 hours walk, for the young and fit. The top is 13km, 3:15 hours walk, not for me.

The forest changes as you climb. It is noticeably taller and richer at La Clarita, the second bridge. This is a wonderful spot to be on a sunny morning, as many species are zipping around and displaying over the ravine. It's always difficult for me to march up this far early in the morning, as I'm so distracted by all the goodies lower down, but David has done this a number of times and photographed lots of great stuff. If you have a vehicle it can be quite productive to drive up here for the morning, then work your way back down. The higher you go, of course the more vulnerable you are to the clouds. It often (usually) clouds up in the afternoon, and it can be savagely wet, as a friend describes it. I've seen the dirt road running in at least 6" of water, so you just slosh your way down, more of a stream than a road. But even in the wetter times of the year the mornings are often bright and sunny.

This small bridge at 1700m, La Clarita, is a great spot for Lycaenidae. They're difficult to photograph, as they tend to be displaying out on the leaves over the ravine and too far away. But every now and then one comes close enough for a photo. One of the most exciting is *Paiwarria episcopalis*, a species that is very sexually dimorphic. The females are a bland grey with a white stripe, while the males are brilliant. Another sexually dimorphic hairstreak at this elevation is *Airamanna columbia*. It helps to have lots of

Paiwarra episcopalis (left, LT), *Airamanna columbia* (center [male; LT] and right [female; DG]) (photos: LT - Leo Tabasco; DG - David Geale)

Lucillella aphrodite (left [male; photo by Bob Behrstock] and center [female; photo by David Geale]), *Mesosemia vemania* (right; photo by Leo [Michelle] Tabasco)

time to be at this location. Also some of the spectacular (and uncommon) Riodinidae can be found here, like the endemic *Lucillella aphrodite*, *Mesosemia vemania* and *M. portentosa*, and the more widespread *Euselasia bettina* is very reliable.

As you climb higher up the mountain, the plants and the butterflies change. There is a good stretch about 1800-1900m that is relatively flat with a good eastern exposure that can be quite productive on a sunny morning. I had my only sighting of *Teratophthalma monochroma* (see Garwood, Lep Soc News 55(1): 38 [Spring 2013]) here one morning, where someone had done their business in the bushes and left some toilet paper. Nothing brings in the higher elevation butterflies like human poop. David had this species once near the farmhouse, so it gets around.

without, and about ten species of *Pedaliodes*. Fortunately Tomasz Pyrcz later helped me sort them all out.

You can drive to about 2,600m, then you can walk a bit higher. On a sunny day the views are spectacular (see back cover). You can also look for the rare *Hypanartia charon*, which we've seen up here a couple of times (see back cover). The first time I saw it, I 'assumed' it was just *Hypanartia dione*, without thinking how high we were. Fortunately one of my friends got some shots of it, and I just about had kittens later looking at his photos. Once we knew it was up there, David got good shots of it on a later trip.

Montezuma is also a fabulous spot for skippers. We've photographed a number of unknown species, just waiting for someone to collect a series and describe them. Some are

At the third signed spot, Cajones at 2,000m, there used to be a simple wooden table for a lunch spot, but it has rotted away. This is still a nice spot to spend some time if you have a vehicle. There has been a landslide with a big pile of scree on one side that opened up the trees, and one year there was an *Yanguna* that guarded this open area. We saw him several times, every time we went by when it was sunny, but he didn't like cameras. But David put out some bait, and he came down next to the road, but he still wouldn't sit for good photos. He turned out to be *Yanguna spatiosa*. We also had *Y. cosyra* at lower elevations, near the bridge.

As we went higher we were up into satyrland, and that's mostly what we saw. Several *Panyapedaliodes*, one of the prettiest of which is *Panyapedaliodes muscosa*, lots of the striking *Parataygetis lineata*, some with the white line and some

Top - *Yanguna* species: *spatiosa* (left) and *cosyra* (right); Bottom: *Panyapedaliodes muscosa* (left), *Parataygetis lineata* (right) (Photos: left two by Kim Garwood, right two by David Geale)

Top: "*Thoon*" sp. (left), "*Talides*" sp. (center), "*Damas*" sp. (right); bottom: *Carystina aurifer* (left), *C. mielkei* (right). (photos by David Geale)

nicely marked, like this one that I'm calling a *Thoon,* just to put a name on it. Another big skipper with red eyes and a pale fringe I'm calling a *Talides*. Interesting that both of these unknown species have a pale fringe; they're not just plain brown jobs. A third undescribed skipper that is fairly common we're calling a *Damas*, and it also has a beautiful yellow fringe. And we've had two species of the strongly marked *Carystina,* both *C. aurifer* and *C. mielkei*.

It's a good spot to work on those dark spreadwings. When they are on bait, they often let you lift their hindwings with a stick, so you can photograph both sides. This is critical for many of these very similar dark skippers, as they can look quite similar from just the dorsal. It can also be difficult to photograph them well, to show the subtle markings, when you have a dark bug on a bright, sandy background. For example, on *Ebrietas badia*, the ventral is a pale orange/tan, different from others in that genus. One of my favorites, when fresh, is the beautiful *Mictris crispus* with a

Top: *Ebrietas badia* (photos by David Geale)
Bottom: *Mictris crispus* (photos by Kim Garwood)

Top: *Telemiades centrites contra* (photos by Kim Garwood); bottom: *Potomanaxas laoma fumida* (left; photo by Kim Garwood), *Dalla eryonas* (right; photo by David Geale)

pale blue ventral. This one likes to hang out under the first big bridge. I've also seen *Telemiades centrites contra* once, on a spitwad. I didn't know what it was until I raised the wing and saw the bright yellow on the edge of the ventral.

And it's a great area for the complicated *Potamanaxas* genus. We have seen at least eight species so far, with help from Nick Grishin to sort them out. I like *P. laoma fumida*, which is common there at the first bridge. It's amazing how well they can blend into the gravel.

Then there's *Dalla*, in the *Hepteropterinae*. We have seen seven species so far. I really like these cloud forest skipperlings, but they can be very difficult to sort out. And there are so many possibilities. Lamas has 95 species in his Atlas of Neotropical Lepidoptera. Fortunately Bernard Hermier has spent endless time helping with id's on all our skippers. An easy one is *Dalla eryonas*, which is fairly common there. I've shot it right at the farmhouse where we stay. But the others can be really tricky. Sometimes you can spend all day just photographing this group, to sort them out later.

I was here in February 2013 with David, and it was supposed to just be for 3-4 nights. But there was a strike by the coffee pickers, and the center of the strike was in the coffee town of Apia, which is right on the one road out. So we were stuck, and ended up staying for 12 days at Montezuma. At least it was a great place to be stuck in. We had plenty of food and no problems, and we got to chase butterflies every day. We were still finding new species on our last morning.

You can walk down from the farmhouse a couple of hundred meters to another small bridge, and this can be worth checking out. There are nine species of *Mesosemia* at Montezuma, so far, and several of them seem to like this little bridge, down the sides by the water. This is a good spot for *Mesosemia pacifica* (see front cover). We usually see more females, with the cream band on the forewing, than the males. Maybe their host plant is somewhere around there, though we've never seen any egg laying. It's good for skippers too, with *Gorgopas chlorocephala* and more *Pyrrhopygini*.

Several other people, aside from those mentioned above, have been a huge help in id'ing our photos. Thanks to: Stephane Attal; Andrew Brower for those confusing *Heliconius* (Colombia seems to be a hotspot); Bernard Hermier, who has looked at many thousands of our skipper photos; James Mallet; Andrew Neild; Gregory Nielsen; Julian Salazar; Aaron Soh and Keith Willmott. Sorry if I have left anyone out, as many people have been consulted over the years.

Juan Guillermo Jaramillo V. and I are building PDF checklists for many locations in Colombia, and they are available online at my website, **www.neotropical butterflies.com**, for downloads. We are sharing these for free, and asking people that get photos they can't id to please submit them to me, so we can add them to our database of Colombian butterflies. The Montezuma PDF is over 240 pages long, so it's a large file. We're updating it as we add new photos and make corrections. If you find any errors, or have photos you would like to submit, please let me know. Thanks, **kimgrwd@sbcglobal.net.**

A persistent population of *Hodebertia testalis*, a milkweed leaftier, in Florida (Pyraloidea: Spilomelinae)

James Hayden[1] and Marc C. Minno[2]

[1]6400 SW 20th Ave., Apt. 126, Gainesville, FL 32607 **jehayden63@gmail.com**
[2]600 NW 35th Terrace, Gainesville, FL 32607 **marc.minno@gmail.com**

Hodebertia testalis (Fabricius, 1794) is an average-sized spilomeline pyraloid, varying from 11 to 13 mm in forewing length. The wings are whitish in real life, but fade to pale orange in museum specimens. The forewing costa, prothorax, and palpi are reddish-orange. There is a postmedial band of small, faint grayish spots on the forewings and hindwings, and other similar spots are in the forewing cell and elsewhere.

H. testalis is widespread in the Old World, ranging through southern Europe, Africa, tropical and subtropical Asia, and Australia (Shaffer and Munroe 2007; Rennwald 2016). It is also known from Cuba (Barro et al. 2011), Puerto Rico (Schaus 1940), and Costa Rica (Janzen and Hallwachs 2009). The species has many synonyms and combinations (Nuss et al. 2015); historically, it is most often placed in *Pyrausta* Schrank or *Palpita* Hübner, and most information can be found under the name *Pyrausta incoloralis* (Guenée). Until recently, it has not been recorded from the continental U.S.

In March 2016, Steve Nanz called JH's attention to photographs on Bugguide.net of moths raised in Harris County, Texas in 2008 (http://bugguide.net/node/view/1145829). The photographs had been identified as *Hahncappsia coloradensis* (Grote and Robinson), but the host (milkweed) caused Nanz to consider *Hodebertia*. The specimens were not kept, but the photographs show the diagnostic characters. As far as known, this was the first declared record in the continental U.S.

With this in mind, JH soon afterward discovered that MM had reared one in Florida many years beforehand. MM collected the larva in Broward County, Florida, in North Markham County Park, on 1 October 1983 (MM code M-593) and reared a female specimen (Fig. 8). He included it in a large donation to the Florida Museum of Natural History, McGuire Center for Lepidoptera in 2004. It is labeled "Reared on willow," but this was probably mislabeled. After finding the specimen in MM's donation, JH confirmed it by dissection (Fig. 12) and advised him about the discovery. Could *H. testalis* still be present in Broward County?

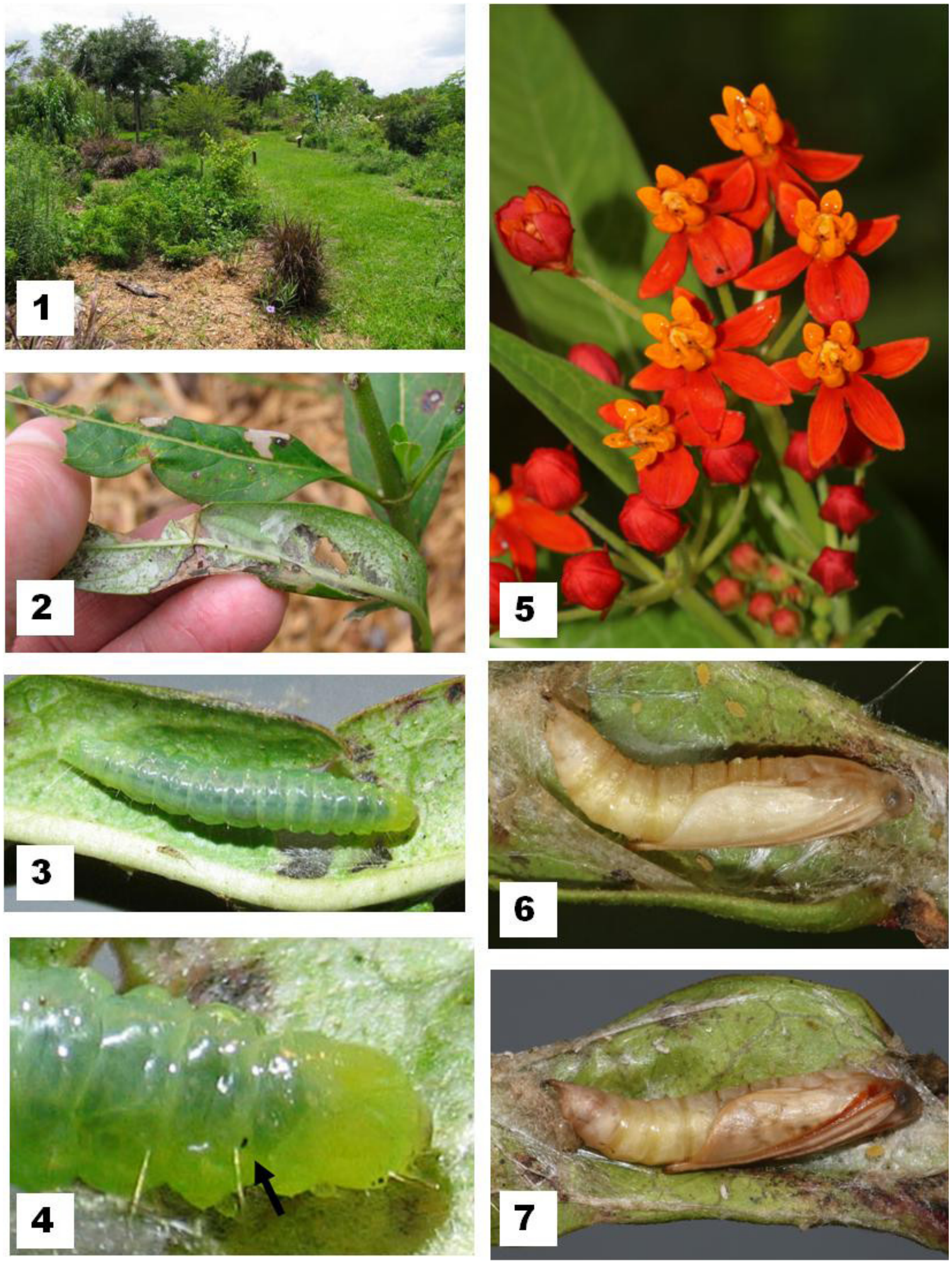
Late in the afternoon on May 27, 2016 MM revisited Markham to look for *H. testalis* and found a large butterfly garden in the southwestern area of the property (Fig. 1). Milkweeds present included several individuals each of *Calotropis gigantea* (L.) W. T. Aiton, *Gomphocarpus physocarpus* E. Mey., and *Asclepias curassavica* L. These were hurriedly examined, but seemed to be free of *H. testalis*. The last tropical milkweed (*A. curassavica*) plant examined was about three feet tall with leaves only at the tips of the stems. There were five nests with pyraloid pupae present among the leaves of this plant (Figs. 6, 7). The larvae had overlapped two leaves and tied them together with silk. The pupae were all facing toward the leaf tips. The eyes were darkening on some individuals. Five adults of *H. testalis* subsequently emerged beginning on May 29 and the last on June 1, 2016 (Figs. 9, 10). We were astonished that after more than 30 years MM was able to rediscover *H. testalis* in less than an hour of searching at Markham Park!

JH visited the same site on June 20, 2016 and found one leafy *A. curassavica* plant inhabited by larvae (Figs. 2-4), two of which emerged the first week of July. The specimens are deposited in the MGCL. A few other tropical milkweed plants in the garden were not inhabited. More *A. curassavica* plants were scattered through the landscape, but we have not had time to survey the whole population.

The nondescript moth resembles the pyraustine *Hahncappsia coloradensis* in the reddish-orange forewing costa. It differs in that 1) the frons is smoothly rounded, not produced as a tubercle, 2) the hind wing postmedial line is pronounced like that on the forewing, 3) males lack a forewing retinacular bar, and 4) females have two rather than three frenular bristles. *Pleuroptya silicalis* (Guenée) is structurally similar, but that species is darker yellow and does not have a reddish costa. The female genitalia (Fig. 12) have no signa. The male genitalia (Fig. 11) have a cross-shaped uncus, valvae with three sickle-shaped fibulae, and a hair pencil near the valve base.

Hodebertia Leraut belongs to the *Diaphania* group *sensu* Munroe (1995) of Spilomelinae (Crambidae) (Richard Mally, pers. comm.). Many genera in this group feed on laticiferous plants, especially Apocynaceae and Moraceae. Externally, the most similar Nearctic relative is *Palpita quadristigmalis* (Guenée), which is faintly yellow-white but lacks hind wing lines. The genitalia of *Palpita* Hübner species are very different, males having fibulae near the edge of the valva, and females having two thorn-like signa and the ostium bursae ingressed into the seventh sternite. The larvae of most *Palpita* species feed on Oleaceae. Munroe (1995) indicated that *testalis* required a new genus, and Leraut (2003) separated *Hodebertia* from *Palpita* Hübner on these characters. Adults key to couplet 2' (*Palpita*) in Hayden and Buss (2013), but the wings have prominent lines and the corpus bursae is unarmed. *Diaphania costata* (Fabricius) is nearly pure white without lines, and males have an expanded antennal scape, subcostal androconia, and black anal tuft.

The larva of *H. testalis* is almost uniformly plain green (Fig. 3). The pinacula are not contrastingly dark except for a black spot on the mesothoracic subdorsal pinaculum (Sevastopulo 1946) (Fig. 4), and the prothoracic shield is unpigmented. The larva keys to *Palpita magniferalis* (Walker) or *Glyphodes sibillalis* Walker in Allyson (1984).



Figs. 1–7. Markham Park Butterfly Garden and immature stages of *Hodebertia testalis*. **1)** Markham Park Butterfly Garden. **2)** larva under web on underside of leaf observed 6/20/2016. **3)** Larva collected 6/20/2016. **4)** Detail of larva with arrow indicating black spot on second thoracic segment. **5)** Inflorescence of *Asclepias curassavica*. **6, 7)** Two pupae collected on 5/27/2016.

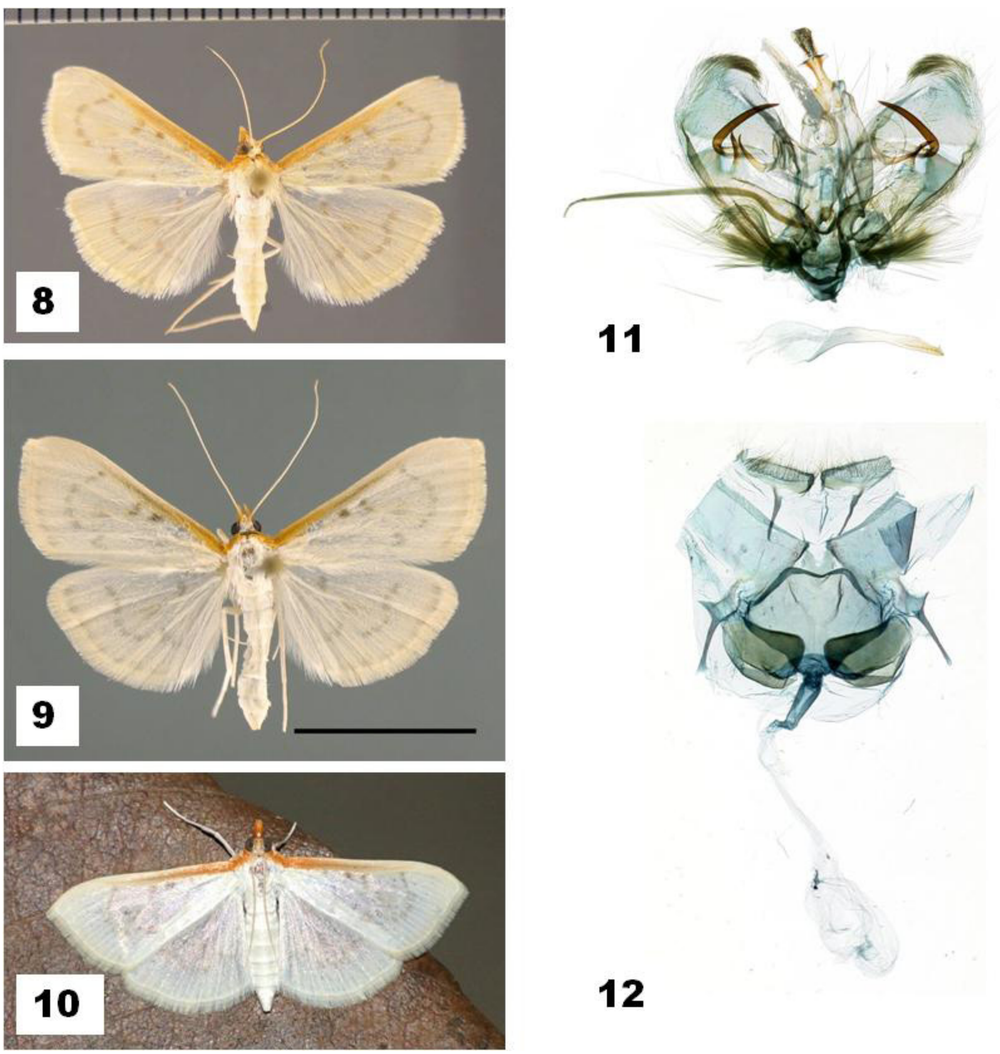
The recorded hosts of *H. testalis* are many asclepiadoid milkweeds, including members of *Asclepias* L., *Calotropis* R. Br., *Caralluma* R. Br., *Gomphocarpus* R. Br., *Leptadenia* R. Br., *Pergularia* L., and *Stapelia* L. (Leraut 2012, Robinson et al. 2001, 2010, Sevastopulo 1946, Shaffer and Munroe 2007). Records on *Sida rhombifolia* L. (Robinson et al. 2001) and *Hibiscus* (Martiré and Rochat 2008) need corroboration, but could be an alternative host family. Interestingly, *A. curassavica* is the only recorded host plant in the New World (Wolcott 1950, Janzen and Hallwachs 2009). We cannot think of any related spilomelines in North America that feed on the same plant genera. *Palpita flegia* (Cramer) feeds mainly on *Cascabela thevetia* (L.), and *Diaphania costata* feeds mainly on the apocynaceous tribe Vinceae, especially *Vinca* L. and *Amsonia* Waler.

The larva makes a thin covering of silk webbing on the underside of the host leaves in which to hide. Sometimes the leaves of the host are rolled or curled into the nest as well (Fig. 2). The webs and larvae are difficult to see due to their color. Pupation occurs in a rolled leaf or between overlapped leaves.

Asclepias curassavica (Fig. 5) is commonly planted in gardens throughout Florida and has long been naturalized in the southern part of the state. The dispersal ability of *H. testalis* is unknown. It is possible that this moth occurs more widely, but has been overlooked and not collected. However, tropical milkweed is a popular and highly watched plant in the southeastern US, because of butterfly gardeners looking for and raising monarchs (*Danaus plexippus* L.) on it. If *H. testalis* has not spread to other areas, why hasn't it dispersed? We welcome any additional observations of *H. testalis* that anyone can offer.

Acknowlegments

We kindly thank Andy Warren and Mary Karcher (MGCL) for accessioning and curating the original specimen. Steve Nanz first identified *H. testalis* from Texas and brought the matter to our attention. Bernard Landry kindly provided a copy of Martiré and Rochat (2008).



Figs. 8–12. *Hodebertia testalis*. **8)** Original female reared by MM in 1983 (scale mm). **9)** Female reared by MM from pupa collected 5/27/2016 (scale bar 5 mm). **10)** Live female reared by MM from pupa collected on 5/27/2010. **11)** Male genitalia of specimen reared June–July 2016, left androconium extended (MGCL Slide 3549). **12)** Female genitalia of specimen reared Oct. 1983 (MGCL Slide 3456).

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continued on p. 186

A biting midge, *Forcipomyia* (*Trichohelea*) sp. (Diptera: Ceratopogonidae), an ectoparasite of the Toltec Roadside Skipper, *Amblyscirtes tolteca prenda* (Hesperiidae)

Mark Salvato[1], Holly Salvato[1] and William Grogan, Jr.[2]

[1]1765 17th Ave. SW, Vero Beach, FL 32962 **anaea_99@yahoo.com**
[2]Florida State Collection of Arthropods, Florida Dept. of Agriculture and Consumer Sciences, 1911 SW 34th St., Gainesville, FL 32614-1700 **William.Grogan@freshfromflorida.com**

The Toltec Roadside-Skipper (*Amblyscirtes tolteca prenda* Evans) occurs locally within desert environments in southeastern Arizona and northwestern Mexico. Freeman (1993) discussed the distribution, morphology and natural history of *Amblyscirtes*, including *A. t. prenda*. In addition, Jim P. Brock (pers. comm.) has documented several parasitoids of larval *A. t. tolteca* in Sonora, Mexico.

On 3 August 2015 MS and HS observed and photographed a female biting midge (Diptera: Ceratopogonidae) attached to the ventral hindwing of an adult *A. t. prenda* (Fig. 1) at Harshaw Creek in Patagonia, Cochise County, Arizona.

Photographs of the midge were sent to WG who identified it as *Forcipomyia* (*Trichohelea*) sp., most likely *F.* (*T.*) *baueri* Wirth. *Forcipomyia* (*T.*) *baueri* is an ectoparasite known to occur within the United States (Arizona; California; Florida; Maryland) and Mexico (Veracruz) (Wirth and Messersmith 1971; Borkent and Grogan 2009). In addition to *F.* (*T.*) *baueri*, Wirth and Messersmith (1971) indicated that *F.* (*T.*) *goniognatha* Wirth and Messersmith, *F.* (*T.*) *leptognatha* Wirth and Messersmith, and *F.* (*T.*) *mexicana* Wirth have also been recorded from Arizona. Globally, species of *Forcipomyia* (*Trichohelea*) spp. are the only biting midges that have been recorded as ectoparasites of adult Lepidoptera (Wirth and Messersmith 1971; Lane 1984; Kawahara *et al*. 2006; Borkent and Grogan 2009; Sourakov 2013).

Tarsal claws of female *F.* (*T.*) spp. are equipped with a distinctive basal spine and comb in the inner margin (Lane 1984; Kawahara *et al*. 2006). Lane (1977) indicates that these claws may provide added support when clinging to wing scales of Lepidoptera, and that host options for *F.* (*T.*) spp. may be limited to species with specific scale characteristics.

In southern Arizona *F.* (*T.*) *baueri* has been documented as an ectoparasite of three lycaenids: *Callophrys gryneus siva* W.H. Edwards, *Euphilotes enoptes* Boisduval, and *Celastrina lucia* (W. Kirby) (Bauer 1961; Ehrlich 1962). To our knowledge this is the first known report of an adult female *F.* (*Trichohelea*) as an ectoparasite of *A. t. prenda*.

Acknowledgment

We thank Jim P. Brock for sharing observations and for helping us to obtain various publications related to *A. tolteca*.

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Fig. 1. An adult female biting midge *Forcipomyia* (*Trichohelea*) sp. attached to the ventral hindwing of *Amblyscirtes tolteca prenda* in Patagonia, Arizona (Cochise County) (Photo by H. L. Salvato).

The Marketplace

IMPORTANT NOTICE to ADVERTISERS: If the number following your ad is "583" then you must renew your ad before the next issue if you wish to keep it in the Marketplace!

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The following fascicles of the MONA series are for sale: 6: Pt.1; 7: Pt.1; 13: Pts. 1A, 2A, 1B, 2B; 15: Pt.5; 20: Pts. 1, 2A, 2B; 21; 26: Pt.1. I am asking for $35 each except 15 Pt.5 for which I am asking $65. Also, the Guide to the Olethreutine Moths of Midland North America (Tortricidae) for $10. I want to make sure mothers get these immaculate issues.

Contact: Larry Line, 6827 Redberry Road, Clarksville, MD 21029; **mothmanlarry@gmail.com**. 584

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The aim of the Marketplace in the **News of the Lepidopterists' Society** is to be consistent with the goals of the Society: "to promote the science of lepidopterology...to facilitate the exchange of specimens and ideas by both the professional and the amateur in the field,..." Therefore, the Editor will print notices which are deemed to meet the above criteria, without quoting prices, except for those of publications or lists.

We now accept ads from any credible source, in line with the New Advertising Statement at the top of this page. **All advertisements are accepted, in writing, for two (2) issues unless a single issue is specifically requested.** All ads contain a code in the lower right corner (eg. 564, 571) which denotes the volume and number of the **News** in which the ad first appeared. **Renew it Now!**

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Advertisements should be under 100 words in length, or **they may be returned for editing.** Some leeway may be allowed at the editor's discretion. Ads for Lepidoptera or plants must include full latin binomials for all taxa listed in your advertisement.

The Lepidopterists' Society and the Editor take no responsibility whatsoever for the integrity and legality of any advertiser or advertisement. Disputes arising from such notices must be resolved by the parties involved, outside of the structure of The Lepidopterists' Society. Aggrieved members may request information from the Secretary regarding steps which they may take in the event of alleged unsatisfactory business transactions. A member may be expelled from the Society, given adequate indication of dishonest activity.

Buyers, sellers, and traders are advised to contact state department of agriculture and/or ppqaphis, Hyattsville, Maryland, regarding US Department of Agriculture or other permits required for transport of live insects or plants. Buyers are responsible for being aware that many countries have laws restricting the possession, collection, import, and export of some insect and plant species. Plant Traders: Check with USDA and local agencies for permits to transport plants. Shipping of agricultural weeds across borders is often restricted.

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Techniques for making micro-moth blocks

John B. Heppner

McGuire Center for Lepidoptera and Biodiversity, Florida Museum of Natural History, University of Florida
Gainesville, FL 32611 **jheppner@flmnh.ufl.edu**

Blocks used for the double-mounts needed for micro-moths mounted on minuten pins have always presented problems for production. The techniques for making micro-blocks noted below are what is being done at McGuire Center.

In the past, most researchers of microlepidoptera used strips cut from polyporus fungi, producing a pleasing white micro-block that would firmly hold a minuten pin and also remain steady on the main insect pin. Dealers generally supplied short strips (ca. 4 inches long) of polyporus cut to 3 x 3 mm size, which then were simply chopped at desired lengths for making the micro-moth micro-blocks. Some 15 years ago, however, the main supply of polyporus fungi was removed from use when authorities declared the polyporus fungi endangered in Britain, possibly all of Europe. Although this large fungus grows on trees throughout Europe, the main market for the polyporus strips came through entomological dealers in England, then transshipped throughout the world. Previously, some curators also used soft woods, like the tropical balsa wood, but wood is not spongy enough to firmly hold pins adequately. Likewise, in western North America some tried using the pith of the flower stems of yucca plants, but this also is not spongy enough to be useful, is brownish, and is hard to obtain nationwide on a large scale. Even cardboard or thick card stock was used, but also not satisfactorily. Stiff plastic foam-like materials, like styrofoam, are also inadequate, since these also have no spongy properties to be able to hold pins firmly, and the pins, micro-blocks and micro-moths then quickly become loose enough to twirl around.

With the lack of polyporus for double mounts, substitutes included various plastic foams or even rubber-like neoprene materials. These proved to be too flimsy for adequate use with minuten pins or too rough a grade of foam. The problem actually involved plastic foam usually not being dense enough. These kinds of foam sheets used for packing are of various stiffness and smoothness, depending on the number of air bubbles and plastic polymers used in the formation of the foam sheets; much of the foam we now use in unit trays and insect boxes has large air bubbles and is fairly firm and spongy but not dense enough to adequately hold minuten pins. In recent years, a dense plastozote-like foam is manufactured where the air bubbles are extremely small, producing a fine, smooth, and very dense white foam which is stiff yet spongy. This has proven to be perfect for minuten pin double-mounts, even better than polyporous.

The main supplier of the densest spongy foam appears to be in Taiwan. This very dense, smooth, and heavy white plastozote-like polyethylene foam is commercially used for padding of heavy but delicate machinery and other items for shipment. This type of plastic foam is much denser and heavier than the typical plastazote-type smooth foam from England used in finer unit trays. In the USA, plastozote-type dense foam of various densities can be obtained, but the sources have not been checked to verify that the foam is identical to the dense and spongy foam from Taiwan. For example, the Foam Factory (www.foambymail.com) sells polyethylene cross-linked foam of 2lb density and 3/8 inches (10mm) thick in 48 x 24 inch sheets for $15: this thickness is the closest to 10mm thickness and may be stiff enough for micro-mounts, but the flexibility (sponginess) has not been tested. Another foam available is called "closed-cell" polythylene foam, with 6 lb density and likely to be more rigid, but is usefully available only in 1/2 inch (13 mm) thickness at $54 per sheet of 108 x 23 inches, which would allow production of longer micro-blocks, usable if spungy but a bit longer than desired for most micro-moths. Plastozote itself can be ordered direct from England, from Thames Valley Supplies (www.thamesvalleysupplies.com): their LD70 firm density plastozote sells for about $55 per large sheet of 1.7 x 0.85m (ca. 66 x 33 inches), but 3 sheets is the minimum order, plus shipping to the USA. However, in the USA smaller quantities of perhaps the same or similar plastozote can be ordered from Bioquip Products (www.bioquip.com), with 3/8 inch (10mm) thick sheets of 16 x 18 inches for $6.25 each (cheaper per sheet when bought in cartons of 24 sheets, at $135 per carton), plus shipping. However, as noted for Foam Factory items, the actual density may not be the same as the material from Taiwan, which is about twice the density of regular plastozote (maybe LD120 or more).

The very dense foam used at McGuire Center is 10 mm thick and comes in sheets that can likely be ordered in various sizes, with our stock size being about 40 cm long and 30 cm wide, originally from Taiwan (actual source unknown and shipped to us from the Taiwan National Museum). As the figures on the next page show, the foam sheets are cut into 3mm wide strips (Figs. 1-2), becoming then 10 mm high and 3mm thick, and 40 cm long. These long strips are then chopped into desired micro-blocks by vertically cutting the height (10 mm) of the strips into 3 mm sections (Fig. 3), thus producing micro-blocks that are 10 mm long and 3 mm square. Longer micro-blocks could be made by cutting the strips into lengths desired and then horizontally chopping out the micro-blocks from the longer sections, or using foam sheets of 1/2 inch (13mm) thickness, but we find the 10mm length to be adequate for virtually all micro-moths (those with long legs have the legs placed either side of the main pin). Foam sheets that are about

Fig. 1-12. Making micro-moth blocks: 1) Balsa razor stripper-cutter. 2) Razor stripper cutting foam strips (razor blade adjusted to 3mm wide cuts). 3) Single-edged razor vertically cutting 4 strips of foam at 3mm width. 4) Micro-block transfer to box. 5) Razor turned over to scrap off the micro-blocks into the holding box. 6) Assemblage of finished micro-blocks (several hundred). 7) Types of wodden step-blocks (cut into steps ot with holes of different depths). 8) Simple step-block made of foam and card stock layers taped together and over a mini-petri dish for correct pinning height. 9) Micro-block pinned with step-block. 10) Finished double mount. 11) Box of finished double mounts (several hundred). 12) Prepared micro-moth on double mount, with locality label beneath (note that moths are positioned with legs directed to the #4 insect pin, not sideways with the wingtips near the insect pin).

30 x 40 cm in size (10 mm thick) produce about 95 strips each, enough for making about 12,600 micro-blocks from one such small sheet of foam.

The procedure for cutting the foam strips and micro-blocks, however, requires very sharp razors: a razor balsa cutter for the strips and then single-edged razors for chopping the micro-blocks. It has been found that the foam is so dense that most razors will only be useful for chopping micro-blocks for one or two batches of 4 strips (40 cm long). The micro-blocks are chopped using 4 foam strips at a time (Fig. 3), since this provides a firmer base when holding the strips together on which to chop the micro-blocks away from the strips, rather than single strips, and also is more efficient in the number of cuts to be made. Once a razor is dulled too much, chopping the micro-blocks becomes difficult to do neatly. A strip razor in the strip cutter, however, lasts longer than the razor for micro-block chopping. One generally has to hold the single-edged razor in the fingers for chopping the micro-blocks, since razor holders do not hold the razor at the correct height to allow the micro-blocks to be cut through the 10 mm depth of the foam strips. One has to chop the micro-blocks very carefully perpendicular to the strip lengths so the micro-blocks come out with even and straight edges (Fig. 3).

When cutting the long foam strips, the razor cutter has to be held level so the razor in the cutter is precisely perpendicular to the foam sheet (Fig. 2). Then the strips come out even in width for the entire length of each strip. The cutting razor may vary the strip thickness if the cutting angle is not uniform the length of the strip. Cutting the foam strips requires a steady hand and a firm pressure on the cutter razor to keep it tight on the foam edge, level, and to cut through the dense foam. A little practice makes all of this fairly simple.

When chopping micro-blocks, the only hindrance is the natural static electricity produced in cutting and chopping plastic foam. This necessitates moving the micro-blocks from the chopping razor onto the rim of the micro-block box after each cut (Fig. 4), then turning the razor over (Fig. 5) and scraping them into the box from the box edge. The static electricity will keep the micro-blocks on the razor until scraped off. Even so, many micro-blocks will "jump" away and will have to be retrieved into the micro-block box from time to time. Once a moth is on the double mount (Fig. 12), the static from the plastic foam is subdued (grounded) by the insect pin and is no longer a problem.

The entire operation, from cutting foam strips to chopping the micro-blocks can be done in a few hours to produce about 1600 micro-blocks from 12 foam strips of 40 cm length. Doing any more than this in one day tends to tire the hands and fingers too much from holding the razor firmly and the repetitive movement back and forth to the micro-block box to deposit the finished micro-blocks. The micro-blocks can be stored in boxes and be ready for pinning (Fig. 6), as can the finished double mounts (Fig. 11). This is a further benefit to using such foam micro-blocks, since museum pests would also in the past eat the polyporus as well as the specimens.

For double mounts, we use standard procedures for micromoths by pinning micro-blocks with a #4 insect pin at one end (Figs. 8-10). We use a uniform height of 15 mm from the top of the insect pin for the micro-block position (Fig. 10), then add the moth on its minuten pin for the final preparation stage, prior to labeling the specimen with a locality label below the micro-block (Fig. 12). A standard insect step-block can be used, either wooden as often sold by dealers (with steps cut in or with holes of different depths) (Fig. 7), or one can also fabricate a simple foam and paper step pinning assembly taped together, with a central hole, and made so the height is correct to position the micro-block (Fig. 8). Holding the minuten pin for preparation of specimens and adding a specimen to the micro-block is best done using a small forceps designed for pulling hairs, such as women's eyebrow forceps, and they must have square-edged and evenly ground inner tips so the minuten pins can be held firmly. When procedures are done with uniform methodology, a neat and museum-quality result is obtained, ready for final curation of specimens (Fig. 12).

More Announcements:

Continued from p. 201

Lepidopterists' Society Statement on Diversity, Inclusion, Harassment, and Safety[1]

During the Executive Council (EC) Meeting on 6 July 2016 in Florissant, Colorado, it was proposed that the Lepidopterists' Society adopt a Statement on Diversity, which would include a policy on harassment and safety. With the valuable input of past EC members, this statement was drafted over several weeks by the current EC membership. This statement is important to help our members feel safe during Society events, and provide the necessary means to resolve situations should they occur. The following statement was approved by the EC on 13 November 2016.

"The Lepidopterists' Society values diversity among our membership, just as we value diversity within the biological communities we study. We welcome into our Society and encourage the participation of all individuals who are interested in Lepidoptera regardless of age; gender; gender identity; sexual orientation; race; ethnicity; cultural background; nationality; religion; physical or mental ability; professional status; opinions on collecting, observing, and photographing; and all other characteristics and activities that make our members unique.

"The Lepidopterists' Society is dedicated to providing a safe, hospitable, and productive environment for everyone attending our events. We therefore prohibit any and all intimidating, threatening, or harassing conduct during these events. Harassment includes, but is not limited to: offensive gestures or verbal comments; the sending or sharing of offensive images, videos, emails, texts, or voicemails; deliberate intimidation; stalking, following, harassing photography or recording; sustained disruption of talks or other events; inappropriate physical contact; and unwelcome attention. Participants asked to stop any harassing behavior are expected to comply immediately. This policy applies to all event speakers, staff, volunteers, exhibitors, and attendees.

"The Society may take any action it deems appropriate in dealing with an event participant who engages in harassing behavior, ranging from a simple warning to expulsion from any Society sponsored events to loss of membership in the Society.

"If you are being harassed, if you notice that someone else is being harassed, or if you have any other concerns, please do not hesitate to contact the Society's designated ombudsperson, who will work with the appropriate Society leadership to resolve the situation. The designated ombudsperson will always be identified by name in the event's program book, along with their contact information. If needed, the Society will also help participants get in touch with convention center/hotel/venue security or local law enforcement, and otherwise assist those experiencing harassment, to enable them to feel safe for the duration of our events."

[1]Based in part on the Entomological Society of America's Statement on Diversity & Inclusion and Code of Conduct

John V. Calhoun, President

Cocoon spinners weave a story of adaptation

Michael M. Collins

Associate, Carnegie Museum of NH, Invertebrate Zoology, Pittsburgh, PA & 215 Prospect St., Nevada City, CA 95959
michaelmerlecollins@comcast.net

Cocoon-spinning larvae weave a story of adaptation, written in their chromosomes, and expressed as an intricate structure of silk that protects the enclosed pupa from severe weather and natural predators. With the exception of a series of intriguing papers from the 1950s to 1970s (reviewed in Tuskes et al. 1996, pp 21-23), spinning behavior and the biophysics of the cocoon in saturniids have not been well-studied, in spite of the group's popularity. Today's technology presents an opportunity for both the amateur and professional to conduct valuable research, using organisms that are easy to rear and observe in the lab (Collins 2011). What are the mechanical properties of the cocoon of a given species? How effective is the cocoon against predation? What are the thermal properties of a cocoon, for example the possible role of albedo in light-colored cocoons of desert species? Are cocoons chemically protected against predation or fungal infection? To attract attention to these and related topics, the following photo essay illustrates stages in cocoon spinning in *H. columbia gloveri* (Strecker), using images of reared stock from Mono Co. CA, near Sonora Pass.

Following a wandering phase the pupating larva selects a spinning site, often characteristic for a given saturniid species, but not well-understood in terms of response by the larva to key environmental stimuli. The larvae of *gloveri* typically wander from the immediate host and select a protected site near to ground level, usually in dense growth. The grey to light brown cocoons have a striated or banded appearance (see below), which adds to their crypsis, especially against shreddy bark (e.g. *Prunus, Purshia, Rosa, Salix*) or within a grassy understory. The cocoon is double, but only the construction of the outer envelope is easily studied.

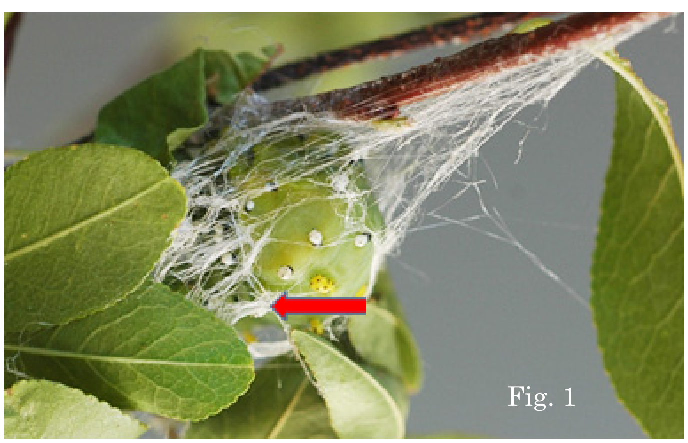

Fig. 1

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Fig. 2

Fig. 3

The larva (Fig. 2, 3) seems to give special attention to attachments to leaves, stems, and twigs, which support the entire envelope. The larva faces the top in Fig. 2, and has turned to face the bottom in Fig. 3.

Fig. 1 (at left). The larva first spins an attachment to a branch or twig. Bands (arrow) are among the first features incorporated into the outer envelope. The larva turns around periodically within the silken superstructure to give symmetry and a uniform density of woven strands to the outer cocoon. The basic outline of the emergence valve (at right) is constructed at this time.


Fig. 4


Fig. 5

The larva (Fig. 4, 5) now begins final construction of the valve, forming a conical structure of parallel fibers, through which the adult moth will subsequently emerge. The larva will stretch out to its full length in spinning these fibers. Just before eclosure the adult moth releases a special "cocoonase" enzyme, which dissolves the glue holding the silk strands together.

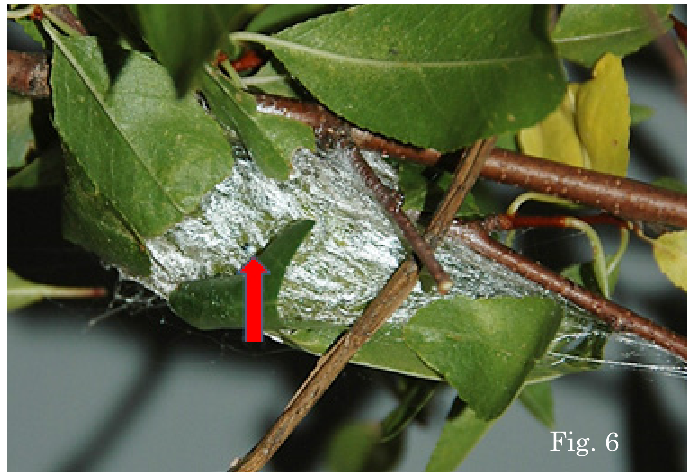
Fig. 6


Fig. 7

By turning around at regular intervals the larva uniformly fills in the outer envelope (Fig. 6). It is still possible to observe the larva; a blue scolus is just visible left of center (arrow). After about a day, the larva secretes a fluid that permeates the cocoon, darkens the fibers, and when dry makes the cocoon somewhat water-proof (Fig.7). The strands and bands of silk in the outer envelope resist this effect and give the cocoon a silvery, striated appearance, adding to its crypsis. Over time exposure to the elements tends to bleach the newly spun cocoon (see Fig. 8).

Fig. 8. Cocoon of *H. c. gloveri* in the wild, spun on host *Rosa woodsii*, Mono Co. CA along Walker River. The placement, cryptic texture, and color of these cocoons make them difficult to find, even by experienced collectors, and presumably by predators also.

Recent records of *Hyalophora columbia* (S. I. Smith, 1865) (Saturniidae) in New York State

Janet R. Mihuc

Paul Smith's College, P. O. Box 265, Routes 86 & 30, Paul Smiths, New York, 12970 **jmihuc@paulsmiths.edu**

Keywords: *Larix*, Adirondack Mountains, Saturniidae, *Hyalophora*

The Adirondack Park encompasses an area of 2.5 million hectares of state and private lands in northern New York. In this state park, the Adirondack Mountains contain over 10,000 hectares of lowland boreal communities. The distribution of these communities in the northeastern United States will be reduced by climate change (Jenkins 2010). One important boreal tree species, the tamarack, *Larix larcina* (Du Roi), is the primary host plant of the Columbia silk moth, *Hyalophora columbia columbia* (S. I. Smith, 1865) (Saturniidae: Saturniinae). Ferguson (1972) and Tuskes et al. (1996) speculated that this species should be present in northern New York but no specimens exist in the New York State Museum (T. McCabe, personal communication). Three *H. columbia* specimens in the United States National Museum have collection dates of 1865 and are labeled 'New York' (Ferguson 1972). There is one specimen of *Hyalophora columbia columbia* at the American Museum of Natural History dated 20 June 1892 from New Windsor, NY (D. Grimaldi, personal communication). There are no records of *H. columbia* specimens from New York in the museum collection specimen data available through the Symbiota Collections of Arthropods Network (SCAN 2016) and no record of this species in New York field season summaries of the Lepidopterist's Society. Recent photo-documentation of *H. columbia* exists for similar latitudes in Vermont, New Hampshire, Maine and Ontario (Lotts & Naberhaus 2015, Moth Photographers Group 2016). Here I report photo-documented sightings of *H. columbia* from New York along with results from placement of virgin females at field sites in 2012-2014. The southernmost location of *H. columbia* specimens from the eastern half of the United States is Jackson Co., Washtenaw Co. and Wayne Co. Michigan (Lotts & Naberhaus 2015, SCAN 2016, Moth Photographers Group 2016) which is within three degrees latitude of the sightings reported herein.

My first record of *H. columbia* in New York came from a male I collected at a MV light on 4 June 2004 in Saranac, NY (Clinton Co.). In 2011, eggs were obtained from a wild female collected on 9 June at a MV light in Paul Smiths, NY (Franklin Co.) (Table 1). The larvae were reared on *Larix laricina*. Two females from the overwintered cocoons were placed in mating cages at four locations in May 2012. One female placed at The Wild Center in Tupper Lake, NY successfully attracted a male *H. columbia* on the second night after eclosion (Figure 1). Another female placed in Lake Placid, NY successfully attracted a male on the second night after eclosion (Table 1). Both of these females had been placed at other locations without success on the first night after eclosion (Table 2). Most of the larvae from both pairings were released in Tupper Lake and Lake Placid at the sites where the females had mated, but I reared four larvae on *L. larcina* and overwintered the cocoons. In May 2013, two females from the overwintered cocoons were placed in mating cages in four locations (Table 2) but no males were observed and none of the resulting eggs were fertile.

I received a female *H. columbia* from the Wild Center in May 2013. This female was collected at a MV light on the building but it could have been an adult from the cohort of caterpillars I released at that location the previous year. The female mated with a male that I collected at a UV light in Saranac, NY (Table 1). The larvae were reared in a wire mesh enclosure containing cut branches of *Larix laricina*. Of the 50 cocoons that overwintered from this pairing, 37 eclosed and 19 of those were female. Weights of the cocoons that did not eclose ranged from 0.5 to 1.18 grams each whereas the lowest weight recorded for an eclosing cocoon was 1.37 grams. The low cocoon hatch rate may have been due to low body weight as no parasitism was observed. All but one of the 19 females emerged between 28 May and 31 May 2014. The last female emerged 4 June. Females were placed individually or in pairs in mating cage traps described in the following paragraph. Cage traps were placed at field sites beginning on the day of eclosion and for up to 3 consecutive nights thereafter. In the majority of cases, the traps were moved to a different location each day. All females began laying eggs on the third, fourth or fifth day after eclosion. *Hyalophora columbia* eggs from previous years had hatched in 13-14 days so all eggs laid by each female were kept for 20 days to allow for hatching. Field sites were chosen based on presence of the host plant and proximity to locations of documented sightings. Field sites used in 2014 were located in Clinton, Franklin, Essex and Hamilton counties at least one mile from the location of any documented sighting (Table 2). Traps were placed within or on the edge of stands of *Larix laricina*. No males were captured or observed at any of the traps placed in the wild and none of the eggs laid by the females were fertile.

The mating cage used in 2012 and 2013 was a 17.5 cm x 15.2 cm cylinder of 1.3 cm wire mesh (hardware cloth) with a plastic lid attached to each end. A male arriving at the cage would have to mate with the female through the wire mesh. In 2014 I designed a mating cage trap because I wanted to place individual females in as many locations

Table 1. Locations where *H. columbia* was documented with either a photograph or a specimen collected by the author. Coordinates are listed as latitude and longitude in decimal degrees (D.D). Photos of *H. columbia* from the first Lake Placid site listed in the table can be found on iNaturalist.

County	Town, coordinates	Date	Source
Clinton	Saranac, 44.659N, -73.800W	4 June 2004*, 31 May 2013	Specimen collected at UV light
Essex	Lake Placid, 44.267N, -73.951W	23 May 2012	Wild male mated with virgin female in mating cage.
	Lake Placid, 44.263N, -74.013W	8 June 2011	Project Silkmoth sighting*
	Newcomb, 43.970N, -74.221W	5 June 2014	Butterflies and Moths of North America sighting
Franklin	Paul Smiths, 44.434N, -74.249W	9 June 2011	Specimen collected at MV light
	Lake Clear, 44.380N, -74.203W	4 June 2013	Project Silkmoth sighting*
	Coreys, 44.219N, -73.304W	13 June 2012	Project Silkmoth sighting
	Tupper Lake, 44.219N, -74.438W	25 May 2012	Wild male mated with virgin female in mating cage.*
St. Lawrence	Madrid, 44.774N, -75.081W	28 May 2013	Project Silkmoth sighting*

* photo in Figure 1

Table 2. Locations where mating cage traps were placed but no *H. columbia* were recorded. Each trap contained 1-2 *H. columbia* virgin females. Traps were in place for 1-3 nights between 22 May and 2 June in the year listed. Coordinates are listed as latitude and longitude in decimal degrees.

County	Coordinates	Year	Site name
Clinton	44.514N, -73.891W	2014	Silver Lake bog 1
	44.510N, -73.885W	2014	Silver Lake bog 2
	44.613N, -73.758W	2012	Pup Hill Road
	44.757N, -73.939W	2013	Merrill
	44.470N, -43.819W	2013	Hawkeye
Essex	44.429N, -74.040W	2014	Fletcher Farm road
	44.392N, -74.056W	2014	Kanze
	44.377N, -73.823W	2012	Wilmington
Franklin	44.373N, -74.502W	2014	Spring Pond bog 1
	44.369N, -74.498W	2014	Spring Pong bog 2
	44.374N, -74.191W	2013	McMaster Road
	44.480N, -74.129W	2013	Rainbow Lake
Hamilton	43.952N, -74.767W	2014	Shingle Shanty Preserve 1
	43.951N, -74.739W	2014	Shingle Shanty Preserve 2
	43.946N, -74.779W	2014	Shingle Shanty Preserve 3
	44.050N, -74.585W	2014	Whitney
	44.054N, -74.614W	2014	Burn road
	44.074N, -74.705W	2014	Sabattis railroad
	44.081N, -74.670W	2014	High Pond bog

as possible and knew that assistants would be placing the traps at remote sites and not retrieving them until the next day. The trap design was intended to prevent males from escaping during the day after the trap had been placed in the field. Each trap was constructed out of a 3.8 l clear PVC square container with a lid (U.S. Plastics Item 072037). Holes (3 mm diameter) were drilled at 2.5 cm intervals in the lid and on all four sides of the container. The female was housed in the top third of the container above 1.3 cm mesh hardware cloth. On the two widest sides of the container, 7.6 cm x 5 cm openings were made just below where the female was housed. The openings were covered with a flap of polyester window screen taped to the inside to allow males to access the female but to prevent them from leaving after mating. Traps were placed at each site before dark and site GPS coordinates were recorded. Because no males were captured in the mating cage traps, the efficacy of this design was not verified.

I have compiled photo-based sightings of *H. columbia* through an online citizen science project. In May 2010 I established Project Silkmoth, a website where sightings of adult saturniids can be submitted (Mihuc 2010). The target geographic area of the project is northern New York, defined as the area of the state that is north of a line from Syracuse to

Figure 1. Photos of Hyalophora columbia from locations in New York: 1a) Clinton Co., Saranac, 4 June 2004 (photo by J. Mihuc); 1b) Essex Co., Lake Placid, 8 June 2011 (photo by L. LaPan); 1c) Franklin Co., Tupper Lake, 25 May 2012 (photo by J. Mihuc); 1d) Franklin Co., Lake Clear, 4 June 2013 (photo by M. Johnson); 1e) St. Lawrence Co., Madrid, 28 May 2013 (photo by B. Doman)

Albany (i.e. 42.5 degrees north latitude). Twelve species of saturniids are considered 'target species', including *H. columbia*. Information requested on the required sighting form includes date, location with address or coordinates, habitat category, host plant species in the area, method of identification and light type. The form also contains a space for optional information on weather condition and details about each moth. Photographs are required for certain species, including *H. columbia*, for verification of species identity.

Through Project Silkmoth, photo-documented sightings of adult *H. columbia* have been received from four locations, representing three of the four northernmost counties in New York (Figure 1). Specimens were collected by the author in the other northernmost county (Clinton Co.) in 2004 and 2013. Of the two photo-documented sightings of *H. columbia* in the Butterflies and Moths of North America (BAMONA) database (Lotts & Naberhaus 2015), one represents the southernmost sighting for the state, in Newcomb, located in Essex Co., New York (Table 1). Photo documentation of *H. columbia* in New York can also be found on the iNaturalist website (SCAN 2016). The location of the iNaturalist sightings in Lake Placid, NY are on the 14ha property where I successfully located a wild male using a caged female in 2012 (Table 1). The 2004 specimen from Clinton Co. is vouchered at the New York State Museum. In May 2016, three male *H. columbia* specimens were collected in Paul Smiths, NY and are vouchered at the Carnegie Museum of Natural History.

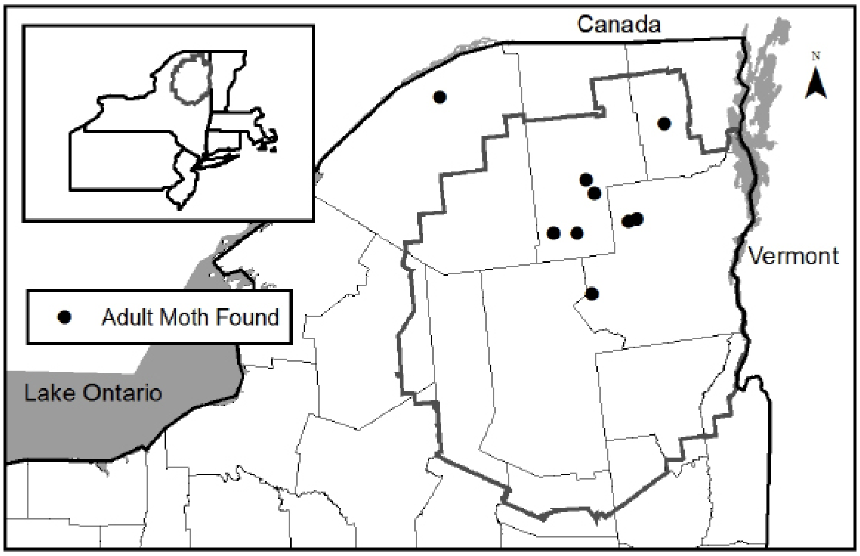

Figure 2. Map of *Hyalophora columbia* sightings from New York. The Adirondack Park boundary is shown in bold. Dates and further information appear in Table 1.

All of the documented locations of *H. columbia* described herein can be seen on the map in Figure 2. The geographic spread of the locations where *H. columbia* was found suggests that multiple populations exist within and around the Adirondack Park. The westernmost sighting, in St. Lawrence County, is 78km from the nearest sighting in adjacent Franklin County. The easternmost sighting, in Clinton County, is 43km from the nearest sighting in adjacent Franklin County. These distances are greater than the 12km flight distance of the closely related *Hyalophora cecropia* (Linnaeus, 1758) reported by Waldbauer and Sternburg (1982) and the 36km flight distance of *Callosamia promethea* (Drury, 1773) reported by Toliver and Jeffords (1981). Further study of the distribution of *H. columbia* in New York is warranted considering the existence of credible 19th century specimens and near lack of 20th century records. Climate modeling data available on the USDA Forest Service Climate Change Atlas indicate that the geographic distribution of *Larix laricina* is expected to shrink in the future (Landscape Change Research Group 2014). Other coniferous tree species within the boreal forests of the Adirondack Mountains are predicted to decline in abundance with climate change (Jenkins 2010, Landscape Change Research Group 2014). The locations of *H. columbia* reported herein represent a source of baseline data for future comparison.

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I thank Lydia Wright for sharing her expertise in rearing silk moths and her help in locating silk moths in Paul Smiths, New York. I thank the Nature Conservancy's Adirondack Land Trust for granting access to the Spring Pond Bog preserve and the Silver Lake Bog preserve. Shingle Shanty Preserve and Research Station granted access and Steve Langdon placed mating cage traps at sites on preserve lands. Angelina Ross and Kevin Ohol were helpful in placing mating cage traps at locations in Hamilton County, NY. I am grateful to Ed Kanze and Larry Master for allowing placement of female moths in mating cages on their property. The Wild Center in Tupper Lake, NY allowed female moths in mating cages to be placed on the property and provided live adult moths found at lights on the building. I thank Paul Smith's College for hosting the Project Silkmoth website. Chloe Mattilio produced the map used in this article. Initiation of Project Silkmoth was funded by a Cullman Foundation grant from the Northern New York Audubon Society.

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From the Editor's Desk

James K. Adams

Dry, warm, and smokey describes the air here in Dalton, GA, as I wrap up this issue of the News. Needless to say, there is NOT much happening Lepwise currently. At least this issue of the News is jam packed with an awful lot of stuff, including announcements about Lep Soc 2017. Hope to see you there! And happy holidays!

A non-intrusive technique for marking Duskywings (genus *Erynnis*) and other butterflies

Gard W. Otis[1] and Jessica E. Linton[2]

[1]School of Environmental Sciences, University of Guelph, Guelph, Ontario N1G 2W1 Canada **gotis@uoguelph.ca**
[2]Natural Resource Solutions, Inc., 225 Labrador Drive, Unit #1, Waterloo, Ontario N2K 4M8 Canada
jlinton@nrsi.on.ca

The Mottled Duskywing, *Erynnis martialis*, is considered to be a "Species at Risk-Endangered" in both Ontario (Government of Ontario, 2016) and Canada as a whole (Government of Canada, 2016). The recent (2008) discovery of a relatively large population of this species in eastern Ontario provided an opportunity for intensive research in 2015 on its habitat utilization (Otis and Aguilar, in prep.). However, because of its endangered status, that study was restricted in scope because, in the absence of a permit under the *Endangered Species Act* (OESA, 2007), we were not allowed to handle the butterflies which greatly limited our ability to mark them. Marking butterflies with distinctive, individual marks is essential if one wants to quantify survival, longevity, population size, outcomes of interactions, and in most cases movements within and between populations.

Researchers generally utilize one of two techniques to mark butterflies. Ehrlich and Davidson (1960) developed a system of dots applied with permanent marking pens near the edges of the undersides of the wings. Marks on one side indicated the numbers 1, 2, 4, and 7; marks in the same positions on the other side indicating the numbers 10, 20, 40, and 70 (see diagrams in Ehrlich & Davidson, 1960, Southwood, 1978; Brussard 1981). This numbering system can be extended to 100s and even 1000s, if required, by placing additional dots medially on the wings (Southwood, 1978; Brussard, 1981) and by applying different colors of dots. This system was used by Paul Ehrlich and associates in their long-term studies of *Euphydryas*; by Ward Watt et al. (1977) on *Colias*; by Frances Chew and associates (Chew, 1981; Nakajima *et al.*, 2014) on *Pieris*; and by Peter Brussard and asssociates (1970, 1981) on *Erebia*. With the advent of inexpensive fine-tipped permanent markers and drawing pens, most researchers have switched to writing actual numbers on the ventral surfaces of hindwings, sometimes on one wing only (e.g., Haaland, 2015; Li et al., 2016; Polic *et al.* 2014; and many others), other times on both hindwings (e.g. Ehrlich and Gilbert, 1973). Most marking systems require recapturing the butterflies to see the marks and identify individuals.

A large proportion of researchers release the butterflies into their environment immediately after marking and handling, with the underlying assumption that butterfly behavior has not been affected by the trauma caused by handling them. However, several butterfly species, upon release, do alter their behavior in ways that reduce their likelihood of being recaptured. There has been no systematic review of which taxa exhibit evasive behaviors, however they have been inferred by reductions in recapture frequency in *Graphium* (Singer and Wedlake, 1981), *Papilio* (Lederhouse, 1982), and *Melanargia, Maniola, Colias,* and *Thymelicus* (Morton, 1982). Watt *et al.* (1977) stored butterflies after capture at ambient temperatures (<20C), forcing them to bask to warm up before resuming flight. They showed no evidence of "capture release trauma," leading the authors to conclude (p. 4):

> *"Apparently the agitation involved in capture is behaviorally erased by time spent at low body temperature, so that solar warmup after release leads to normal flight behavior."*

A similar absence of evasive behaviors was observed in Macoun's Arctics (*Oeneis macounii*) that were allowed to cool to ambient temperature before marking and release (Burns, 2013). In a small but important study, Kemp and Zalucki (1999) empirically showed that *Hypolimnas bolina* males that were released immediately after marking exhibited evasive flights that significantly reduced recapture probability in comparison to individuals that were chilled before release or were marked without handling. Bull et al. (2006) chilled Monarchs (*Danaus plexippus*) for 10 minutes to minimize the effects of handling on their behavior.

We wanted to develop a marking technique for future research on the endangered Mottled Duskywing (Hesperiidae: *Erynnis martialis*). Our goals were:

1. To handle the butterflies as little as possible, to minimize damage such as loss of wing scales, loss of legs, and structural damage to wings that may affect subsequent flight, survival, mating, and/or oviposition;
2. To minimize handling trauma and associated atypical behavior following release;
3. To apply unique markings that enable identities of individuals to be determined visually without requiring recapture.

We first experimented with marking of Mottled Duskywings in June, 2015. We very slowly approached basking butterflies in the morning when the temperature was cool, then applied a spot of liquid acrylic paint (see paint details below) with a fine paintbrush to an upper wing surface, a technique inspired by Singer and Wedlake (1981). On the first day, two researchers successfully marked 10 individuals in an hour when the temperature was ~16°C; as the day warmed and the butterflies became more active they took evasive flights before we could get close enough to mark them. The second day was warmer (>18°C) and we only managed to mark one butterfly in an hour. When touched with the paintbrush, they flew only a few meters away from where they had been marked. We observed several individuals for several hours close to where they had been marked, suggesting that this technique of marking without handling had not disturbed them (see Kemp and Zalucki, 1999). However only a single spot of paint could be applied before the butterfly flew, and it was difficult to control the position, size, and shape of the marks. Consequently, it was difficult to determine their identities when we re-sighted them. That, coupled with the low numbers we could mark per day, made this method impractical.

In 2016 we set out to improve our technique so that we could mark duskywing butterflies at any ambient temperature while still minimizing damage and post-handling trauma. We experimented first (19-24 May) with Juvenal's Duskywings (*Erynnis juvenalis*) because they emerge earlier in the spring than Mottled Duskywings, are not endangered, and are relatively common at a site close to where we live. With the first set of butterflies (n=6), we removed them from the net with our fingers and adjusted their wings so all four wings were fully exposed. We then marked the dorsal wing surfaces with a unique pattern using dots of liquid acrylic paint in the 1-2-4-7 positions (*sensu* Ehrlich and Davidson, 1960) about halfway between the body and the outer wing edge, after which we released the butterfly. With this method we experienced several difficulties. The butterflies struggled while being held and usually lost many wing scales. One butterfly that struggled a lot during handling was injured to the extent that it could not fly properly when released; another lost a leg. When we released the butterflies they flew relatively short distances (2-5+ m), but we could not determine if they subsequently altered their behavior due to our handling. For several butterflies that received a spot of paint in the "4" location (on the leading edge of the hindwing), struggling while being held sometimes caused the forewing to shift posteriorly and touch the paint on the hindwing, resulting in the fore- and hindwings being stuck together. Those individuals flew away upon release, but presumably with reduced speed and agility.

The difficulties described above led to us to experiment with chilling Juvenal's Duskywings (n=36 butterflies in total) prior to handling and marking them. We maneuvered them from the net it into a small plastic jar (4 cm diameter; 4 cm height) without touching them. The jar was submerged in ice in a small cooler that we carried to the field site (Fig. 1). Chilling for 6 min caused them to cease body movements (n=10). We could then shake them gently into the palm of a hand, hold them with our fingers, and spread their wings for marking. Despite having been chilled for 6 minutes, some still struggled when held and we experienced the problems described above for unchilled butterflies, albeit to a reduced extent. Longer chilling duration (7 min (n=2); 8 min (n=1); 10 min (n=5)) helped to reduce movements of the butterflies and had no apparent negative effects on the butterflies, but slowed our marking process and increased the duration of butterfly basking upon release.

Our biggest improvement came with the realization that grasping the butterflies was not necessary if we did not mark the hindwings. Our final and highly successful technique involved chilling butterflies as described above for only 5 minutes. This was still sufficient to cause them to stop moving. They were then shaken into a hand (Fig. 1) and marked with 1-3 dots on the left forewing with combinations of dots that yielded identities of 1 to 6 and on the right forewing for numbers 10 to 60. With this method, we marked 18 Juvenal's Duskywings, of which only one shifted its wings and had its fore- and hindwings stuck together.

Subsequently we tested this method on Mottled Duskywings (6 June 2016), after having obtained the required permits for research with an endangered species. Again our method proved to be excellent: a team of three people caught and marked 22 individuals in 2.5 hours) with no apparent damage to them (Figs 2 and 3). After marking, we could usually carry the butterfly in cupped hands to the site of capture and place it on a twig where, after basking for a few minutes, it rejoined the population. One male Juvenal's Duskywing entered into a territorial chase with another male only 5 minutes after having been marked. By not marking the wing edges, even very worn individuals could still be identified when sighted on subsequent days (Fig. 4). There was no evidence that the marking process affected duskywing behavior.

For the marks, we purchased several liquid, non-toxic, water-based "fluid" acrylic paints made by Golden Artist Colors Inc. (Golden®, New Berlin, New York**).** These can be bought at art supply stores in a many bright colors in small volume containers (1 fl. oz; 30 ml) at a reasonable cost (http://www.goldenpaints.com/products/colors/fluid). For maximum visibility on the dark brown duskywings, the colors we selected were: Hansa Yellow Opaque (#2191), Pyrrole Red (#2277), Teal (#2369), Vat Orange (#2403), and Titanium White (#2380). We also created a light purple by diluting Dioxazine Purple (#2150) with an equal volume of Titanium White, and a lime green by diluting Permanent Green Light (#2250) with an equal volume of Titanium White to which we added a small amount of Hansa Opaque Yellow. Additional colors could be created by mixing 2 or more paints of different colors. These paints are easily applied with a fine brush, grass stem, or stick. The marks

Clockwise from top left:

Figure 1. Field set up for marking butterflies. After chilling a Juvenal's Duskywing on ice for 6 minutes in a cooler, it has been gently shaken into Gard Otis' hand while Jessica Linton is gently applying yellow acrylic paint marks.

Figure 2. Mottled Duskywing number "Orange #1" in the palm of a hand immediately after receiving a spot of Golden® "Vat Orange" paint in the number 1 location along the leading edge of the left forewing. While still chilled, the marked butterflies were carried back to the site of capture where they were gently transferred to a twig. After several additional minutes of basking they took flight and rejoined the population.

Figure 3. This Mottled Duskywing has received three spots of green acrylic paint on its left side along the leading edge of the forewing (number 1), trailing edge of the forewing (number 2), and base of the forewing (number 4), making this "Green #7" (i.e., 1 + 2 + 4 = 7). It is basking on one of the two host plant species utilized by Mottled Duskywings in Ontario, Prairie Redroot (*Ceanothus herbaceous*).

Figure 4. This very worn Juvenal's Duskywing, Red #2, in fresh condition when marked on 23 May, 2016, was photographed by a visitor to the reserve five days later, on 28 May. We avoided marks close to the outer edge of the wings because of the risk they would be lost through wing wear. The permanency of the acrylic paint marks is evident. Photograph taken by Julie Reid and used with her permission.

dry quickly, are permanent, and are highly visible. We could readily determine the identities of individuals (see Fig. 3) through close-focussing binoculars (focus range to ~2m) without recapturing them and potentially damaging them further.

To summarize, we developed a marking technique that met all of our objectives. By chilling the butterflies on ice for a relatively short period of time (5 min), we could mark them without the need to grasp them with our fingers to position the wings or to identify them, thereby minimizing damage to them. The marks, dots of fast-drying non-toxic water-based acrylic paints, were conspicuous and allowed for individual identification with binoculars. The chilling provided another benefit in that there was no evidence that the butterflies experienced any behavioral changes due to handling. These considerations were particularly important to us because the the Mottled Duskywing is rare and endangered in Canada. The brightly colored paints we used were easy to see, but they also made the butterflies more visible to predators, a violation of one of the assumptions of mark-recapture studies (Southwood, 1978). Less contrasting colors could be used that would overcome this concern. With three dots on each forewing (numbers 1-6 and 10-60), 42 individually recognizable individuals can be marked with each color. With our 6 colors, 252 butterflies could be uniquely marked. This is sufficient for uncommon species, but it would not be suitable for species with large populations. Our method is most applicable to butterfly species that bask dorsally and relatively close to the ground; lateral baskers would need to be marked identically on both hindwings if one wants to avoid recapturing them for identification (e.g., Ehrlich and Gilbert, 1963).

In reviewing the literature on mark-release-recapture studies, we were surprised to find that so many researchers continue to release butterflies immediately after marking without apparently considering the possible influences that handling may have on estimates of dispersal and survival. The evidence of such effects is widespread, having been documented in species belonging to many families (e.g., Papilionidae, Nymphalidae, Pieridae, Hesperidae). Although chilling of butterflies slows the marking process, doing so either before or after handling minimizes the effects of handling and marking. Chilling is strongly recommended except for those species that have been shown to be unaffected by netting, handling and marking.

ACKNOWLEDGEMENTS

Ivan Aguilar (2015) and Charlotte Teat (2016) assisted with fieldwork. Jenna Quinn and Allie Abraham of *rare* Charitable Research Reserve, Cambridge, Ontario, facilitated our study of Juvenal's Duskywings at the reserve. Lisbeth Sider provided advice on acrylic paints. We are grateful for funding and cooperation to JEL from the Ontario Species at Risk Stewardship Fund, Cambridge Butterfly Conservatory, and the Halton Region Conservation Authority; and to GWO from the Undergraduate Student Research Assistantship program, University of Guelph.

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Pat and Eric Metzler at the 2016 Lep Soc meeting. (Photo by James K. Adams)

Metamorphosis

Chris Grinter

Charles Bordelon, near Sanderson, Terrell Co., Texas, 2012, with a lot of Buffalo Gourd (*Cucurbita foetidissima*) in the background.

Charles W. Bordelon Jr., of Beaumont, TX, passed away at home, on Sept. 25, 2016, age 57, after a 9 month heroic battle with cancer. Charles recieved a B.S. degree from Lamar University, and pursued postgraduate studies. He worked as a cable installer and later for his father, Charles Sr. in publishing. I met him briefly in 1977, and in 1993, began a friendship and collaboration, on the study of the Lepidoptera of TX. He took over as the Lepidopterists' Society Season Summary coordinator for Texas in 1995, which he turned over to Mike Rickard in 2016. I joined with Charles on a survey of Lepidoptera of Big Thicket Nat'l Preserve in 1995 and later we worked together on similar surveys of Big Bend Nat'l Park, Guadalupe Mts. Nat'l Park, The Davis Mts, the Texas Hill Country, and Caprock Canyonlands, which continued through the early 2000s. These surveys concluded with self-published illustrated checklists. Checklists also were produced for the Lower Rio Grande Valley in 3 volumes, on Butterflies and moths. These were sold or distributed to many people. About 30 published articles also appeared in the News of the Lepidopterists' Society and Southern Lepidopterists' News. We also collaborated with others on the description of a new moth, Schinia varix. One butterfly, Poanes aaroni bordeloni, was named for him by the late Ron Gatrelle, and other patronyms will be coming soon. Our combined collections (Texas Lepidoptera Survey Research Coll.), is to be donated to the Florida State Coll. of Arthropods, McGuire Center, Gainesville,FL.

Charles (like many of us) began an interest in Lepidoptera at the age of 4, when his mother gave him a butterfly net, and never quit. He loved and excelled at field work, but also became very adept at moth and butterfly identification and scientific writing. He discovered many new records of Lepidoptera for Texas and many were also new for the USA. We went all over Texas, with occasional trips to Florida, Georgia, California, New Mexico, Arizona, Colorado, and Wyoming, and on several of these, we were joined by Susan Lee, Charles' wife. Our most memorable trip was a 2 week visit to Yasuni Nat'l Park in Ecuador, in 2002.

I have so many wonderful memories of our times together in the field. Some of these include: The night in Big Bend at Ladybird's cabin, when he nearly petted a fox and a skunk sat in my lap; the night of a million fireflies and many snakes at the Lance Rosier Unit in the Big Thicket; a trip with Susan, to New Mexico and Arizona, including a visit to the Grand Canyon Nat'l Park, where we saw a Condor; nights in cabins, with a roaring fire in Cloudcroft, NM and Pine Top AZ, where we found a paradise along the creek; the "catch" in Alamo, TX, when Charles netted a perfect *Historis odius*, sitting 20 feet up on the Alamo Inn. It was the first TX voucher. The last trip was in the summer of 2015, when we went to Colorado and Wyoming, where we found a particularly beautiful place at Columbine Pass, in the Uncompaghre Plateau, saw the Tetons, and found our first Hayden's Ringlets.

Charles enjoyed all aspects of nature and this included all kinds of insects, birds, reptiles, plants. He created, in his Beaumont backyard, a veritable oasis of native plants. He was well known by the Lepidoptera community and well liked, especially after you met him. He will certainly be greatly missed.

His parents pre-deceased him and he is survived by 1 brother and 2 sisters, many aunts, and cousins, and of course, Susan Lee, his wife. [*Submitted by Ed Knudson, Houston, TX.*]

Hodebertia testalis in Florida

Continued from p. 170

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Sesiid pheromone attractant information update

William H. Taft, Jr.

Research Contributor, A. J. Cook Research, Michigan State University, 1430 W. Locher Road DeWitt, MI, 48820
billandgussie@earthlink.net

In the Lepidopterists' Society publication titled: Basic techniques for observing and studying moths and butterflies by William Winter (2000), the author included a number of specific subject appendices including Appendix H – Sesiid Pheromones. In Table 3 of Appendix H, titled "Pheromones and the sesiid species they attract", there were 54 sesiid species that had no chemical attractant data associated with them. In an effort to fill in these gaps, I have compiled a list of North American sesiid species with new or updated pheromone attractant data.

A number of these attractants have not been developed for pest monitoring or have any direct commercial application, however custom pheromone blends can be purchased from several pheromone suppliers including Pherobank located in the Netherlands (**www.pherobank.com**) and Alpha Scents in the United States (**www.alphascents.com**).

Table 3. Supplement species information to Table 3 of Appendix H in W. Winter, 2000

Species	pheromone attractant $	special notes
Zenodoxus mexicanus	ZZA****	Greater peachtree commercial lure
Z. palmii	ZZA/EZA 50:50	Alphascents custom lure
Cissuvora ampelopsis	EZOH/ZZOH 50:50	P. robiniae commercial lure
Paranthrene robiniae	EZOH/ZZOH 50:50	Alphascents commerial blend
P. simulans	EZ 2-13A/ZZA 50:50	Alphascents custom lure
Euhagena emphytiformis	EZOH ***or ZZA	
Vitacea scepsiformis	EZA/EZ 2,13A/ZE 3,13A 88:6:6	Alphascents EZA - custom blend
Melittia cazabaza	EZA***	Lesser peachtree borer lure
M. snowii	EZ 2,13OH/ZZA 99:1	Pherobank custom lure
M. gloriosa	EZ 2,13 OH/EZ 2,13 A/ZZA/Z13A 94:2:2:2	Pherobank custom lure
Sesia apiformis	ZZOH/EZ 2,13 Octadecadienal 40:60	Pherobank commercial blend
Tirista argentifrons	ZZA***	Greater peachtree commercial lure
Synanthedon arctica	EZ 2-13A/ZZA 99:1	Graperoot borer lure
S. bolteri	EZ 2-13A/ZZA 99:1	Graperoot borer lure
S. geliformis	ZZA or ZZA/ZZOH (50:50)****	
S. helenis	EZ 2-13A/ZZA 50:50	Alphascents custom lure
S. proxima	EZA/EZ 2,13A/ZE 3,13A 88:6:6	Alphascents EZA - custom blend
S. polygoni	ZZA or ZZA blend	Greater peachtree commercial lure
S. sigmoidea	EZ 2-13A/ZZA 99:1	Graperoot borer lure
Palmia praecedens	ZZA or ZZA blend	Greater peachtree commercial lure
Carmenta apache	EZ 2-13A/Z 13A 96:4	P. asilipennis blend
C. arizonae	ZZA/EZA***	
C. auritincta	EZ 2-13A/ZZA 99:1	Graperoot borer commercial lure
C. ogalala	Scentry L103*	Scentry commercial lure
C. phoradendri	ZZA/EZA blends***	
C. prosopis	EZA/ZEA/ZZA/EEA EZ 2,13A 95:2:1:1:1	Pherobank custom lure
C. querci	ZZA or ZZA blend	Greater peachtree commercial lure
C. rubricincta	EZ 2-13A/ZZA 50:50	Alphascents custom lure
C. tecta	ZZA or ZZA blend	Greater peachtree commercial lure
C. wellerae	EZ 2,13OH/ZZA 99:1	Pherobank custom lure
Penstemonia clarkei	ZZA or ZZA blend	Greater peachtree commercial lure
Alcathoe autumnalis	ZZA***	Greater peachtree commercial lure
A. pepsioides	ZZA/EZA 50:50	Alphascents custom lure
A. verrugo	ZZA/EZ 2,13A/EZ 2,13OH**	Moths trapped in 2015
Hymenoclea palmii	ZZA or ZZA blend	Greater peachtree commercial lure

$ Best or reported lure
* Chuck Harper report
** Kelly Richers tentative comment
*** Texas Lepidoptera Atlas, Vol. VII, SESIODEA, Knudson & Bordelon
**** Tropical Lepidoptera Aug. 1993, Vol. 4, Supplement 4, Brown and Mizell
ZZA = Z,Z 3,13 A; EZA = E,Z 3,13 A; ZZOH = Z,Z 3,13 OH; EZOH = E,Z 3,13 OH

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Holarctic Butterflies -- Part 2

George O. Krizek[1], Kyle E. Johnson[2], Paul A. Opler[3], and Steven J. Cary[4]

[1]2111 Bancroft Place NW, Washington, D.C. 20008
[2]445 Russell Labs, University of Wisconsin-Madison, 1630 Linden Drive, Madison, WI 53706-1598
kejohnson4@wisc.edu
[3]P. O. Box 2227, Loveland, CO 80539 **paulopler@comcast.net**
[4]202 Solana Drive, Santa Fe, NM 87501 **sjcary@earthlink.net**

In the Fall 2015 issue of the News of the Lepidopterists' Society (57:3, pgs. 103-107) we presented a photoessay of Holarctic butterflies, sensu stricto; here we continue with a supplement.

The first set of images show additional species which appear to be (based on present knowledge) clearly Holarctic species (*Hesperia comma, Parnassius eversmanni, Colias nastes, Boloria improba, B. chariclea, Erebia magdalena, E. mackinleyensis, E. pawloskii, Oeneis melissa,* and *O. alpina*). After these is one species (*Vanessa virginiensis*) which could be marginally considered Holarctic since it occurs naturally in the Canary Islands and often wanders into southwestern Europe (Higgins & Riley, 1970). Lastly are a few species formerly considered Holarctic (*Plebejus argyrognomon, Boloria titania, Erebia youngi,* and *Oeneis norna*) but are now considered to be solely Palearctic. Some of the above (and others) warrant further taxonomic reearch, so our perceptions of what species are actually Holarctic may still change with time.

We thank Matjaž Černila, Jurij Rekelj, Per-Olof Wickman, and Christer Wiklund for kindly sharing some of their images. Dr. Alois Pavlicko from the Czech Repulbic helped us get in touch with several Eurasian photographers, and deserves our thanks.

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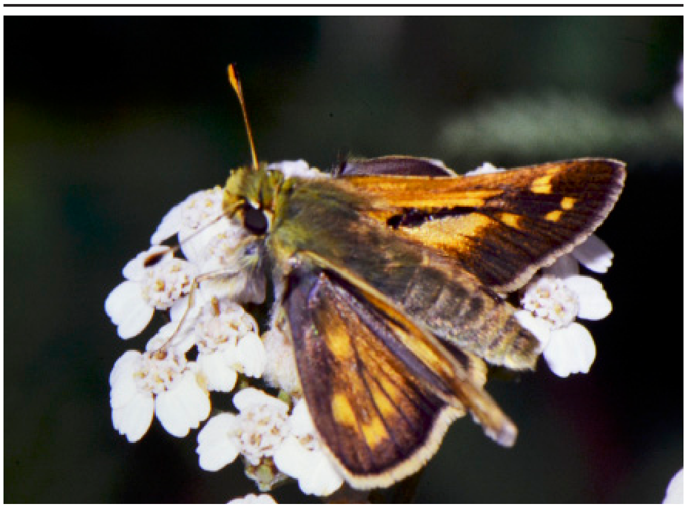


***Parnassius eversmanni*,** NE SIBERIA, far E. Magadanskaia Oblast, Omsukchanskii raion, Omsukchanskii khr.,Kapranovskii pass/1300m/env., road to Osadochnyi village, 62°09'39" N/ 155°17'23" E, 1000-1200m, June 30, 2006, Photo by Matjaž Černila

***Colias nastes*,** SWEDEN, Abisko, Photo by Crister Wiklund

To left: ***Hesperia comma*,** SWITZERLAND, Zermatt, August 3, 2001, Photo by George Krizek.

Boloria improba**,** SWEDEN, Darfallahku, close to Nikkaluokta, Lapland, July 4, 2011, Photos by Per-Olof Wickman

Boloria improba**,** ALASKA, Brooks Range, Dalton Hwy., Galbraith lake, East side of lake (Dry Tundra), 68°26.545'N/ 149°20.440'W , 1050m, June 22, 2015, Photo by Jurij Rekelj

Boloria chariclea**,** NEW MEXICO, Wheeler Peak, Taos Co., July 17, 2007, Photo by Steven J. Cary

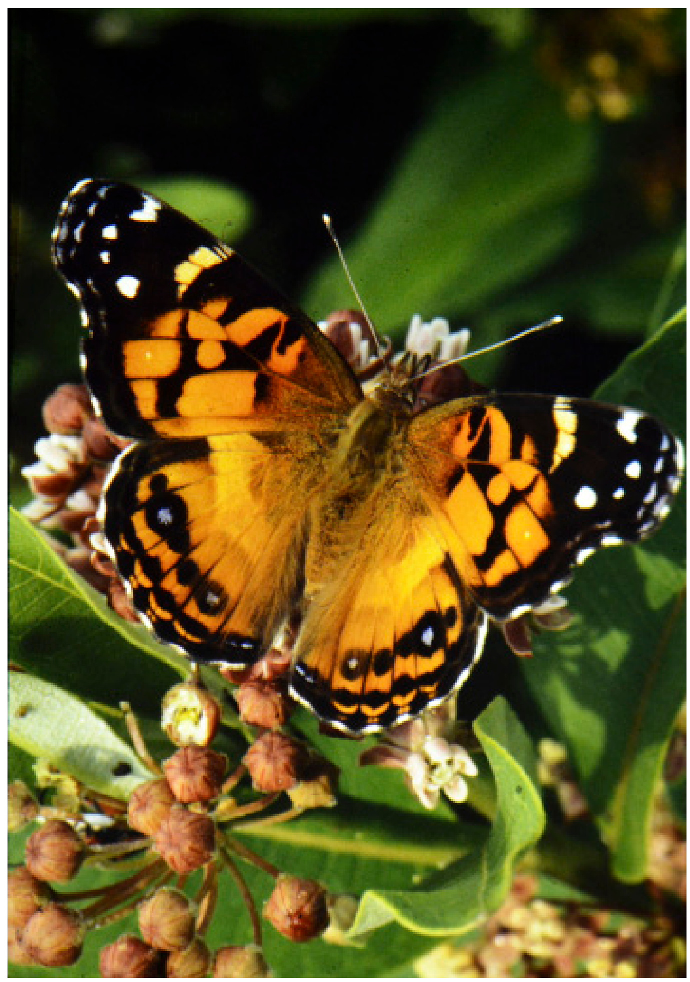
Erebia magdalena**,** UTAH, Uintah Mountains, July 15, 2014, Photo by Steven J. Cary

Erebia mackinleyensis**,** ALASKA, Brooks Range, Dalton Hwy., Galbraith lake, East side, mountain peak (rocky tundra), 68°25.860'N/149°18.725'W, 1360m, July 2, 2015, Photo by Jurij Rekelj

***Erebia pawlovskii*, **WYOMING, Clay Butte., July 22, 1996, Photos by George Krizek

***Oeneis melissa*, **NEW MEXICO, Truchas Peak, Ariba Co., June 28, 2014, Photo by Steven J. Cary

***Oeneis alpina*, **ALASKA, North Slopes, Dalton Hwy., Hill by the Oksrukuyik Creek, (Dry Tundra), 68°40.892'N / 149°05.502'W, 800-820m, June 23, 2015, Photo by Jurij Rekelj



***Vanessa virginiensis*, **MARYLAND, Potomac, June 20, 1981, Photo by George Krizek

Plebejus argyrognomon**,** CZECH REPUBLIC, Praha, July 10, 2015, Photos by George Krizek

Boloria titania**,** ITALY, Courmayeur, August 6, 1986, Photo by George Krizek

Erebia youngi**,** NE SIBERIA, Far E. Magadanskaia Oblast, Khasinskii okrug, gory Del-Urekchen, Karamken pass, 60°19' N/151°11.5' E, 800-900m , July 7, 2006, Photo by Matjaž Černila

Oeneis norna**,** SWEDEN, Abisko, July 1985, Photo by Crister Wicklund

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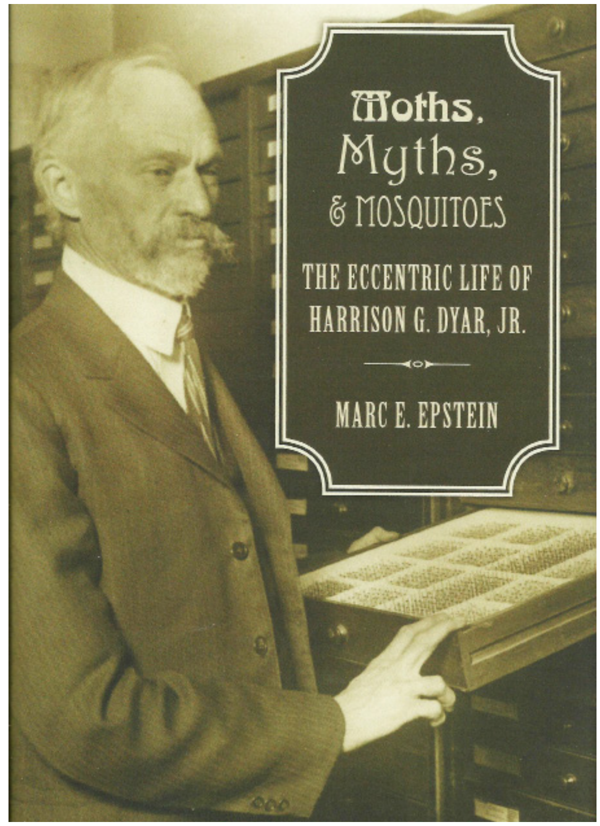
Membership Updates

Chris Grinter

Includes ALL CHANGES received by 2 November 2016. Direct corrections and additions to Chris Grinter, **cgrinter@gmail.com.**

New Members: *Members who have recently joined the Society, e-mail addresses in parentheses.. All U.S.A. unless noted otherwise.*

Erin R. Barbeau: [redacted by request] (erinreneebb@gmail.com)
William Beck: 15660 N Roadrunner Ridge Lane. Tucson, AZ 85739 (billbeck001@gmail.com)
Dylan Cipkowski: 187 Old Queechy Rd, NY 12029 (dacipkowski@gmail.com)
Patricia Esther Corro: Universidad Panama, Programa de Maestría en Entomología, Vicerrectoría de Investigación y Postgrado, Estafeta Universitaria 0824. Panama City, PANAMA. (estherpatricia04@gmail.com)
Ansley Curry: 212 Governors Court. Cartersville, GA 30121 (aecurr3488@ung.edu)
Marsha E. Dwyer: [redacted by request] (mdwyer@aggies.ncat.edu)
Makani Layne Fisher: 1127 North 140 East. Orem, UT 84057 (makanifisher@comcast.net)
Patrick Grof-Tisza (Ph.D.): 1130 L. Street. Davis, CA 95616 (pgroftisza@ucdavis.edu)
Craig Hensley: 3350 Park Road 31. Spring Branch, TX 78070 (craig.hensley@tpwd.texas.gov)
Linda Jane Hunt: 10322 Twinedew Place. Columbia, MD 21044 (Raven10322@hotmail.com)
Lucas Kaminski (Ph.D.): Av. Ijui, 133, Ap. 26. Porto Alegre RS BRAZIL (lucaskaminski@yahoo.com.br)
Akhlesh Lakhtakia: 1811 Red Lion Drive. State College, PA 16801 (akhlesh@psu.edu)
Taryn Lewczyk (Ph.D.): 2019 Oleander Street, Apt 3. Baton Rouge, LA 70806 (tarynxvii@gmail.com)
Eleanor McCabe: 29 Spring Street, Apt 6. Newmarket, NH 03857 (ellieannmccabe@gmail.com)
Karen Mintiens: 1916 Stockton Road. Phoenix, MD 21131 (karenmints@comcast.net)
Carmen Mo: [redacted by request] (carmen@nevada.unr.edu)
Nadya Muchoney: Biology Dept, MS 0314, Univ. of NV, Reno. 1664 N. Virginia St. Reno, NV 89557 (nmuchoney@nevada.unr.edu)
David Lee Myers: 120 Commercial Street. Astoria, OR 97103 (david@DavidLeeMyersPhoto.com)
Franz Pühringer (M.D.): Häusern 4 St. Konrad. Upper Austria 4817 AUSTRIA (f.puehringer@sesiidae.net)
Florian Radke: 183 Stanhope Street. Brooklyn, NY 11237 (florian@florianradke.com)
Elizabeth M. Rothwell: [redacted by request] (emr342@nau.edu)
Frederico Riva: [redacted by request] (friva@ualberta.ca)



Bryan Wesley Shields: 2265 South 58th Street. Fennville, MI 49408 (bwshields@frontier.com)
Steven Taylor (Ph.D.): 1116 W William Street. Champaign, IL 61821 (ceuthophilus@gmail.com)
Jean Francois Treves: 7 Sweetwater Lane. Hilton Head, SC 29926 (treves.u@gmail.com)
Reginald Webster (Ph.D.): 24 Mill Stream Drive, Charters Settlement, NB CANADA E3C 1X1 (reginaldwebster@rogers.com)
Asha Wijerathna: [redacted by request] (wijerath@ualberta.ca)
Wade Lee: [redacted by request] (wlee801@gmail.com)

Address Changes: *All U.S.A. unless otherwise noted.*

Charles M. Barksdale (Ph.D.): 151 Crim Lane. Elgin, TX 78621 (cmbarksdale@cox.net)
Teá Kesting-Handly: 26 Arborway. Jamaica Plain, MA 02130 (tea.kestinghandly@gmail.com)
Ken Stead: 16321 Kelly Woods Drive, Apt 186. Fort Myers FL 33908 (kstead@sympatico.ca)
Mamoru Watanabe (Ph.D.): 1-4-1 Kuriki, Isogo, Yokohama, Kanagawa 235-0041 JAPAN (papilio-platycnemis@nifty.com)

Correction:

Susan Reigler: Dept. of Biology, Indiana University, Southeast 4201 Grant Line Rd. New Albany, IN 47150 (sreigler@ius.edu) - originally published state as NY, corrected to IN.

Book Review

Marc. E. Epstein. 2016. *Moths, Myths, and Mosquitoes: the Eccentric Life of Harrison G. Dyar, Jr.* Oxford Univ. Press, New York, New York. xxvi+325 pp. $39.95. (cover to left)

If there was ever a story worthy of its own soap opera, this is it. The vibrant life of Harrison Gray Dyar, Jr. (1866-1929), known as "Harry" to his family, fused together the experiences of at least five individuals: entomologist, essayist, engineer, genealogist, and real estate tycoon. While his activities spawned fanciful myths, his reality reads like folklore. The term "eccentric" understates the essence of this colorful character, who is richly revealed in this new book. Epstein clearly lays out the purpose of publishing Dyar's biography: "I hope to tip the balance a bit more toward his bequest to science, while chronicling his illustrious personal life and complexities." In this he certainly succeeds.

Dyar's father – one of twelve siblings – was an inventor who, among other things, developed a type of telegraph that influenced the later electrical telegraph by Samuel F. Morse (of Morse code fame). Dyar's aunt conducted a séance for President Abraham Lincoln and his wife, Mary Todd Lincoln. From a young age, Dyar Jr. was exposed to the spiritualist movement, which alleged that the dead have the desire to communicate with the living. Not surprisingly, this activity contributed to an unusual childhood for Dyar, prompting him to seek an escape within the study of natural history, especially insects. He rapidly rose in prominence and eventually authored over 650 publications on Lepidoptera, in which he described over 3,000 genera and species. He also described 48 genera and 628 species of flies, and 28 species of wasps. The hallmark of his career was the study of the larval stages of insects, leading to his revolutionary work in mosquito control, as well as his remarkable "Law of Geometric Growth," which assumes that geometric growth occurs between successive stages (instars) of Lepidoptera larvae. Dyar is most often remembered by lepidopterists for his voluminous work "A list of the North American Lepidoptera and key to the literature of this order of insects," which was published in early 1903.

Unfortunately, rivalries with fellow entomologists dogged many of Dyar's endeavors, though most of it was self-wrought. Dyar had a quick temper and could be extremely condescending, both in writing and in person. He preferred to get along only with a select few. He fought authority and abhorred bureaucracy, prompting him to criticize management of the Smithsonian Institution, where he worked (at times without pay) for many years. He openly quarreled with other entomologists, such as John B. Smith, whose feud with Dyar is well known in the annals of historical entomology. Epstein finally debunks the popular myth involving Smith and the moth name "*dyaria*" (pronounced "diarrhea"), which in reality was not intended as a derogatory swipe at Dyar (nor was the name proposed by Smith!). Dyar had an intense passion for the study of insects, but his judgement was sometimes questionable. For example, he once attempted to establish an Asian limacodid moth around Washington, D.C., which was mercifully unsuccessful. None of this, however, compares to the poor judgement he exhibited with regard to his love life and his unusual hobby of digging elaborate tunnels beneath Washington, D.C.

Accused of bigamy, the saga of Dyar's two marriages is extraordinarily complex, involving adultery, fraud, and deceit. These escapades are meticulously deconstructed in the book, underscoring Dyar's misdirected genius. The tale of the tunnels is just as captivating, explaining how Dyar began digging them for fun and exercise, excavating intricate labyrinths for a decade. Once thought to have been constructed by bootleggers or war spies, it was a complete surprise when authorities discovered that the tunnels were the work of a peculiar entomologist who dug them as a pastime. Some tunnels were lined with concrete and brick, and illuminated with lights. Epstein suggests that Dyar's burrowing habit was driven by a compulsion to finish whatever he started, or it perhaps calmed him in some cathartic fashion. In the end, Dyar suffered a stroke while at work at the Smithsonian, just before his 63rd birthday. His tunnels, which continued to collapse into the 1950s, ultimately brought his two families together: an inspiring tribute to the odd mastermind who created them.

Epstein's writing is clear and enthralling, drawing the reader from one surreal event in Dyar's life to another. The book is loaded with figures and maps (some in color) that trace Dyar's life and times. The acknowledgements are extensive, showing the level of research and scholarship that went into the book's production. The chapters are arranged in logical fashion, though there is a large amount of crossover due to the subject matter. A fitting epilogue includes some final thoughts about Dyar's marriages, entomological value, and final resting place. Also included is an extensive chronology of Dyar and his family, listing all the relevant happenings within the context of world events. Exhaustive notes, which provide additional facts by chapter, are worth wading through, as they contain an incredible amount of valuable information.

I found few aspects of this book to criticize, beyond a handful of typos (such as a reference to Plate 9 on page 74, when it should be Plate 8), which are of minor significance. Some readers may be distracted by the apparent disregard for chronology in some places, but this seems unavoidable. Some minute details may appear unnecessary, but the astute reader will understand their inclusion within the larger portrait of Dyar's life. I highly recommend this book to anyone interested in history, entomology, or nature. If you are looking for a biography that puts reality television to shame, search no further.

John V. Calhoun, **bretcal1@verizon.net**

Where have all the butterflies gone? Well, many of them are still here!

Jeff Pippen

101 Forest Oaks Dr., Durham , NC **jeffpippen9@gmail.com**

Many comments I read on social media reflect sentiments such as "where are all the butterflies?", or "why am I not seeing as many butterflies as I did last year?" And no doubt some of the perceived decline in butterflies is real, due in part to habitat loss, land use changes, and, in some areas, mosquito spraying. Scott Hoffman Black's article in a recent issue of *Wings* (2016) and reprinted in the *News of the Lepidopterists' Society* (2016) raises some critically important issues concerning many likely declining butterfly populations in North America. On the other hand, I often wonder if *some* of the "doom and gloom" sentiments, usually posted early in the summer season on social media, are simply a reflection of the fact that butterfly population numbers of many species tend to build as the summer progresses. Hence, later in the season there are more butterflies (in terms of sheer numbers) than there are earlier in the season. I wonder if some of those late season higher numbers are lingering in the minds of those who lament "fewer butterflies" on social media when the next season rolls around, especially when these posts appear before late summer. In this article, I present a couple of examples of experiences this past season where butterflies abounded.

Figure 1. Pink and purple flowers like this Joe-Pye Weed (*Eutrochium fistulosum*) in the North Carolina mountains attracted hundreds of butterflies including this Aphrodite Fritillary (*Speyeria aphrodite*) this past August.

This past August 23-24, Harry LeGrand Jr. and I headed to the North Carolina mountains for two days of butterflying. Despite a sunny forecast, we fought clouds and dew-draped vegetation both mornings so butterfly encounters were low. But perseverance paid off as the afternoon sun came out. By the end of the second day, we tallied over 1800 individual butterflies comprising 53 species in and around the meadows, wetlands, and roadsides of Caldwell, Watauga, Wilkes, Ashe, and Alleghany Counties. The big nectar attractions were the scores of Joe-Pye Weed (*Eutrochium*) (figure 1), Ironweed (*Vernonia*) (figure 2), and Elephantsfoot (*Elephantopus*) that were covered with leps. It was a great trip and butterflies were "everywhere" once the ambient conditions improved! Appendix 1 lists the species from this adventure.

Figure 2. A Meadow Fritillary (*Boloria bellona*) dives into Ironweed (*Vernonia sp.)* for nectar.

My second example involves the annual Durham NC Butterfly Count that I have been compiling since 1999. After reading several comments expressing concerns over a lack of butterflies in the eastern US, including my residential state of North Carolina, I thought I'd look at some actual data based on the Durham Butterfly Count. Figure 3 shows the total number of butterflies tallied annually in August on the Durham Count. The linear trendline is nearly flat, indicating no significant change over a 17-year period. To account for varying effort from year to year, figure 4 shows the number of butterflies tallied per field party hour. Again the trendline is relatively flat, perhaps even trending toward an increase in butterfly sightings. In fact, for the past two years, the observers on the Durham count have found higher than average numbers of butterflies. Our 17-year average is 3839 individuals (125/party hour). In 2015 we totaled 4155 butterflies (143/party hour) and in 2016 we racked up an impressive 5096 individuals (164/party hour)!

We are all aware of myriad challenges that butterflies and other denizens of natural habitats face in the coming years. Fortunately, however, the stories are not all doom and gloom, and great opportunities still exist for adventurers seeking butterflies!

Literature Cited

Black, Scott Hoffman. 2016. "North American Butterflies: are once common species in trouble?" *Wings* 5-9.

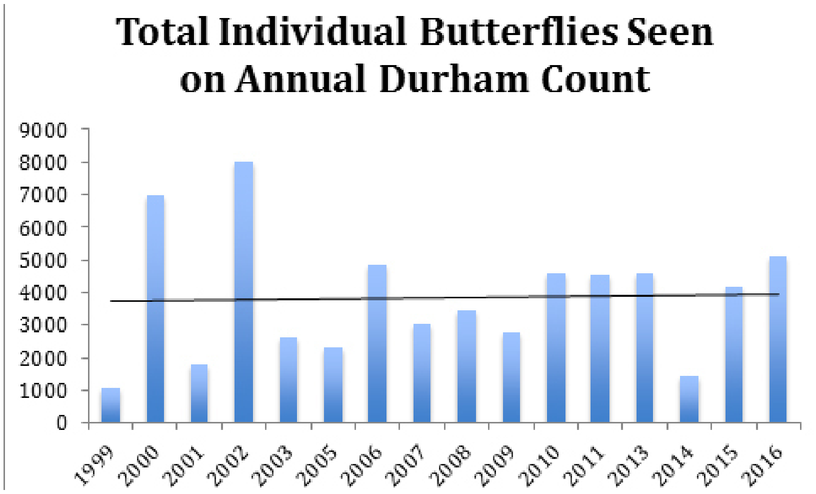


Figure 3. A linear trendline shows that the average number of butterflies observed during the annual Durham Butterfly Count has been stable since inception of the Count in 1999. The Count was weathered out in 2004 and 2012.

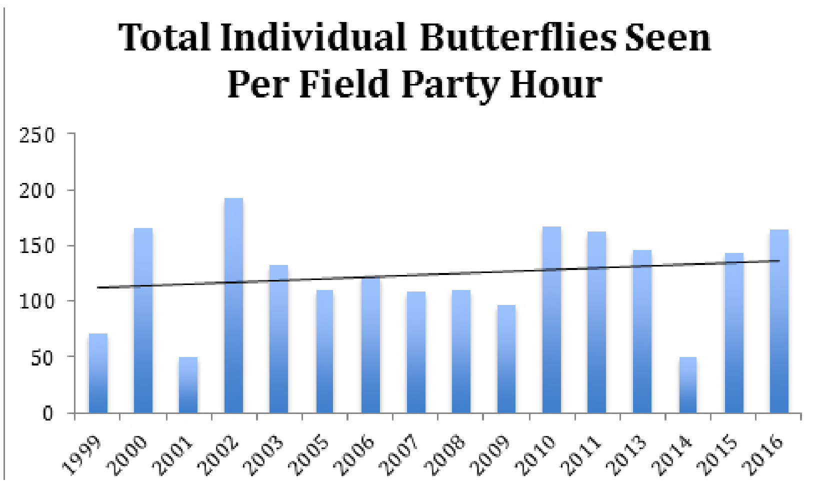


Figure 4. Since the Durham Count started in 1999, the number of individual butterflies observed per field party hour has been fairly constant.

Appendix 1

LeGrand and Pippen sighted the following butterflies 23-24 August in the northwestern NC mountains.

Pipevine Swallowtail, *Battus philenor*
Black Swallowtail, *Papilio polyxenes*
Spicebush Swallowtail, *P. troilus*
Eastern Tiger Swallowtail, *P. glaucus*
Cabbage White, *Pieris rapae*
Clouded Sulphur, *Colias philodice*
Orange Sulphur, *C. eurytheme*
Cloudless Sulphur, *Phoebus sennae*
Little Yellow, *Pyrisitia lisa*
Sleepy Orange, *Abaeis nicippe*
Gray Hairstreak, *Strymon melinus*
White M Hairstreak, *Parrhasius m-album*
Eastern Tailed-Blue, *Cupido comyntas*
Summer Azure, *Celastrina neglecta*
Variegated Fritillary, *Euptoieta claudia*
Great Spangled Fritillary, *Speyeria cybele*
Aphrodite Fritillary, *S. aphrodite*
Diana Fritillary, *S. diana*
Meadow Fritillary, *Boloria bellona*
Silvery Checkerspot, *Chlosyne nycteis*
Pearl Crescent, *Phyciodes tharos*
Eastern Comma, *Polygonia comma*
Question Mark, *P. interrogationis*
Red Admiral, *Vanessa atalanta*
American Lady, *V. virginiensis*
Common Buckeye, *Junonia coenia*
Red-spotted Purple, *Limenitis arthemis astyanax*
Viceroy, *Limenitis archippus*
Northern Pearly-eye, *Lethe anthedon*
Creole Pearly-eye, *L. creola*
Appalachian Brown, *L. appalachia*
Carolina Satyr, *Hermeuptychia sosybius*
Common Wood-Nymph, *Cercyonis pegala*
Monarch, *Danaus plexippus*
Silver-spotted Skipper, *Epargyreus clarus*
Golden Banded-Skipper, *Autochton cellus*
Hoary Edge, *Achalarus lyciades*
Horace's Duskywing, *Erynnis horatius*
Zarucco Duskywing, *E. zarucco*
Common Checkered-Skipper, *Pyrgus communis*
Least Skipper, *Ancyloxypha numitor*
Clouded Skipper, *Lerema accius*
Fiery Skipper, *Hylephila phyleus*
Peck's Skipper, *Polites peckius*
Crossline Skipper, *P. origenes*
Southern Broken-Dash, *Wallengrenia otho*
Little Glassywing, *Pompeius verna*
Sachem, *Atalopedes campestris*
Delaware Skipper, *Anatrytone logan*
Zabulon Skipper, *Poanes zabulon*
Dun Skipper, *Euphyes vestris*
Lace-winged Roadside-Skipper, *Amblyscirtes aesculapius*
Ocola Skipper, *Panoquina ocola*

Announcements:

Nominations for Karl Jordan Medal 2017

The Karl Jordan Medal is an award in recognition of published original research on the Lepidoptera that may be given biennially by the Lepidopterists' Society at the Annual Meeting. Nominations of publications must be of exceptional quality and focus on the morphology, taxonomy, systematics, biogeography and natural history of Lepidoptera. The criteria (*J. Lep. Soc.*, 26: 207-209) emphasize that the work may be based on a single piece of research or on a series of interrelated works. These publications must be at least three but not more than 25 years old. The latter is to assure that the awarded work(s) have been used by the scientific community and stood the test of time. The Jordan Medal is not intended to be a career award for service rendered to the study of Lepidoptera inasmuch as the Society already has such an award, Honorary Life Member. In addition, the nominee does not have to be a member of the Society in order to qualify. A complete list of lepidopterists who have previously received the Karl Jordan Medal is available on the Lepidopterists' Society website http://www.lepsoc.org/society_news.php.

Formal nominations for the Karl Jordan Medal will be accepted from any member of the Lepidopterists' Society and should be sent to Dr. Jacqueline Y. Miller, McGuire Center for Lepidoptera and Biodiversity, Florida Museum of Natural History, University of Florida, P.O. Box 112710, Gainesville, FL 32611-2710 or via email (jmiller@flmnh.ufl.edu). Please include a list of the specific publications for which the candidate is nominated, a support letter outlining the significance of the work(s), and if possible, a copy of the nominee's curriculum vitae, no later than 15 February 2017.

The Joan Mosenthal Dewind Award -- Now accepting applications

Joan Mosenthal DeWind was a pioneering member of the Xerces Society. A psychiatric social worker by profession, she was also an avid butterfly gardener and an accomplished amateur lepidopterist. Her contributions of time, organizational expertise, and financial support were essential to the growth and success of the Xerces Society over the past 30 years. In Joan's memory, Bill DeWind established this student research endowment fund. Since 2003, the Xerces Society administers two $3,750 awards for research into Lepidoptera conservation.

Details -- Submission Requirements:

The DeWind Awards are given to students who are engaged in research leading to a university degree related to Lepidoptera conservation and who intend to continue to work in this field. All proposals must be written by the student researcher. Proposed research should have a clear connection to Lepidoptera conservation and must be completed within one year from receiving funds. Applicants may be graduate or undergraduate students; however, please note that all but one awardee, to date, have been pursuing graduate research. Applications from countries outside the United States will be considered but must be written in English and international applicant work cannot involve work in the United States.

Submission Deadline: Monday, January 2, 2017, at 11:59 PM PDT. Award winners will be announced by March 31, 2017, with the awards given by May 2017.

Instructions and format: All proposals must be submitted by email to dewind@xerces.org. The proposal should be attached as a single file in PDF format. The subject line of the email should read "DeWind Award Proposal 2017."

Proposal Format: (all text should use 12 pt font and one inch margins)

1. Cover page (1 page)
    a. Title. List the title in Bold.
    b. Contact information. Provide the name and contact information for the applicant and his or her major advisor. Include institutional affiliations, complete mailing address, and country. Also provide an email address and telephone number (include country code if outside the United States).
    c. Abstract. Include a project summary immediately following the title and contact information. The summary should be limited to 100 words and should not exceed one paragraph.

2. Proposal body (2 pages). Begin with a clear statement of the problem or objectives, follow with a clear methods section, and end with a substantial conclusion. The proposal should include a discussion of potential conservation applications and results, and what products, if any, will result from this work.

3. Additional information. On separate pages, please include all of the following information: cited literature, detailed project budget, project timeline, and a short (2 pages or less) CV. It is the goal of the DeWind Award that the funds be used for direct research-related expenses; overhead and/or administrative fees are considered ineligible.

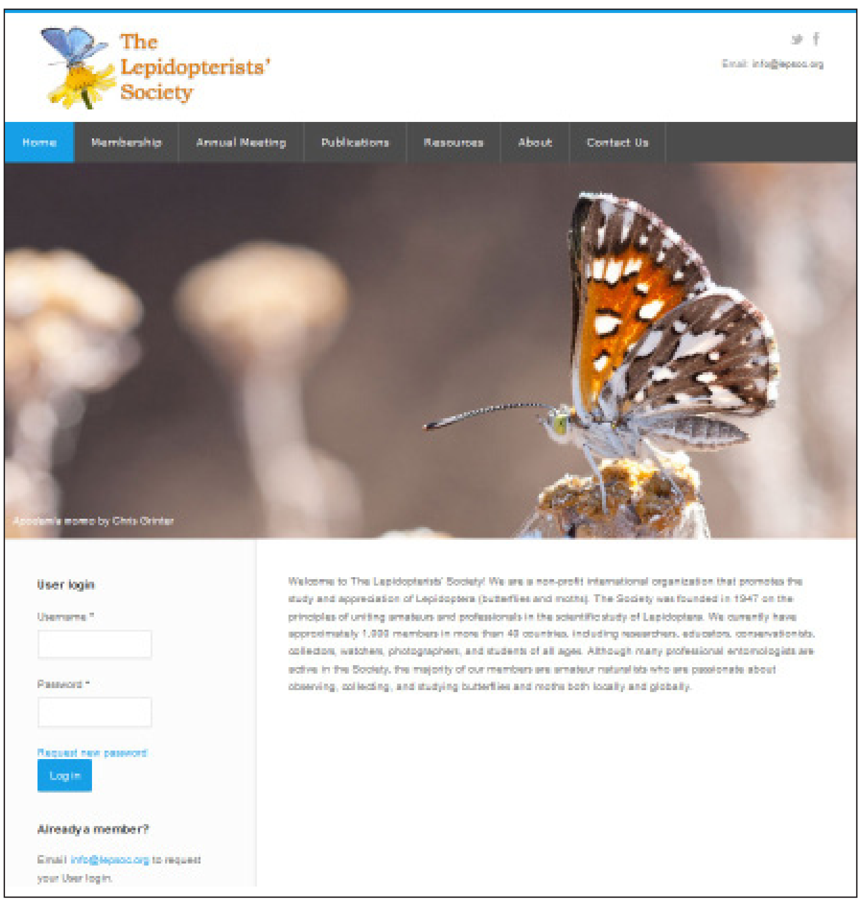
4. Please include all of the materials as a single attachment. No other attachments or supporting materials should be included.

For more information and to view previous winners visit: **http://www.xerces.org/joan-dewind-award/**

LepSoc attends the XXV International Congress of Entomology and releases a new website!

The Lepidopterists' Society sponsored a non-profit exhibitor's booth at the XXV International Congress of Entomology (ICE2016), which also served as the 2016 annual meeting for the Entomological Society of America (ESA). The meeting was the largest entomological gathering in history, with 6,682 registered participants from 102 countries and 5,396 formal presentations. Needless to say, the number people, size of the venue, and thousands of presentations was overwhelming! The Lepidopterists' Society booth was staffed Sunday through Friday by Christi Jaeger, Hanna Royals, and Todd Gilligan. We also received lots of help from Charlie Covell and Brian Scholtens, who graciously took time out of their meeting schedule to give the others a break. We had lots of LepSoc-branded freebies, and handed out 300 bags, nearly 1,000 pens, 500 notepads, more than 1,000 of the newly designed LepSoc brochures, and several hundred extra Journal and News copies. We also gave away 30 free student membership cards, made possible by our "sponsor a student" membership drive (which will continue for 2017). The most popular items at the booth were the butterfly and moth displays provided by Jackie Miller and the McGuire Center for Lepidoptera & Biodiversity. Jackie also helped by transporting the displays and boxes of publications and materials to and from Gainesville before and after the meeting. Overall, the booth was a great success, and during the course of the meeting we were visited by several thousand people, easily the most exposure the Society has to the worldwide entomological community.

ICE2016 booth volunteers. Todd Gilligan, Hanna Royals, Brian Scholtens, and Christi Jaeger (apparently searching for morphos?). Not shown, Charlie Covell and Jackie Miller.



On September 1, in preparation for the ICE 2016 meeting, the LepSoc released a new version of their website, **www.lepsoc.org**. The new site was designed and coded (entirely for free!) by Todd and Ella Gilligan over the past year using a Drupal content management system. Significant changes over the old site include: new logo and consistent branding with new brochures and meeting displays; new design allows for automatic scaling and functionality across devices; new online membership database that is updated in real time with changes, new members, renewals, etc.; member login that allows access to Journal content hosted by BioOne; and many back end changes that will allow for compatibility with future updates, web browsers, and devices. When first visiting the new website , you will notice a "User login" box on the left side of the screen. Logging into the website is NOT required to view any content unless you are attempting to update your membership information or access recent Journal articles hosted at BioOne. Member logins are not active by default, so if you would like a user login for the site, email **info@lepsoc.org** with a request. We hope you enjoy the new LepSoc website!

The 66th annual meeting of the Lepidopterists' Society will be held from Sunday July 30 - Tuesday August 1, 2017 at the Marriott University Park in Tucson, Arizona which is within walking distance to the University of Arizona Insect Collection (**http://www.uainsectcollection.com**). This event is hosted by the Department of Entomology in the College of Agriculture and Life Sciences, University of Arizona. The University of Arizona has recently been ranked the top entomology program in the United States with particular specialties in Biodiversity, Integrative Pest Management and Pollination.

We are looking forward to contributed papers, and special symposia will be for Arizona Lepidoptera, biodiversity (graduate student research), and pollination. Organized activities such as visiting and working in the collection, learning how to be more involved with the Lepidopterists' Society, and guided collecting and photography adventures await.

Online registration and abstract submission is open at **https://lepsoc2017.eventbrite.com.** Registration includes facility fees, snacks and the BBQ. Additional tickets for the banquet buffet are available for purchase on the website. Tucson has an airport near town with shuttle and taxi services to the conference location. Hotel accommodations can be made at the Marriott University Park for a discounted rate (**https://goo.gl/CMZrpx**). There is off street parking near the hotel for an additional fee. The Executive Council will meet on Saturday at 9:00 AM in Forbes Hall on campus. Onsite registration check in begins on Saturday at 4 PM followed by a reception with a no-host bar at Gentle Ben's restaurant next door to the Marriott. Registration check in will continue on Sunday morning with the conference beginning around 10 AM. The BBQ will be on Sunday evening (price included in registration) and the Banquet will be on Tuesday evening (additional ticket purchase). Additional schedule information will be posted and disseminated on the Lepidopertists' Society Website, Facebook and Twitter accounts. Please email **meeting@lepsoc.org** with any questions or concerns.

Forbes Hall, which houses the insect collection, on the Univ. of AZ, Tucson campus; the EC meeting will be here on Saturday.

If you would like to become a member of the Lepidopterists' Society please visit **https://www.lepsoc.org/content/new-membership**. Membership has its privileges with a reduced registration fee to the annual meeting, access to current and past issues of our quarterly scientific publication, *the Journal of the Lepidopterists' Society* and of our newsletter, *News of the Lepidopterists' Society*. Members are also eligible for various awards including travel awards to help defray some of the costs to attend the meeting (**https://www.lepsoc.org/content/awards**). Beyond all that, the real strength of this society is its people! We are a supportive group of amateurs and professionals, collectors and photographers, students and retirees, all united around our mutual appreciation of moths and butterflies.

Tucson is a metropolitan city with an airport, university and a wide variety of dining options. The meeting venue has multiple food venues within walking distance in addition to a light rail which can take you downtown for even more dining options. Tucson was recently named a UNESCO World City of Gastronomy, the first in the United States because of its long history of sustainable agriculture and local ingredients. Tucson Botanical Gardens (**https://www.tucsonbotanical.org**), Tohono Chul Gardens (**http://www.tohonochulpark.org**) and the Mission Gardens (**http://www.tucsonsbirthplace.org/tucsons-birthplace/mission-garden-project/**) all interpret this history and provide good opportunities for moth and butterfly photography.

The areas around Tucson offer a wide variety of collecting and photographing opportunities for all insects, especially Lepidoptera. Special attention has been made to account for both the monsoon and the moon phase to schedule this event. A few collecting and photography field trips will be organized and participants are welcome to organize their own informal adventures based on their personal wishes. The Arizona Insect Collection is especially looking forward to hosting moth and butterfly experts in the collection and are including a golf cart service to bring people to and from the collection during the meeting.

Please direct your questions or concerns to **meeting@lepsoc.org**. Hope to see you in Tucson this July!

2017 Organizing Committee (Katy Prudic, Wendy Moore, Gene Hall, Jacqueline Miller, Jennifer Zaspel)

Citheronia splendens, reared from larva, Peña Blanca Lake area, Santa Cruz Co., west of Nogales (photo by James Adams)

PayPal is the easy way to send money to the Society

For those wishing to send/donate money to the Society; purchase Society publications, t-shirts, and back issues; or to pay late fees, PayPal is a convenient way to do so. The process is simple: sign on to www.PayPal.com, and navigate to "Send Money", and use this recipient e-mail address: **kerichers@wuesd.org**; follow the instructions to complete the transaction, and be sure to enter information in the box provided to explain why the money is being sent to the Society. Thanks!

Second Edition of Butterflies of Alaska

Butterflies of Alaska, A Field Guide, Second Edition, Kenelm W. Philip (posthumous) and Clifford D. Ferris. iv + 110 pages, spiral bound with durable covers; 8.5" x 11". The now known 80 resident, 5 casual species, and one currently unconfirmed species are illustrated in full color. Each species entry includes information about geographic distribution, habitat, basic biology, flight period, diagnostic characters, and field behavior. A species index and plant index are included. The second edition includes the recently described (2016) *Oeneis tanana* as well as addressing several taxonomic issues; a flight-period graphic has been added. The book pages have been reformatted such that there is coverage of only one species per page. Maps have been updated to reflect additional records obtained after the first edition was published. $30 plus postage. ISBN 978-1-945170-60-7. Available from BioQuip and various Alaska booksellers.

Kirby Wolfe's new website on the Tiger Moths of Costa Rica

"I want to bring to your attention my new site for the Tiger Moths of Costa Rica, with photographs of 370 species (so far) of identified live Arctiinae. They can be accessed by typing into the web address bar: **https://www.flickr.com/photos/kirbywolfemoths/collections,** or by Googling "Kirby Wolfe Tiger Moths", then in Flickr navigating either to Albums or better yet to More>Collections where you can find the moths divided into their respective tribes and subtribes. This can simplify specific searches significantly. I am frequently adding new material. The site format is very restrictive and I'm hoping to get some help for developing a better site in the future, but for now it kind of works and it's free." Enjoy the photos and the information!

Corrections to items in the Fall 2016 News (Vol. 58:3)

First, the editor would like to apologize to Handly and Dennehy for not matching captions with photos of the Amphion floridensis larva (pg. 122). Clearly, the first photo shows feeding, not the second!

Jim Scott sent some corrections to nomenclature in the table of butteflies encountered at the Lep Soc meeting (pgs. 148-150). *Oeneis chryxus altacordillera* is actually *Oeneis calais altacordillera*. Nick Grishin indicates that it is distinguished from *chryxus* by several nucleotide substitutions, wing pattern, mate-locating behavior and oviposition/hostplant difference. Also *P. cotundra* is considered a subspecies of *P. lupini* by many.

Call for Season Summary Records

It is once again the time of year to prepare your submissions for the annual Season Summary report. The annual report is sent as a hardcopy to members each year, and each year's data is also incorporated into the on-line database. Take the time to access the Season Summary database through The Lepidopterists' Society home page (http://www.flmnh.ufl.edu/lepsoc/) and do a few searches. The value of the on-line database increases as your data gets added each year. Please take the time to consider your field season and report range extensions, seasonal flight shifts, and life history observations to the appropriate Zone Coordinator. Zone Coordinators, their contact information, and the scope of their zone appears on the inside back cover of every issue of the "News".

There are a number of factors that make it necessary for the Zone Coordinators to meet a reporting deadline each year. As a result, you should have your data to the Zone Coordinator(s) no later than **December 15, 2016**. In most of our Nearctic zones, you have long since put away your cameras, nets, bait traps, and/or lighting equipment by that time anyway.

All records are important. Reporting the same species from the same location provides a history for future researchers to use. Report migratory species, especially the direction of flight and an estimated number of individuals. Again, all of these records may be useful in the future.

Season Summary Spread Sheet and Spread Sheet Instructions

The Season Summary Spread Sheet and Spread Sheet Instructions are available on the Lepidopterists Society Web Site at **http://www.lepsoc.org/season_summary.php.** The Zone Coordinators use the Season Summary Spread Sheet to compile their zone reports. Please follow the instructions carefully and provide as much detail as possible. Send your completed Season Summary Spread Sheet to the Zone Coordinator for each state, province or territory where you collected or photographed the species contained in your report.

Important reminder to contributors using MAC computers to submit records

PC operating systems save dates based upon a 1900 format, whereas MAC operating systems save dates based upon a 1904 *default* format. The Lepidopterists' Society master database is maintained in PC format. As a result, if you submit your season summary records on an Excel spreadsheet generated on a MAC to a Zone Coordinator who operates a PC system, without first disabling the default date setting, the dates will be off by 4 years and 1 day. If you submit your season summary records on an EXCEL spreadsheet generated on a MAC to a Zone Coordinator who operates a MAC system, without first disabling the default date setting, the dates will appear proper to the Zone Coordinator but the dates will be off by 4 years and 1 day when they are incorporated into the master data base. In some cases, MAC system dates sent to a Zone Coordinator operating a MAC system are off 8 years and 2 days (we haven't figured that one out). The following are instructions so that this problem will never rear its ugly head again.

Instructions

When a MAC user sits down to enter the very first record of the season, he/she must create a <u>new</u> Excel file. **Before typing in any data,** go to "Tools", then "Options" or "Preferences" depending upon your version of Excel, "Calculations", and ***uncheck*** the 1904 box. Once the data is entered, save this file, and close. If supplemental data is entered directly into this file by keypunching it in, there will not be any problems. However, do NOT paste in MAC data from another file into your file without first ensuring that the 1904 box was *unchecked* in their file PRIOR to entering any of data. Unfortunately, once data has been entered in a file, it does NOT do any good to retroactively *uncheck* the date box!!!

By following these few steps, it is a simple matter to accommodate MAC records. However, you, as the original contributor, must ensure that those steps are taken. Improperly dated records will be rejected and your important records will not get into the database.

Photographs for Front and Back Covers

Please submit photos for the front or back covers of the Season Summary to the editor of the News, James K. Adams (**jadams@daltonstate.edu**). Photos can be of live or spread specimens, but <u>**MUST**</u> be of a species that will actually be reported in the Season Summary for this year.

Leroy C. Koehn, Season Summary Editor, 3000 Fairway Court, Georgetown, KY 40324-9454, **Leptraps@aol.com**

John (Jack) Franclemont, Founding Editorial Board Member of the Moths of North America series, Cornell University professor and curator, with his dog Cho.

Plea for Charitable Gifts and Estate Bequests: Lepidopterology Needs You

Butterflies and moths, and their caterpillars, have found their way into the lives of all members of our Society, whether collector, gardener, photographer, watcher, lister, or scientist. For many of us, Lepidoptera have been a formative and even consuming aspect of our journey on this planet, enriching myriad aspects of our summers, vacations, and daily lives. Any of us that have been watching for more than a couple decades have likely experienced worrisome declines in the faunas of some of our favored haunts. The world's biota is facing many challenges: development, habitat degradation, exotic species invasions, hotter summers, extended droughts, and other unpredictable and unprecedented events. More changes loom on the horizon. Our collections are becoming an increasingly important part of documenting the planet's historical biodiversity. In the future, scientists and conservation biologists will be mining the world's collections for our specimens in the same way paleontologists visit museums today to study fossils. The integrity, longevity, and legacy of our collections can be maintained through charitable gifts and allocations by Society members in our estate plans to support natural history collections, curators and collection managers, and systematic entomology in general. The Wedge Entomological Research Foundation (WERF) is seeking support to endow positions in systematic entomology, initially with enough money to hire a young postdoctoral researcher, but eventually to support fulltime scientists to work on the taxonomy and biology of Lepidoptera.

Douglas Ferguson's passing left an enormous void in our ability to advance our knowledge of Geometridae of North America. Over the past decade, many career Lepidopterists at museums and universities have retired. There is an urgent need to replace some of these positions so that the work of species description, monography, curation, collection-based research, and student training can continue.

Endowments dedicated to the study of Lepidoptera are the only way to ensure that funds and administrative commitments can be maintained in perpetuity. We encourage those who are in a position to make a charitable gift to carefully consider the wording of their gift. If monography and revisionary taxonomy, curation, and collection-based research are important to you, restrict your gift to such. Broadly defined positions, e.g., focused on "Lepidoptera research" at a University, might be filled by a climate-change scientist, an insect-plant ecologist, theorist, or any of a dozen other worthy disciplines—but these would not appreciably bolster the state of lepidopteran systematics and collection-based research at the target organizations and institutions. Also, keep in mind that large charitable gifts and estate bequests can be used to leverage matching dollars from partner institutions, especially from those with a history of supporting lepidopteran taxonomy.In these uncertain economic times, estate gifts are one of the best ways, for those of us that can, to invest in the future of lepidopterology and our collections, which for many of us will be our longest legacy on this planet: well-preserved specimens are expected to last centuries and beyond. The Wedge Entomological Research Foundation (publishers of the Moths of North America series) is seeking charitable gifts to endow postdoctoral and curatorial positions in lepidopteran systematics—to be hosted at the Smithsonian Institution. If you think that you may be in a position to help or may have already named WERF in your estate planning, please contact Eric Metzler (**spruance@beyondbb.com**).

David L. Wagner, **david.wagner@uconn.edu**

Announcements continued on p. 175



Doug Ferguson, Ron Hodges, and Richard Dominick. Three of the six Founding Editorial Board Members of the Moths of North America series, published by the Wedge Entomological Research Foundation. The foundation is named after the Wedge Plantation, home to the Richard and Tatiana Dominick, where the MONA series was conceived.

Conservation Matters: Contributions from the Conservation Committee

Speciation, hybridization, and conservation quanderies: *what are we protecting anyway?*

J. R. Dupuis[1] and Felix A. H. Sperling[2]

[1]Dept. of Plant and Environmental Protection Sciences, Univ. of Hawai'i at Mãnoa, Honolulu, Hawai'i 96822
[2]Dept. of Biological Sciences, Univ. of Alberta, Edmonton, Alberta, Canada T6G 2E9 **felix.sperling@ualberta.ca**

There are few scientific disciplines more prone to social quandaries than conservation biology. Its multidisciplinary and synthetic nature lends itself to conflicts among science, money, laws, and social values, which are encapsulated in questions like *what should you do with limited funding but seemingly endless needs?* In insect conservation, these quandaries often have an added layer of taxonomic uncertainty. When a unique population is discovered in some remnant patch of wildland, the first question is usually *is this a different species/subspecies*? A 'yes' can open the floodgates to discussions of endemism, legal protection, and conservation prioritization. What may have started as a weekend collecting trip, and the excitement of a new discovery, now involves conservation authorities, politicians, expert opinions, and land owners leery about new restrictions on their land.

In recent decades, questions about species identification and ranking have increasingly been answered with DNA-based approaches, which can provide a wealth of information and carry a lot of weight in conservation biology. Yet these genetic tools often raise as many questions as they answer - a frustrating outcome when money is on the line or timelines are urgent. For instance, what should a conservation biologist do when new genetic data fail to support the evolutionary distinctness of an endangered species that has already had millions of dollars spent toward its protection? Here, we consider two butterflies, Lange's metalmark and the Ozark swallowtail, to explore some of these questions.

Lange's metalmark

Lange's metalmark, *Apodemia mormo langei* (Figure 1), was one of the first insects to be considered *federally endangered* under the US Endangered Species Act (ESA) in 1976. It is found only in the Antioch Dunes National Wildlife Refuge (NWR) on the banks of the San Joaquin River downstream of Sacramento, California. Ecologically, Lange's metalmark is restricted to sand dunes, as its larval host plant *Eriogonum nudum psychicola* depends on this dynamic and shifting habitat. However, sand mining beginning in the early-mid 20th century destroyed the once extensive dunes of the area and reduced suitable habitat for this species to ~1.3 hectares in 1979 (Powell and Parker 1993). The Antioch Dunes NWR was established in 1980 to protect Lange's metalmark, as well as two rare plant species, and was the first NWR established with the explicit purpose of protecting rare animals or plants. Since then, extensive conservation efforts have taken place to stabilize populations of Lange's metalmark, including the establishment of a captive breeding program, planting of *E. n. psychicola*, hand-clearing/herbiciding invasive plants, and experimental grazing. Despite these efforts, population numbers are still precariously low, with competition from invasive weeds and wildfires proving to be formidable opponents.

While Lange's metalmark has a wing pattern that is distinct from most of the *A. mormo* species complex, it has little to distinguish it genetically. Using mitochondrial DNA and nuclear microsatellite markers, we found that Lange's metalmark was no more genetically distinct than any other population of the Mormon metalmark (*A. mormo*) complex in California (Proshek *et al.* 2015, *open-access article*). We observed localized patterns of genetic differentiation, as expected given the low vagility and colonial nature of this butterfly, and some populations with relatively higher genetic diversity than the population at Antioch Dunes. We also found some of the morphological characteristics that distinguish Lange's metalmark in individuals from other populations. We are following up with genome-wide single nucleotide polymorphism surveys. These methods still only sample a small fraction of the genome, but preliminary analyses of these data support and expand on the pattern

Figure 1. Adult Lange's metalmark, *Apodemia mormo langei*. Public domain, USFWS, from **https://commons.wikimedia.org/wiki/File:Dorsal_view_of_an_endangered_lange_metalmark_butterfly.jpg**

of local genetic differentiation found in our previous study (Oliver, Dupuis, and Sperling *et al. in preparation*).

Ozark swallowtail

The Ozark swallowtail, *Papilio joanae*, is a relatively unknown creature with a distribution that is localized, as its name suggests, to the Ozark Plateau of Missouri (Figure 2). Its describer, J. Richard Heitzman, spent years amassing rearing records, generating crossing lines, and observing habitat associations to form the basis of our biological knowledge of this species. In appearance, it is almost indistinguishable from *P. polyxenes*, the black swallowtail, and ecologically it uses some of the same hosts, at much the same time of year. However, it flies and selects oviposition sites almost entirely under the forest cover, a unique behavioural trait among its relatives in the Old World swallowtail (*Papilio machaon*) species group.

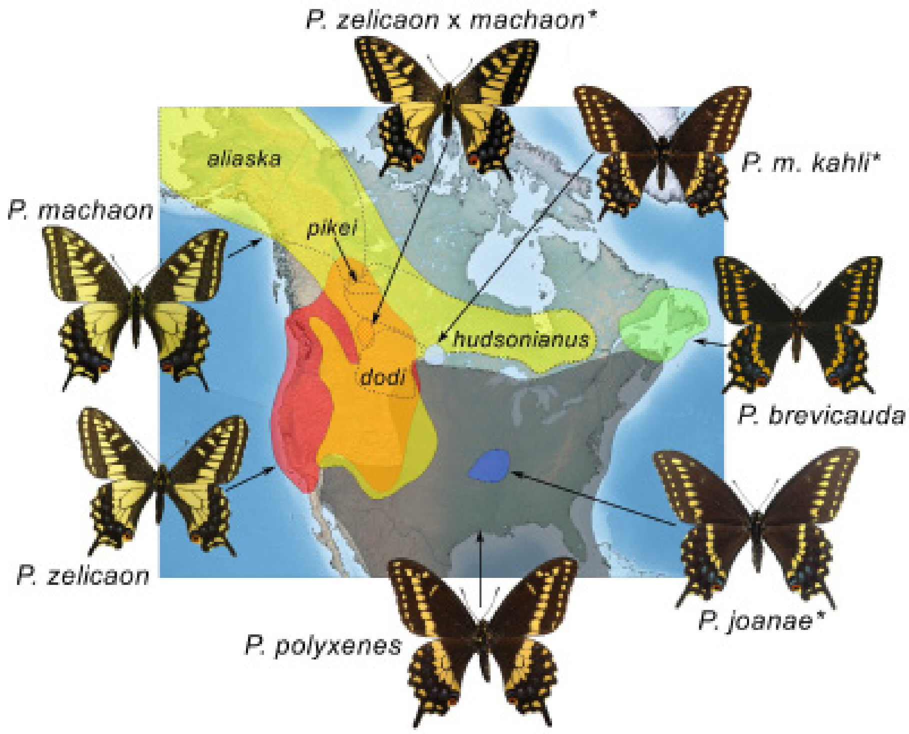


Figure 2. Generalized range map of current distributions of the *Papilio machaon* species complex in North America, from Dupuis and Sperling (2015). Putative hybrid taxa are indicated with an asterisk. Dashed lines indicate approximate ranges of *P. machaon* subspecies pertinent to Dupuis and Sperling (2015). Map image: public domain from www.simplemappr.net, *Papilio joanae* holotype (photograph by John Tewell).

Although the morphology of the Ozark swallowtail is almost identical to that of the black swallowtail, it shares several small characteristics with the Old World swallowtail, most notably a pupil connected to the margin of its hindwing eyespot. This morphological enigma served as the original impetus for investigating the genetic relationships of the group. Sperling and Harrison (1994) discovered that the Ozark swallowtail shared mitochondrial DNA signatures with the Old World swallowtail, suggesting a hybrid origin between the black and the Old World swallowtail. We recently corroborated the initial mitochondrial results, but found that nuclear microsatellite markers showed closer relatedness between the Ozark and the black swallowtail (Dupuis and Sperling 2015, *open-access article*). The mitochondrial DNA lineage of the Ozark swallowtail is shared with only a single subspecies of the Old World swallowtail, *P. m. hudsonianus*, which has a north-eastern distribution in North America (Figure 2). This makes a compelling phylogeographic story of a hybrid origin for the Ozark swallowtail during Pleistocene glaciations (Dupuis and Sperling 2015). Preliminary results using genome-wide markers for the whole species group indicate that, although most similar to the black swallowtail, at fine-scale levels the Ozark swallowtail is genomically distinct from both of its original parental species (Dupuis and Sperling *in preparation*).

Given its localized distribution, the Ozark swallowtail has been listed as "vulnerable" by some conservation organizations (Schweitzer *et al.* 2011), but as "unrankable" in other conservation prioritizations due to lack of information. There are few verified records of the species from recent years. The most recent one that we are aware of is a single individual from 2006, and before that only four individuals were recorded between 1995 and 2006. Many of the regularly-visited collection localities discovered by Heitzman have reportedly been overgrown or replaced with houses.

What to do?

The fact that Lange's metalmark is not more genetically distinctive than many other of the local Mormon metalmark colonies in California raises a fundamental question - *what are we protecting anyway*? If Lange's metalmark passes the bar as a genetically distinct population entity, then perhaps we should also be protecting many other local colonies of Mormon metalmarks and other species that might be just as vulnerable to extinction (some of which are more genetically distinct than Lange's metalmark). Conservation biologists use the term "evolutionarily significant unit" to identify and delimit populations of organisms with particular evolutionary potential (see summary in Proshek *et al.* 2015), even when some of these units are not formally recognized as species or subspecies. However, invertebrates are at a disadvantage when it comes to federal protection of such unique populations, as the ESA only recognizes conservation units below the subspecies level in vertebrates. Lange's metalmark's only saving grace, as far as federal legislation goes, is its formal subspecific status, although the taxonomy of the entire Mormon metalmark complex is tenuous at best (Proshek *et al.* 2015). Because it happened to have a recognizable wing pattern, Lange's metalmark was viewed as distinct, while other populations that have no obvious identifiable traits, but are genetically distinctive, remain invisible to legal protection. In an interesting contrast, the species status of the Ozark swallowtail has done little to help gain its protection.

On the other hand, if it is a particular habitat or location that we are intending to protect with a legislated endangered species as the flagship or umbrella species (see Caro and O'Doherty 1999), then the Antioch Dunes NWR would seem to fit the bill. Lange's Metalmark serves as a culturally recognized flagship of this remnant dune habitat, along with the two endangered plants and a number of other rare species found at the Antioch Dunes (Powell 1978). But it may be instructive to critically evaluate how far conservationists are willing to go to protect such a flagship endangered species. Is the captive breeding program for Lange's metalmark an optimal use of such conservation funds if protecting the NWR habitat is really the main goal? Would conservation funding be better spent in rearing one, or several, of the other rare and unique species found in the Antioch Dunes NWR? If ecosystem preservation or ecological services are the main goals for conservation, perhaps other less "species-centric" conservation approaches would be more appropriate, cost-effective, or ecologically sound.

That leads us to ask whether we are really protecting only those biological phenomena that derive their value from the way they are perceived, rather than having intrinsic objective characteristics like genetic or evolutionary distinctiveness. Perhaps we should be more honest about the cases where we are primarily protecting cultural symbols rather than imperiled habitats. If we take this last line of thought seriously, then we would put greater emphasis on understanding, delimiting, and explaining what kinds of "endangered species" phenomena we are societally prepared to put resources into protecting. This would be a very different process, compared to the work we do to determine evolutionary significance based on, say, genetic data or habitat-based considerations. Being more open about cultural subjectivity in conservation prioritization may provide the flexibility to see conservation quandaries in a new light. There may be cases where genetic data and habitat/ecosystem considerations would only act as supporting factors in a conservation decision, rather than the primary criteria for designating endangered species.

Lange's metalmark is certainly a relic of the days before sprawling development of California's coastline. Should its flagship nature and history of conservation efforts trump the fact that it is no more genetically distinct than other nearby Mormon metalmark colonies? Does a hybrid species like the Ozark swallowtail hold the same conservation value as species that we think originated via phylogenetic divergence? Does the fact that it arose in a seemingly unusual manner (through homoploid hybrid speciation) elevate its status as an interesting biological phenomenon? How should the fact that the Ozark plateau is home to many other endemic species affect the conservation prioritization of those species? Does perceiving the Ozark swallowtail as a beautiful, mysterious entity flitting through the trees count just as much? These questions are obviously subjective ones, and we advocate no hard-and-fast answers here.

But perhaps it is time to more critically re-evaluate the motivations for our conservation efforts. These two butterflies differ considerably in their evolutionary histories, but even more so in their histories of conservation. While one is being brought back from the brink through enormous efforts and costs, we are not certain if the other has been seen in the past ten years. Conservation biology is arguably as much in our heads as it is in nature: humans have significantly altered the planet, and now want to stem losses and make amends with a small number of species. Recognizing the primarily cultural underpinnings of many of our prioritizations could lead us in some interesting directions, such as refreshing public trust and understanding of conservation biology's scientific and societal goals.

ACKNOWLEDGEMENTS

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Moths of the Nature Place, Colorado, Lep Soc 2016

Jim Vargo

P.O. Box 97, Mishawaka, IN 46546 **varlepski@aol.com**

The three columns in the following are the Hodges #, followed by the new Pohl, Patterson and Pelham (2016) checklist #, and then species name. All species were seen July 6-10, 2016.

ADELIDAE

214	210104	Cauchis sedella

DEPRESSARIIDAE

976	420183	Ethmia semilugens

GELECHIIDAE

2090	421009	Chionodes lugubrella

CARPOSINIDAE

2322	480014	Bondia fidelis

SESIIDAE

2581	640122	Synanthedon polygoni

TORTRICIDAE

2847	620612	Olethreutes glaciana
2953	620821	Eucosma rupestrana
3014	621019	Pelochrista ridingsana
3030	621016	Pelochrista ragonoti
3288	621262	Epinotia castaneana
3404	621275	Dichrorampha simulana
3584.1	620235	Anopina internacionana
3712	620408	Sparganothis vocaridorsana
3755	620116	Aethes smeathmanniana
3765	620192	Platphalonia campicolana
3776	620181	Phtheochroa huachucana
3794	620175	Phtheochroa aureoalbida

CRAMBIDAE

4726	800992	Eudonia torniplagalis
4904	801078	Evergestis simulatilis
4912	801086	Evergestis obliqualis
5060	801540	Pyrausta insequalis
5132	801212	Choristostigma elegantalis
5343	800931	Crambus perlella
5388	800967	Crambus dimidiatellus

PYRALIDAE

5514	800077	Aglossa cacamica
5748	800279	Pima fosterella
5764	800298	Catastia actualis
5800	800354	Sciota termitalis
5824	800390	Pyla aequivoca
5841	800409	Dioryctria abietivorella
5850	800417	Dioryctria ponderosae
5861.4	800424	Dioryctria durangoensis
5888.1	800477	Lipographis unicolor
6024	800265	Bandera binotella
6053	800670	Peoria approximella

SATURNIIDAE

7726	890033	Coloradia luski

URANIIDAE

7650	910002	Callizia amorata

GEOMETRIDAE

6289	910711	Macaria coloradensis
6472	910887	Stenoporpia glaucomarginaria
6589	911008	Iridopsis emasculatum
6605	911023	Mericisca gracea
6632	911051	Galenara stenomacra
6760	911189	Pero behrensaria
6875	911310	Snowia montanaria
6879.1	911316	Meris alticola
7018	910599	Nemoria unitaria
7191	910026	Dysstroma formosa
7194	910029	Dysstroma brunneata
7213	910049	Ecliptopera silaceata
7264	910102	Hydriomena morosata
7301	910142	Entephria multivagata
7309	910152	Spargania viridescens
7385	910228	Xanthorhoe alticolata
7449	910296	Eupithecia palpata
7484.1	910387	Eupithecia lafontaineata
7575	910415	Eupithecia mutata
7594	910434	Eupithecia anticaria
7600	910439	Eupithecia graefii
7634	910475	Scelidacantha triseriata
7641	910482	Lobophora montanata

NOTODONTIDAE

7924	930013	Odontosia elegans
7940	930028	Furcula scolopendrina
8014	930107	Oligocentria pallida

EREBIDAE

8121	930299	Virbia aurantiaca
8180	930255	Grammia incorrupta
8207	930366	Lophocampa significans
8261	930434	Ctenucha cressonana

NOLIDAE

8985	931125	Meganola fuscula

NOCTUIDAE

8913	931196	Autographa pseudogamma
8914	931193	Autographa californica
8950	931236	Plusia putnami
9183.3	931397	Panthea greyi
9212	931433	Acronicta grisea
9287	932195	Cryphia olivacea
9339	932326	Apamea auranticolor
9344	932304	Apamea plutonia
9351	932307	Apamea alia
9351.1	932306	Apamea xylodes
9355	932312	Apamea unita
9359	932329	Apamea commoda
9362	932310	Apamea indocilis
9364	932314	Apamea sordens
9374	932355	Apamea niveivenosa
9382	932350	Apamea devastator
9412	932392	Neoligia subjuncta
10190.2	931518	Cucullia dorsalis
10273	932865	Polia discalis
10280	932872	Polia purpurissata
10288	933146	Orthodes detracta
10290	933142	Orthodes obscura
10299	932881	Lacanobia subjuncta
10303	932885	Trichordestra tacoma
10313	932895	Papestra brenda
10317.1	932912	Hadena lafontainei
10322	932916	Hadena circumvadis
10370	933017	Lacinipolia lustralis
10373	933020	Lacinipolia incurva
10379	933027	Lacinipolia umbrosa
10395	933042	Lacinipolia pensilis
10406	933053	Lacinipolia olivacea
10415	933069	Lacinipolia strigicollis
10449	932948	Leucania insueta
10530	933086	Anhimella contrahens
10540	933097	Homorthodes carneola
10541	933098	Homorthodes reliqua
10552	933105	Protorthodes incincta
10584	933134	Pseudorthodes virgula
10617	933183	Anhypotrix tristis
10644	933488	Feltia mollis
10727	933346	Euxoa pleuritica
10764	933391	Euxoa stigmatalis
10794	933375	Euxoa setonia
10795	933372	Euxoa pluralis
10804	933396	Euxoa plagigera
10851	933440	Euxoa redimicula
10864	933483	Euxoa flavicollis
10910.1	933224	Anicla espoetia
10918	933532	Diarsia dislocata
10928	933563	Graphiphora auger
10929	933560	Eurois occulta
10930	933561	Eurois astricta
10931	933562	Eurois nigra
10941	933587	Xestia bolteri
10946	933574	Xestia conchis
11000	933564	Anaplectodes prasina
11001	933565	Anaplectodes pressus
11064	932041	Pyrrhia exprimens

UNIDENTIFIED/UNDESCRIBED

Argyresthia sp., Diploschizia undescribed, Homorthodes undescribed, Hadenine undescribed

Reference

Pohl, G.R., Patterson, B., & Pelham, J.P. 2016. Annotated taxonomic checklist of the Lepidoptera of North America, North of Mexico. Working paper published online by the authors at ResearchGate.net. 766 pp.

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Todd Gilligan, Colorado State University, Bioagricultural Sciences and Pest Management, 1177 Campus Delivery, Fort Collins, CO 80523-1177, (970)490-4478
tgilliga@gmail.com

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Submissions are always welcome! Preference is given to articles written for a non-technical but knowledgable audience, illustrated and succinct (under 1,000 words, but will take larger). Please submit in one of the following formats (in order of preference):

1. Electronically transmitted file and graphics—in some acceptable format —via e-mail.

2. Article (and graphics) on diskette, CD or thumb drive in any of the popular formats/platforms. Indicate what format(s) your disk/article/graphics are in, and call or email if in doubt. The InDesign software can handle most common wordprocessing software and numerous photo/graphics software. Media will be returned on request.

3. Color and B+W graphics should be good quality photos suitable for scanning or, as indicated above, preferably electronic files in TIFF or JPEG format at least 1200 x 1500 pixels for interior use, 1800 x 2100 for covers.

4. Typed copy, double-spaced suitable for scanning and optical character recognition. Original artwork/maps should be line drawings in pen and ink or good, clean photocopies. Color originals are preferred.

Submission Deadlines

Material for Volumes 58 must reach the Editor by the following dates:

	Issue	Date Due
59	1 Spring	Feb. 15, 2017
	2 Summer	May 12, 2017
	3 Fall	August 15, 2017
	4 Winter	Nov. 15, 2017

Be aware that issues may ALREADY BE FULL by the deadlines, and so articles received by a deadline may have to go in a future issue.

Reports for Supplement S1, the Season Summary, must reach the respective Zone Coordinator (see most recent Season Summary for your Zone) by Dec. 15. See inside back cover (facing page) for Zone Coordinator information.

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977 Wicks Drive, Palm Harbor, FL 34684-4656
(727)785-0715
bretcal1@verizon.net

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tgilliga@gmail.com

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micromothman@gmail.com

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The Natural History Museum, Cromwell Road
London SW7 5BD
UNITED KINGDOM
+44 20 7942 5716
g.martin@nhm.ac.uk

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Centre for Biodiversity Genomics, Biodiversity Institute of Ontario, Univ. of Guelph, 50 Stone Road East, Guelph, ON, CANADA N1G 2W1
519-824-4120 ext.55672
kekkonen@uoguelph.ca

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Division of Math and Science, Eureka College
300 E. College Ave.
Eureka, IL 61530-1500
miketol@eureka.edu

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9417 Carvalho Court
Bakersfield, CA 93311
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kerichers@wuesd.org

Assistant Secretary & Assistant Treasurer

Chris Grinter
Illinois Natural History Survey, 1816 S. Oak Street, Champaign, IL 61820-0904;
847-767-9688
cgrinter@gmail.com

Publications Manager

Kenneth R. Bliss
1321 Huntington Trail
Round Rock, TX 78664
(512)850-1700
krbliss@gmail.com

Editor, News of The Lepidopterists' Society

James K. Adams
School of Sciences and Math
Dalton State College
650 College Drive
Dalton, Georgia 30720
(706)272-4427
jadams@daltonstate.edu

Editor, Journal of The Lepidopterists' Society

Keith Summerville
Dept. of Environmental Science and Policy, 131 Olin Hall, Drake University
Des Moines, IA 50311-4505
(515)271-2498
ksummerville@drake.edu

Editor, Memoirs of The Lepidopterists' Society

Kelly Richers
(see Treasurer, above)

WebMaster

Todd Gilligan
(see WebMaster opposite)

Members-At-Large

David Bettman, Shannon Murphy, Brigette Zacharczenko, Christi Jaeger, Caitlin LaBar, Erik Runquist, Megan McCarty, Kathleen Prudic, Mark Walker

Season Summary Zone Coordinators

Refer to Season Summary for Zone coverage details.

Chief Season Summary Coordinator and Editor

Leroy C. Koehn
3000 Fairway Court
Georgetown, KY 40324
(502) 542-7091
leptraps@aol.com

Zone 1, The Far North:

Crispin Guppy
5 Boss Road, Whitehorse, Yukon Y1A 5S9, Canada
(778) 256-1251
csguppy@gmail.com

Zone 2, The Pacific Northwest:

Jon H. Shepard
4925 SW Dakota Ave.
Corvallis, OR 97333
(541) 207-3450
shep.lep@netidea.com

Zone 3, The Southwest:

Ken Davenport
8417 Rosewood Avenue
Bakersfield, CA 93306
(661) 366-3074
kdavenport93306@yahoo.com
with help on moths from Kelly Richers (see Treasurer, this page)

Zone 4, The Rocky Mountains:

Chuck Harp
8834 W. Quarto Ave.
Littleton, CO 80128-4269
(720) 981-5946
cehmoth@aol.com

Zone 5, The Plains:

Michael M. Ellsbury
70855 Highway 8
Fairbury NE 68352-5565
(402) 300-1969
bugsnrails@gmail.com

Zone 6, Texas:

Mike A. Rickard
411 Virgo Street
Mission, TX 78572
(956) 519-0132
Cell: (281) 734-1110
folksinger4@yahoo.com

Zone 7, Ontario and Quebec:

Jessica E. Linton
245 Rodney Street
Waterloo, ON, Canada
N2J 1G7, (519)-489-2568
Cell: (519)-502-3773
jessicalinton86@gmail.com

Zone 8, The Midwest:

Thomas Jantscher
2800 Rustic Pl. Apt. 206
Little Canada, MN 55117-1389, (612) 875-1710
tjantscher@gmail.com

Zone 9, The Southeast:

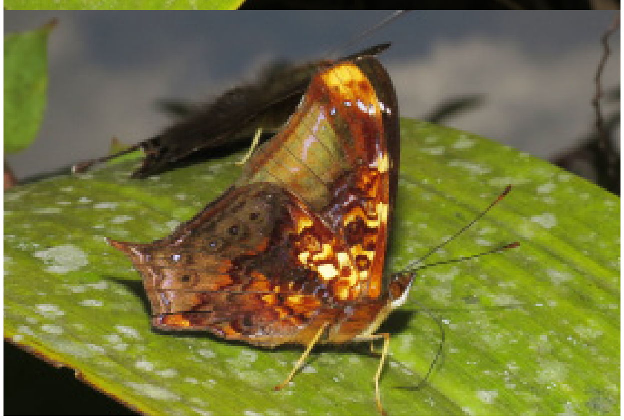
Brian G. Scholtens
Biology Department
College of Charleston
66 College Street
Charleston SC 29424-0001
(843) 637-6224
scholtensb@cofc.edu

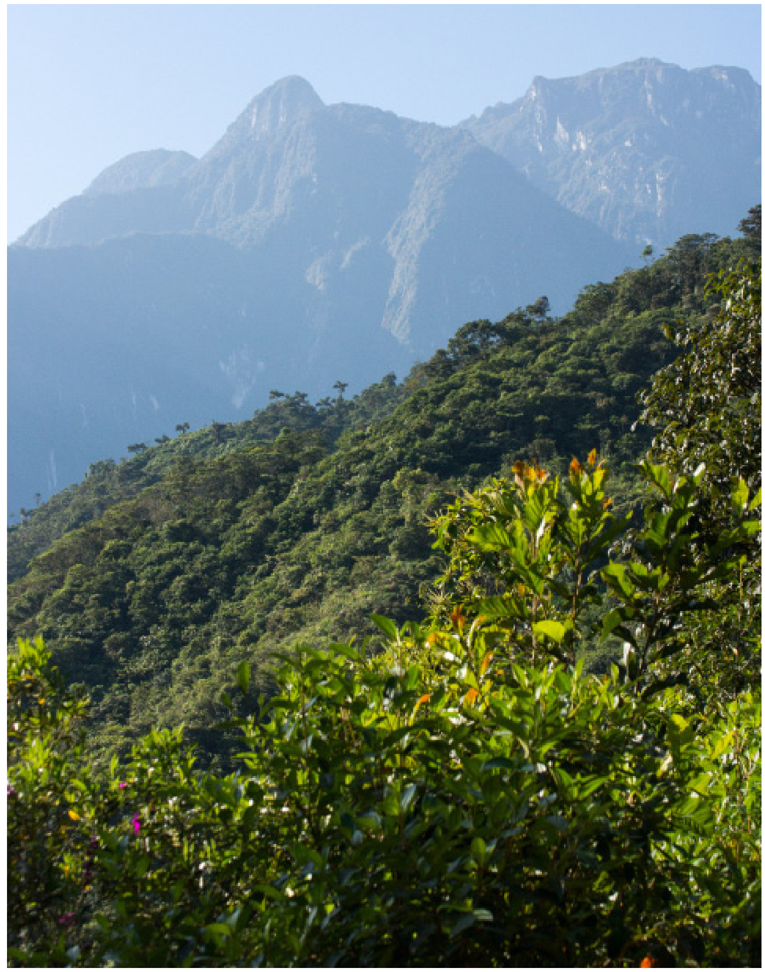
Zone 10, The Northeast:

Mark J. Mello
c/o Lloyd Center,
430 Potomska Rd
Dartsmouth, MA 02748
markmello@lloydcenter.org

Zone 11, Mexico & the Caribbean:

Isabel Vargas Fernandez
Museo de Zoologia, Facultad de Ciencias, Univ. Nacional Autonoma Mexico, Apartado Postal 70-399, D.F., Mexico 04510
ivf@ciencias.unam.mx





Above: *Hypanartia charon*, Montezuma Road, departmento de Risaralda, Colombia, June 14, 2015 (photos by David Geale); Right: scenic mountains at high altitude (2600+ m), above Montezuma, Colombia, January, 7, 2015 (photo by Juan Guillermo Jaramillo) (see article page 159)

Photos from the Lep Soc Meeting, 2016. Above: John Brown, doing something weird with a fork and knife; Above Right: Ian Sagebarth, Peter Houlihan, and Logan Locasio; Right: The Lepidoptera Tattoo contingent (photos by James K. Adams)