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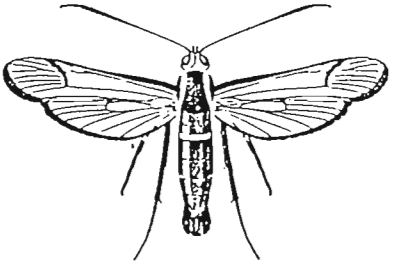
INTERACTIONS BETWEEN SESIIDS AND PHEROMONES

DR. JOHN HOLOYDA

Thanks to recent development of synthetic sex attractants, I have been fortunate enough to spend several hundred hours observing the behavior of males of several commonly occurring sesiid species. These observations seem very consistent, and are different for individual species. Apparently different species react differently with regard to how high a trap is placed, where a trap is placed, and just how persistent individual species are in terms of how long and how cautious the males are as they approach pheromone baits. These and other observations will be discussed below.

First of all, each species seems to prefer a certain height in terms of where the trap is placed. Albuna fraxini (Engelhardt) responds best when the baits are placed very low, between ground level and a foot high. By comparison, males of Synanthedon exitoas (Say) and Podosesia syringae (Harris) prefer that the attractants are between three and six feet above ground level.



Synanthedon pictipes

Certain species can only be trapped effectively if the traps are placed in vegetation, and will not approach pheromone that are placed at the end of a pole or in open environments. A related observation is that identical traps placed in a similar environment about six feet apart often do not produce similar results. For unknown reasons, one trap will often receive almost all of the attention. If the favored trap is placed by yet another trap, the sesiids will still go to the same trap location!

All my observations indicate that males respond to pheromones according to a bell-shaped normal curve; that is, if numbers of specimens of each species are plotted against a 24 hour time interval, each curve begins at zero (no responses), followed by a gradual rise to peak activity response time represented by the top of the curve (maximum numbers arriving at a specific time interval); the numbers then taper off through time until the response is again zero. Individual species which are attracted to the same pheromone component often exhibit different peak activity periods, although the curves may overlap. These respective peak periods, coupled with environmental factors such as height and concealment, may serve as isolating mechanisms preventing the different species from responding to the wrong mate, even when the same pheromone is employed. The peak period for Podsesia syringae, for example, lasts about 0.5 hours, with most males arriving about 9 AM, while Synanthedon exitosa peaks last longer, with peak activity around 11 AM.

There are also differences in how long a sesiid species will stay "mesmerized" at the pheromone. For example, Synanthedon rileyana (Hy. Edwards) males stay for only a few seconds, and are especially wary of anyone approaching. Synanthedon exitosa males, however, are incredibly persistent, often investigating the bait several minutes, and will often follow you around if you are carrying the attractant!

The behavior of individual species near a pheromone source also varies considerably, especially in terms of how easily they can be captured in traps. Podosesia syringae, Synanthedon exitosa, and Synanthedon pictipes (Grote & Robinson) fly directly to the attractant

and are easily captured in numbers. Paranthrene pellucida (Greenfield & Karadinos) approaches very cautiously, taking several seconds to investigate the source from several feet away, and is not easily trapped. Albuna fraxini may only approach as close as three to six feet,often winding its way in and around surrounding vegetation, making capture by an observer difficult. Obviously, the relative numbers taken in traps in a given area do not necessarily accurately represent the relative numbers of all species in that area. Perhaps some species rely on visual cues in addition to the chemical stimulus, especially when the male realizes it is close to the point source.

Those of us interested in the Sesiidae quickly learn how difficult some species are to capture. Without attractants to draw them into traps or slow their flight down enough to capture in a net, we would probably catch few specimens. Oddly, few species are regularly seen and collected using more conventional methods. Although certain species definitely seek nectar or may periodically be collected at rest on host plants or other foliage, collectors do not seem to catch many this way. We can't catch what we can't see!

While the most obvious defense mechanism of these moths is mimicry of wasps, their elusive speed is also a definite advantage. The only time they seem to slow down appreciably is when they begin to inspect a pheromone source. I have spent countless hours trying to observe where they came from and where they depart to, but once a sesiid is about five feet from a pheromone, they seem to literally "disappear" into thin air. Since we can not effectively track them visually, it is likely the other predators also have similar trouble. Their individual mimetic color patterns may be most effective when the moths are feeding, mating, or resting.

Concluding, it appears that the use of synthetic pheromone attractants does not completely control successful capture rates. Different species respond in various ways. Even with pheromones, other factors seem to influence sesiid male responses, making capture of certain species difficult. Close observations are needed to fully begin to understanding the complex behavior of these fascinating animals, and we still have much more to learn.

PROCESSING CLEARWING BORER MOTHS — HERMANN FLASCHKA

Once clearwing moths (Sesiidae) are collected, they must be mounted and prepared for storage in a collection. This article details the processing method that I have developed.

BELLY-UP PINNING (SESIIDS)

One method of pinning sesiids is to treat them like most other lepidoptera. By that I mean push a pin through the middle of the thorax, place the specimen on a spreading board of appropriate size and move the wings, abdomen, and antennae into place. Most lepidopterists are not concerned with the legs and allow them to dangle under the body. More often than not some of the legs will break off, usually when removing the specimen from the spreading board, or when pushing the label up the pin. Aside from esthetic reasons the legs are very important for identification purposes and should be positioned to permit them to be easily viewed.

Mounting sesiids can be extremely frustrating, especially when attempting to position the legs. These are extremely fragile and seem to fall off when just looking at them. For formal mounting, a board is selected that has a groove the width of which is slightly larger than that of the insect's body. This does not work well with Sesiids. When attempting to position a leg an excessive amount of leverage is applied and it will break from the body. Using a board with a much wider groove alleviates the problem when dealing with large species. Small ones still present problems. The thin insect pins (#00 & #000 sizes) which are used to position and hold legs and antennae are extremely difficult to insert into normal spreading boards which are made of basswood. Softer balsa wood boards could prove to be advantageous; however, the vast majority of commercially available boards are of basswood type.

I have developed a method of spreading Sesiids that has been very satisfactory for me and several friends of mine who have ventured to use it. Mounting belly-up! It permits positioning of the legs in full view and without using any excess leverage. Should something go astray and a leg break off, it can be most easily glued in place. The method is described below. The format used is similar to that found in laboratory manuals. I have numbered each step of the procedure along with reference notes. The latter will contain detailed explanations, amplifications, alternatives and general information that would otherwise interrupt the flow of the text.

PROCEDURE

1. Use a block of styrofoam, at least as thick as 1/2 to 3/4 the length of an insect pin and place a piece of adhesive tape in the upper right (or left) hand corner about 2 cm from each edge. The size of the patch will depend on the size of the specimen. It should be large enough so that when the specimen is spread the wings, body, and legs remain within the patch area. For large specimens a patch may not be required as long as the surface of the styrofoam is very smooth. (Note # 1)

2. Make a small grove through the center of the patch using a coin or finger nail. The grove should be of such depth as to allow the wings to be paralliel with the block when the dorsal part of the thorax is inserted. Once the wings are set in position they should be flat against the tape and at 90 degrees to the axis of the body.

3. Place the specimen upside down on the block or place between your fingers or forceps. Insert the appropriate size insect pin through the thorax from the ventral side. (Note # 2)

4. Insert the pin with the moth into the styrofoam block. Do not push the pin deeper than is necessary to hold the specimen firmly in place. (Notes # 3 & 4)

5. Move the antennae into the correct position and hold them down with cross pins placed as close to the base of the antennae as possible. This will permit the covering of the wings with the same paper strips that hold them down.

6. Insert a pin into the styrofoam at each side of the body to keep it from rotating during the positioning of the wings.

7. Using the smallest possible pins (#000), move one pair of wings into the desired position. Repeat on the other pair of wings. (Note # 5)

8. Place a paper strip over one pair of wings and antenna and pin it down. Repeat on the other side. Do not align the paper strips parallel but rather at about a 30 degree angle to the body, covering the outer portions of the wings and the antennae. The strips should meet or overlap in front of the head forming an inverted "V" or arrow pointing forward. (Note # 6)

9 Using two fine pins (#000), move the fore legs into position and pin them in place. Insert the holding pins pointing forwards in order to avoid interference when positioning the other legs.

10. Set the middle and hind legs into position using cross pinning to hold them in place. Do the same with the abdomen.

11. Allow the specimen to dry long enough for the legs to stay in position. Once this point is reached, remove all the holding pins except the ones holding the paper strips. (Note # 7)

12. Allow the specimen to completely dry. During the drying period make several freeze preventing "pushes" to prevent the specimen from becoming "stuck" on its mounting pin. When the specimen is completely dried remove the paper strips. Carefully lift the pin with the moth from the styrofoam block. Working close to the blocks surface push the specimen down the pin with a small pair of forceps and allow the moth to fall gently on the block. Use extreme care to prevent legs and antennae from being broken off.

13. Turn the specimen over and gently push the pin from the dorsal side through the thorax using the existing hole. A magnifying glass or low power stereo microscope will facilitate this operation. Position the specimen at the desired height on the pin and secure it with a small drop of glue. (Note # 8)

NOTES

Note # 1. Styrofoam offers several advantages. It is widely available, (packing strips for appliances, etc.) and can easily be cut into any size. It is extremely soft; even a #000 pin can be easily inserted into it. The softness is very advantageous; once a pin has been inserted to hold down a leg, wing, etc. it can be pushed deeper into the styrofoam to "get it out of the way" and eliminate interference during subsequent steps in the pinning process. When using styrofoam it is important to remember that some chemicals employed by lepidopterists (e.g. ethyl acetate) will dissolve styrofoam. The tiniest drop causes the collapse of the walls of the styrofoam bubbles and a large hole results. When after some use the tape becomes rough with pin holes, simply replace it.

Note # 2. Until you become familiar with inserting the pin from the ventral side, the following guide line should help. In fresh specimens the femora of the forelegs are nicely aligned in the middle of the thorax extended towards the abdomen. If you insert the pin between the femora close to their rear end and push the pin through at the correct 90 degree angles, the pin will emerge in the middle of the dorsal portion of the thorax. Experiment with large "junk" species before attempting small or choice species. Specimens that are distorted from being papered or improperly packaged require special attention. Be sure the forelegs are in the correct positions, one to the left of the pin, the other to the right.

Note # 3. Sesiids do not fold their wings over their bodies like butterflies. Therefore placing the specimen upside down on the block causes no problems. Be sure that the groove has the correct depth. Place the specimen into the groove, push one wing pair down

and check whether the wings are at 90 degrees to the body and lay flat on the tape patch. If the groove is too shallow, simply deepen it. If it is too deep, then prepare a new tape patch and leave the deeper groove for another specimen. Make sure there is sufficient working space between the two patches.

Note # 4. When a specimen is mounted and dried it must be removed from the pin, turned over and re-pinned through the existing hole from the dorsal side. To accomplish this, one must not allow the specimen to get stuck to the mounting pin (the body contents dry and act like glue, freezing the specimen to the pin). It is therefore extremely important not to insert the mounting pin too deep into the styrofoam block. To prevent freezing, simply push the pin several times a little bit deeper into the styrofoam block. For small specimens, the first push should be done a few hours after mounting, followed by one push the next day. No additional pushes should be required. For large specimens a push every 3 or 4 days should be adequate.

Note # 5. The hindwings of Sesiids are double folded and rest -costa-to-costa- directly under the forewings. When it comes to separating and unfolding the wings there is rarely a problem when dealing with large species. Placing the tip of a forceps behind a wing pair and pushing forwards will give the desired result. It is then a simple matter to proceed. Insert a small pin (#0 or less) behind the forewing costa at a quarter of the wings length, move forward and pin into position. In most cases the hindwing will remain linked and follow. If not pin it down separately.

Note # 6. The inverted "V" arrangement of the paper strips allows room for the movement and positioning of the middle and hindlegs without interference from the strips. Be sure to securely pin down the paper strips; otherwise, because the styrofoam is somewhat spongy, the wings may move. The use of thick glass headed pins and heavy paper, preferably folded to achieve double thickness is recommended. Push the glass headed pins holding the paper strips deep into the styrofoam block to provide an unobstructed work area.

Note # 7. It is important to remove these pins early, especially when working with small specimens where very thin ones are used. When removing a pin be sure to pull it in the exact direction of its axis. If this is not done, the pin will bend and when its tip comes out of the styrofoam, it will spring back into the straight position.Thereby a leg or antenna can be catapulted out of sight.

Note # 8. It is a rather common occurrence for Sesiids to grease. De-greasing is accomplished in the usual way by placing them in a solvent. It is important to remember that the glue used to secure the specimen to the pin or to repair damages may dissolve in the liquid. Use glues that are not affected by the solvent. I use finger nail polish to secure the specimen to the pin. Should the need arise to remove the moth from the pin, a drop of finger nail polish remover will soften the glue. For repairing broken appendages I use "Wing Paste" which is available from AMBI Products (11330 Dillon Heights Ave., Baltimore, MD 21228. I am not rewarded for mentioning this or any other product. I simply report my experience; other products may work equally well or better). The glue contracts upon drying and sets rapidly, but contrary to "instant or super glues" not so fast that correcting positions of appendages becomes impossible. The glue as well as nail polish will not be softened by any of the common degreasers that I have used. These include toluene, benzene, benzin, naphtha, 1,1,1, trichloroethane (the latter two are available in hardware stores under the trade name ENERGINE) and trichloroethylene. I do not recommend the last named because in some instances I have noticed signs of corrosion on pins.

GREASING, WING-FOLDS AND OTHER THINGS

DE-GREASING

As mentioned above, Sesiids are notorious greasers. De-greasing is not new to lepidopterists and most of them have developed their own personal method. However, I will elaborate on this subject in regards to Sesiids.

Saturnids and other large moths usually do not grease until well after they are set and determined. This is not the case with Sesiids. I have encountered specimens that have been in a trap for only a very few days and already showed signs of greasing. In many cases determination of greasy Sesiids is very difficult, and thus, aside from esthetic reasons, degreasing becomes almost a necessity. When performing the operation it is important that the specimens remain in the degreasing fluid long enough in order to remove all the fat. If this is not done, greasing will eventually re-occur. When to degrease is a critical question. I recommend de-greasing after the specimen is mounted, set and dried. De-greasing before mounting removes all the body fat and fluids and the specimen is no longer pliable. Mounting a stiff specimen is extremely difficult. It is, however, of the utmost importance that the specimen be bone dry before being placed into the de-greasing solvent. Any moisture remaining in the specimen will cause the appendages to shift and loose the position achieved when originally mounted. It is very difficult to re-mount a fully degreased specimen even after a prolonged stay in the relaxing jar.

Minor resetting is facilitated by using the relaxing fluid sold by AMBI (see above). I recommend making a small board with a wide groove from balsa wood. A drop of relaxing fluid is applied to the wing base; this allows the wing to be moved after a minute or so.

The legs and antennae are treated in the same manner. Once the relaxing fluid has been applied quick action is necessary. The liquid contains organic components which evaporate rapidly and once they are gone the brittleness returns. Wait about 10 minutes before removing pins and paper strips. There is another advantage to this relaxing fluid in that it does not leave spots on scaled portions of the wings, a most welcome feature when used on other lepidoptera, especially those that are white or yellow.

The relaxing fluid can also be quite handy when a leg or antenna is broken off any specimen in the collection. Once the legs and antennae are brittle they will crumble when grabbed by forceps, even with the utmost tenderness being employed. I transfer the loose appendage to a piece of paper by using a slightly moistened pin. I then apply a drop of relaxing fluid and after a minute the appendage is pliable and will stand the pressure of the forceps and can be glued back in place.

WING FOLDS

The hindwings of Sesiids have a peculiar double fold. This is evident with specimens collected in dry traps. The hindwings seem to be absent. However, they are only folded and hidden from view under the forewings. To separate the wings and unfold the hindwing is not difficult when working with large specimens. The situation is different for small specimens and can be aggravating. The wings stick together. The early stage of greasing is the culprit. Separating the wings may be accomplished by moving the wings with a pin placed behind the wing bases near the thorax and gently pushing forward. With a specimen that is already pinned belly-up on the block, another possibility exists. Place a pin on the outer portion of the underside (now facing up) of the forewing, parallel to its surface. Pressing down slightly, carefully move the pin towards the body. Because the hindwing is shorter than the forewing the pin can slide underneath the hindwing, which then can be lifted up and separation is achieved.

Unfolding the hindwings is a problem that I have devoted a considerable amount of time, experimentation and the sacrifice of many specimens, but admittedly without having found a solution satisfactory to all circumstances. Sometimes a short stay in the de-greaser will remove all or just enough grease from the wings to permit the separation and unfolding. The duration of the stay in the de-greasing fluid is critical. A specimen that is left for a long period will become stiff or brittle and their mounting is difficult. Too short a stay leaves some grease and not much of an advantage is gained.

I have noticed that nearly all specimens collected in a soap trap have their wings separated and unfolded. I have therefore attempted to solve the wing problems of small moths captured in "dry traps" by immersing them in a detergent solution. I have in some cases succeeded, however, not every time. There could be a certain type of detergent and a specific concentrations that would prove to be universally applicable. I did not have enough small specimens to sacrifice for extended experimentation.

TRAPS

The physical condition of a specimen is determined by its method of capture, whether by trap or net. I use traps almost exclusively and will limit my comments to trapped moths.

Sticky traps may be of great value to survey work because they are inexpensive and space effective during transport. According to some experts, they catch Sesiids that are "too clever" to enter other traps. I have found sticky traps extremely frustrating and, as hard as I have tried, I have never taken a decent specimen from one. I gave up on them early on.

I now use the Universal Traps which can be obtained from Great Lakes IPM, Inc. of Vestaburg, Mi. (Southern Lep. News. Vol. 11, No 2). There are three parts, the top hat, funnel section, and the bottom. Because the parts, regardless of color, are interchangeable it would be interesting to test various color combinations as to their effectiveness towards trapping different species. Supposedly the yellow-white combination will not attract as many bees and wasps. I use the yellow-white combination to help me find the traps once I have placed them. Unfortunately this color combination may also interest curious humans resulting in theft, damage, or total destruction of the trap.

When emptying traps I remove all the specimens with forceps and transfer them to a film canister (for 35 mm film). At the bottom of the canister I place a few crystals of chlorocresol and I cover them with a disk cut from a paper towel. The disk should have the same diameter as the canister. It is possible to then layer the specimens with additional disks separating them by date and locality. I wet each disk with one drop of relaxing fluid; water can be substituted. I store the canister in a refrigerator or even better a freezer. This retards or prevents greasing. Unfortunately, the canister (at least the clear type that I prefer) do not always seal tightly and as a result evaporation may occur. It is then necessary to add water or relaxing fluid to make up the loss.

I also use the canister to ship material. Before mailing the canister I fill it to the top with crumbled toilet paper in order to prevent the specimens from moving and being damaged in transit. It is of the utmost importance that the packing is very loose; just enough to do its job. Otherwise specimens will be under pressure and become distorted, making subsequent setting quite difficult.

The more frequently traps are checked the better the condition of the specimens and mounting is easy. When traps are left out for a lengthy period, those individuals that become entrapped early will be battered and damaged by those arriving later. This problem becomes more serious when there are great numbers of individuals during peak flight periods. When many moths of the same species are collected there are always several individuals in good condition. However, when more than one species is attracted to a pheromone trap, Murphy's Law always applies! The species that you most desire will be the one that gets the worst.

To prevent problems I have used a "Soap Trap". I fill a 9 inch pie pan (the size can vary) with water containing approximately five drops of liquid detergent. In the center of the pan I place the pheromone on a pedestal less than a 1/2 inch above the level of the solution. I add .05% sodium azide to the soap solution to prevent the formation of mold and algae and to reduce the action of bacteria. I then place a plywood cover over the top, supported and secured with bricks. The cover keeps rain from washing out the trap and the heat of the sun from causing rapid evaporation of the water and pheromone. It also prevents leaves and other debris from entering the trap.

Soap traps must be checked frequently and are preferably used in your yard or nearby locations. I do not recommend leaving specimens in the solution too long. They become soggy and fall apart easily. I suspect that this is caused by enzymes which are not inhibited by the preservative. This condition makes soap traps impractical for use far away in the field. The evaporation problem can be solved by applying the old "chicken watering" method. Fill a small narrow necked bottle completely with water. Using the tip of your finger cover the opening and place the bottle in the solution with the neck down and below the level of the solution. Then remove your finger. Once in the solution the water cannot escape. Position the bottle with the use of coat hangers so that the mouth of the bottle is slightly below the desired water level. As the water in the dish evaporates the level of the soap solution falls below the mouth of the bottle. A bubble of air will enter the bottle and allow enough water to flow out to restore the soap solution level. Thus maintaining the soap solution level is no longer a problem, but it is still necessary to remove moths from traps frequently to prevent the decomposition of specimens.

Once I take a specimen from the soap trap I place it in clean water. The little cups that hospitals use for dispensing pills are ideal for this purpose. I swivel the cup gently and then dip the specimen in another cup containing some alcohol. I allow to dry for a short period of time on a paper towel. The specimen is then ready to be mounted or placed into storage.

Many years ago I was interested in Coleoptera and collected among others ground beetles. One way to collect some species is to put a plastic cup into the soil (the rim even with the ground). The cup contains some molasses that is an attractant for the beetles. The trap should be protected by flat stone plates or a pyramid of heavy stones with some holes left to allow the scent to escape and the beetles to enter. Otherwise the trap will be licked quantitatively clean by raccoons!

I recently learned from Dr. H. Romack Jr. (Cambridge, NY) of traps that contain some vinegar in the water with table salt at high concentration. Vinegar is the attractant and salt the preservative. Neither compound is considered a delicacy by raccoons!

I plan to try out traps containing soap with salt as the preservative and encourage others also to test the possibilities. Admittedly beetles are more robust than Sesiids, but it may work. Salt is a preservative that has been used for quite some time (pickling in brine, rubbing salt into meat, etc.). From a physico-chemical point the following should be considered. When a butterfly or moth is exposed to water dripping from a faucet, no wetting will occur. After all how would the creature survive in a heavy rain storm? However, when soap is added to the water its surface tension is lowered and "wetting" takes place (the principle underlying washing). The Sesiid wing touching the surface of the liquid in a soap trap gets wet and the moth is "held". The question is how will a high salt concentration affect this mechanism? The salt prevents the growth of algae, fungi, and probably bacteria as well. The salt should alleviate the problem with enzymatic decomposition mentioned above. Enzymes act only under rather narrowly limited conditions concerning temperature and chemical environment. With a solution high in salt outside the body osmosis through semipermeable membranes at the abdomen should push out of the specimen and change the composition of the liquid inside to an extent of stopping the action of the enzymes.

If the hypothesis holds soap-salt-traps with the implementation of the "chicken watering" system would allow the placing of those traps far away, checked at lengthy intervals and still obtain good specimens.

I will mention another type of trap that was brought to my attention by Dave Baggett and that I had noticed the advertisements of in some journals: the "Wasp Trap". It consists of a plastic dome to which a reservoir in the shape of a doughnut, cut in half, is attached. Through the hole in the reservoir passes a tight fitting funnel with the narrow end pointing up into the dome. The ring portion of the reservoir is filled with a sugar solution containing some liquid detergent. The contraption is then hung in an area that is desired to be "free of wasps". I replaced the sugar/soap solution with a soap solution and placed an L-103 lure into the upper portion of the funnel. Over a three week period it collected only one (!) P.syringae, while a universal trap nearby was virtually full of them. The best explanation that I can provide for the failure of the wasp trap is as follows: the sugar in the trap solution ferments. Thereby, substances are formed that have a scent that attracts wasps. At the same time carbon dioxide forms, which is denser

than air, and "flows" out of the trap carrying with it the attractants. This sets a trail for the wasps to follow. When used as a Sesiid trap no carbon dioxide forms and no gas motion exists to carry the scent from the lure located high up in the funnel. Would the lure have been placed in the lower end of the funnel it could attract Sesiids. However, they would have no motivation, unless suicidal, to fly up into the dome and drown in the soap solution.

There are many more interesting points related to collecting, trapping, mounting, rearing and studying Sesiids. Other lepidopterists may have found different techniques and solutions to some of the problems mentioned above and I encourage them to send a note or article to the newsletter.

COLLECTION OF CLEARWING MOTHS (SESIIDAE) BY MEANS OTHER THAN PHEROMONES — DAVE HAGGETT

Most of the recent literature discussing Sesiid moths has been dominated by the extremely effective attraction of males to synthetic sex pheromones, especially isomers of 3, 13-octadecadien-1-01 acetate, the corresponding alcohols, or mixtures of these components. Without the biotechnological advances allowing us to employ pheromones, we probably would still know very little about this extremely interesting family of moths. Many collectors now regularly use pheromones as part of their field equipment, and interest in these animals is reaching a peak.

We have learned that many species are much more common than previously thought. Although these moths are day-fliers, they are rarely seen, even by knowledgeable field workers, unless pheromones are used. This article will document capture of several species via methods other than sex attractants. Aside from certain species with economic importance as pests of cultivated plants, the general ecology for many species remains poorly known.

LIST OF SPECIES

Paranthrene asilipennis (Bdv.) : a single female was collected while ovipositing on a young oak sapling stump in a power line clearing near Middleburg, Clay County, Florida, on March 10, 1990. Several others were seen in similar behavior between 1200-1400 hours but were extremely wary and difficult to approach (HDB).

Paranthrene simulans (Grt.) : a female of the form palmii (Hy. Edw.) was collected over flowers of Vaccinium arboreum on May 4, 1988 near Stage Pond in the Withlacoochee State Forest, Citrus County, Florida. While the presence of many males in the area was easily ascertained with pheromones, no others were seen at flowers in spite of diligent searching (HDB).

Vitacea polistiformis (Harr.) : a female was collected using MV light on August 19, 1982 at Torreya State Park, Liberty County, Florida (HDB).

Melittia cucurbitae (Harr.) : a single male was collected while nectaring at the flowers of Phyla nodiflora at Black Creek and U.S. 17 north of Green Cove Springs, Clay County, Florida (HDB). Specimens of both sexes can often be taken at rest on leaves of cultivated squash.

Osminia ruficornis (Hy. Edw.) : several fine series of this tiny species were collected in Gainesville, Alachua County, Florida at rest on the foliage of several plants in a vacant lot during September and early October in 1987. Both sexes, including mating pairs, were found commonly between 1400-1700 hours, and there seemed to be possible preference for a species of Galactia vine present at this locality (CMS, HDB, RMG).

Synanthedon geliformis (Wlk.) : a single specimen was collected at MV light on May 22, 1990 near Marion Oaks, Marion County, Florida by JSK. Several specimens were collected in a fermented bait trap in Jacksonville, Duval County, Florida using a mixture of beer, sugar, crushed apples, and molasses. These were taken in the late 1970's and forwarded to the late Charles Kimball; precise data is not available, but the specimens should be in the Harvard MCZ. I also found males attracted to the base of an old Pecan tree at the same site between 1600-1700 hours, probably seeking freshly emerged females, something I did not realize back then (HDB).

Synanthedon scitula (Harr.) : a single specimen was collected at an incandescent light after dark at the Fontana Dam, Graham County, North Carolina on August 15, 1982 (HDB).

Synanthedon acerni (Clem.) : this species is frequently collected at night using UV/MV lights for collecting other moths. Here is a summary of Florida light captures is provided by county: common, Feb.-Mar. 1987, Hammock Park, Dunedin, Pinellas County (LCU, HDB); Feb.-Mar., and June 1989, Belleview, Marion County (JSK); Mormon Branch, Ocala National Forest, Marion County, Feb. 17, 1990 (JSK); Otter Creek, Levy County, Mar. 30, 1990 (JSK) Munson, Santa Rosa County, May 19, 1989 (JSK); Blackwater River State park, Santa Rosa County, July 3, 1982 (HDB); Holt, Santa Rosa County, August 27, 1990 (JSK); Torreya State Park, Liberty County, Mar. 31, 1979, April

4, 1981, August 5, 1980, and Sept. 4, 1983 (HDB, RMG); Goose Pasture, Jefferson County, April 19, 1990 (JSK); Aspalaga Landing, Gadsden County, Sept. 26, 1986 (HDB); Highlands Hammock State Park, Highlands County, Oct. 11, 1986 (HDB).

Synanthedon alleri Engelhardt : a male was taken at building lights at the Jacksonville Police Academy, Duval County, Florida on June 15, 1977; another specimen was taken at building lights at Blue Springs State Park, Volusia County in October of 1983 (HDB, DP).

Synanthedon refulgens (Hy. Edw.) : this species also seems to come to light at night with some frequency. Torreya State park, Liberty County, Florida, July 5, 1984 (HDB, RMG); Goose Pasture, Jefferson County, Florida, May 27, 1989 (HDB, JSK); McKethan Lake State Recreation Area, Hernando County, Florida, Sept. 19, 1982 (WLA).

Synanthedon sapygaeformis (Wlk.) : several specimens were collected over the flowers of Bidens pilosa, Gainesville, Alachua County, Florida during September and October of 1987 (HDB, CMS).

Synanthedon arkansasensis Duckworth & Eichlin : a single female was collected at MV light at Shell Bluff Landing, Crescent Lake, Flagler County, Florida on May 20, 1990 (JSK).

Carmenta bassiformis (Wlk.) : a single specimen taken at MV light, Benton, Columbia County, Florida on June 30, 1990 (JSK).

Carmenta texana (Hy. Edw.) : a male was collected over flowers of Eupatorium odoratum on January 26, 1980, Key Biscayne, Dade County, Florida (HDB).

Alcathoe autumnalis Engelhardt : series of adults were collected by several members of the Southern Lepidopterists' Society during our south Texas meeting. Adults of both sexes were abundant on or near Clematis vines along the railroad trestle near Madera, Hildago County, Texas on Oct. 21, 1984 (CMS, HDB). Several mating pairs were collected as well.

While pheromones obviously are the method of choice for obtaining series of males, we still need to learn a great deal more about the general ecology and host plant affinities for many of the species. We need more observation regarding nectar sources and behavior for these fascinating moths. It is also worth a note when species other than S.acerni are captured at light. I suspect that malaise traps may be very effective when used in conjunction with pheromones for certain species, since some species are extraordinarily wary around pheromone baited traps and do not readily enter them, even when attracted. They are probably looking for some visual cue when they approach a point source.

Persons whose records are noted above with the initials as follows: HDB = H.D. Baggett; JSK = J.S. Kutis; RMG = R.M. Gillmore; CMS = C.S. Stevens; WLA = W.L. Adair; LCD = L.C. Dow; and DP = D. Profant.

WHERE TO GET WHAT FOR SESIID MOTHS — LEROY C. KOEHN

I currently know of only one supplier of traps and pheromone lures. However, I have listed several other suppliers of entomological equipment who offer items other than traps and pheromone lures.

Great Lakes IPM, 10220 Church Road NE, Vestaburg, MI 48891 Telephone 517-268-5693. Traps, lures, and killing agents. Write or call for a free catalog.

BioQuip Products, P.O. Box 61, Santa Monica, CA 90406, Telephone 213-324-0620 Large selection of supplies for entomology, pins, books, spreading boards, killing agents, etc. Write or call for free catalog.

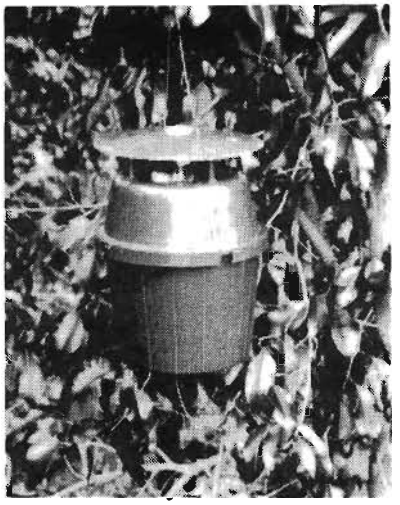
American Biological Supply, Inc.(AMBI), 11330 Dillon Heights Ave., Baltimore, MD 21228 Telephone 301-747-1797 General supplier of biological and entomological equipment. Pins, spreading boards, wing paste, relaxing fluids, etc. Send $1.00 for complete catalog. Cost of catalog refunded with order.

There are numerous publications and books dealing exclusively with sesiid moths. I have listed those most widely used.

Monograph of the Sesiidae of America, North of Mexico. Beutenmuller, W., 1901, Memoir Amer. Mus. Nat. Hist. 1: 217-315. This book is out-of-print. It is well illustrated, although the nomenclature is out of date. It is difficult to locate. However, used book dealers will occasionally offer it. A must book for the serious collector.

The North American Clearwing Moths of the Family Aegeriidae. Englehardt, G.P., 1946, U.S. Natl. Mus. Bull., 190: 1-222. This publication is out-of-print and difficult to obtain. The color illustrations are excellent. (Cont. on Pg.# 46)

(**Editors note** This article has been re-written. The original article was published in Vol 11, No.2 of this newsletter. The purpose is to help others who are interested in collecting and studying these fascinating insects.)



UNITRAP

Collecting sesiid moths has never been an easy task, even after the introduction of pheromones. I tried several times to collect them with rather poor results. I faithfully pinned a pheromone strip to my net and hat before venturing out to collect. This did help me see a few bugs, however, by the time I recognized one as a sesiid, it would vanish instantly. Dave Baggett introduced me to a sticky trap for sesiids. Its principles were similar to a sticky trap for flies and just as effective. (See Southern Lep. News Vol. 8 # 2) I really got stuck on and with sesiids. I became rather frustrated in my attempts to remove delicate sesiid moths intact from the stick-um of the trap. Even once removed the moths had to be cleaned in a solvent. Some how after all that, they lost some of their looks and in the end my patience.

While attending the 1989 spring field meeting at Welaka, Hermann Flaschka introduced me to a new and easy to use sesiid trap that is inexpensive and readily available from a supplier. This is a kill type trap that leaves the specimens in excellent condition. The traps are made of durable plastic that will last for many years (See Fig. #1). I started using these traps in my yard and to date have collected 11 species, and I live in an asphalt and concrete city. During visits to my in-laws in Kentucky I had the opportunity to use them on several occasions and experienced some excellent collecting. The killing agent is a piece of Vapona, cut 2 inches square and wrapped in aluminum foil. Punch 25 or more holes in the foil with a pin to allow the Vapona to escape. When not in use, simply place the aluminum foil blocks in a pint size zip-lock (sandwich bags). This will extend the life of the Vapona as well as trap the poisonous fumes during transit or storage.

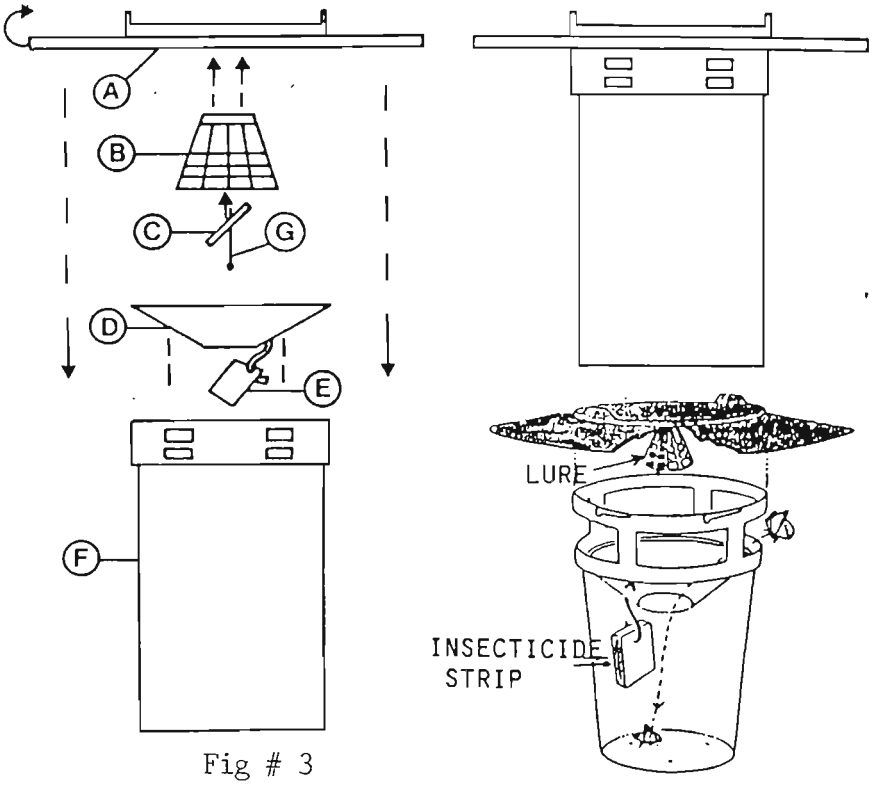


Fig # 3

1) Insert pheromone cage (B) into designed space under lid (A) and push lightly until interlocking.

2) The pheromone lure (C) is suspended with a needle (G) prick to the bottom of the pheromone cage (B).

3) Hang the insecticide strip (E) (Ex:Vapona Type) under funnel (D) with a hook and insert funnel (D) into container (F) up to its place. Make sure funnel is fixed inside.

4) Join container (F) to lid (A) by screwing both pieces together.

5) Attach wire (H) to lid (A) and hang trap to a tree.

UNITRAP

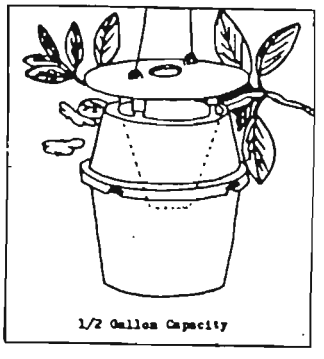


Fig # 2

I take my traps with me whenever I travel, even on business! I pack several in my suitecase. However, each pheromone lure should be placed in an air-tight container to prevent the different pheromones from contaminating each other. I use pint size zip-lock bags for this purpose also. When I get where I am going I find several convenient locations and hang my traps.

There are two styles of traps available, Multi-Pher (Fig # 3) and Unitrap (Universal trap, Fig # 2). Both are funnel type. The traps and pheromones are available from Great Lakes IPM, 10220 Church Road NE, Vestaburg, MI 48891. Write for a free catalogue.

(Cont. from Pg.# 44) The Moths of America North of Mexico: Fascicle 5.1 Sesioidae. Eichlin, T.D. and Duckworth, W.D., The Wedge Entomological Research Foundation. This is the most current and up-to-date publication. It is well illustrated and contains considerable information for each species.

If you know of other books, dealers/suppliers of equipment, and information about sesiid moths which would be of importance to the membership, please let us hear from you.

1990 ANNUAL MEETING REPORT — JEFFREY SLOTTEN

The 12th Annual Meeting of the Southern Lepidopterists' Society was held the weekend of 12-14 October at the Archbold Biological Station near Lake Placid, Florida. The meeting was well attended, and included special guests Dale Schweitzer (Invertebrate Zoologist, The Nature Conservancy), Mark Bailey (Alabama Natural Heritage Program), and Mark Deyrup (Biologist, Archbold Biological Station). Members in attendance included Dave Baggett, John Calhoun, Leroy Simons, Hermann Flaschka and wife Hanna, Marc Minno and wife Maria(plus Angie and Ivan), Tom Neal, Jack Heinrich, John Heppner, Paul Pfenninger, Leroy Koehn, Jeff Slotten, Charles Stevens, Howard Weems, Deborah Lott and husband Terry, Greg Myers, Wayne Miller, William Nix and his parents, and Vincent Golia.

Several members arrived early Friday afternoon and collected around the station. Sesiid traps were set out around the premises and at the nearby McArthur Ranch. Most of the members stayed in the bunk houses at the ranch, which were air-conditioned, equipped with beds, bedding, showers, and full cooking facilities. Meals were provided at the station for those attending. Moth collecting was available Friday evening.

Saturday morning found most of us in the field, where a fair number of butterflies and schinia moths were collected. The larvae and ova of Battus polydamas were found on Aristolochia maxima on the station grounds, and the larvae of all stages and color phases of the sphingid Eumorpha fasciata were absolutely abundant on Ludwigia peruviana growing prolifically around the lake edges near the ranch. The endemic pyralid, Petrophila drumalis, was extremely abundant at the ranch. A fair number of new station records were obtained, in spite of previous efforts of numerous lepidopterists who have visited the station through time.

Saturday evening commenced with a chicken dinner, followed by the business meeting. Jeffrey Slotten and Leroy C. Koehn chaired the session, which focused on the directional goals of the group, exchanged ideas on collecting techniques, photography, and locality information.

Two major issues were addressed, the first and most crucial regarding policy changes of the Florida DNR and collecting in Florida State Parks and Preserves. It seems that a major restructuring has taken place and there was little information given to RA pass holders about the changes, which apparently created serious problems primarily through the basic lack of communication. We had hoped to have several of the DNR Biologist in attendance to convey their thoughts and concerns to us, and allow us to provide input on how to address their apprehension of collecting impact on insects and lepidoptera on parks, as well as to demonstrate some of the benefits we can offer. For the time being we will advise ALL MEMBERS not to collect on parks until the new policies and guidelines are in place, and wait until we receive the new statements directly from DNR. We hope to have information on this in the next issue. Members are also herein informed that collecting on State or Federal Public Land in the Keys has been severely restricted. We hope to provide maps in the next issue, since much of North Key Largo, Big Pine Key, and other areas are now regarded as OFF-LIMITS, except for very special research carefully controlled by permit through the Keys Region District Biologist.

We must realize the constrains that the park rangers and staff face in protecting restricted species, and urge our members to comply with the rules and regulations. If Any of you are caught engaged in collecting activity without the proper permits or permission in the sensitive Keys region, you deserve having the book thrown at you. Ignorance will not be taken as an excuse, and we would encourage park personnel to do their job in protecting the unique Keys fauna. We further urge them to apply the full measure of the law and to prosecute those who are caught. Most of the new land acquisitions on Key Largo have created serious problems for the DNR staff, mainly because no attempt was made to inform insect collectors. However, most of the new areas are posted and you should not be inside these areas unless you have made prior arrangements with the DNR staff. Butterfly collecting in particular will be excessively regulated in the future in the Florida Keys, apparently, due primarily to restricted species such as Papilio aristodemus ponceanus, Chlorostrymon maesites, Strymon acis bartrami, Anaea floridalis, and Eunica tatila tatilista. Specialized research is ongoing with P.a.ponceanus, and Big Pine Key populations of S.Acis bartrami and A.floridalis were monitored in 1989-90 and have been found to be very low numbers. We hope to have an update on the latter in the near future. We also urge your complete cooperation with DNR and USF&WS staff until further notice. Remember also that your individual actions reflect not only on yourself, but on the Southern Lepidopterists' Society as a whole.

The second issue addressed was the formation of a special committee to evaluate moths and butterflies for the revised updating of the Rare and Endangered Biota of Florida invertebrate volume currently be edited by Mark Deyrup at Archbold. Members Tom Emmel, Marc Minno, John Calhoun, Tom Neal, Dave Baggett, Linwood Dow, and Lee Adair contributed input for the next issue, due out late this year.

Forty-nine species of butterflies and skippers and ninety-one species of moths will be discussed. Perhaps DNR and USF&WS will view this as a benefit of invertebrate survey work done here in Florida by our members. We would hope for increased input and cooperative effort in the future.

Following the business meeting, we were treated to a slide presentation by Leroy Simon, who has done incredible things with his camera in recording life histories of many exotic species. John Calhoun presented an excellent talk and slide presentation regarding the habitats of Euphyes dukesi and Philosora catullus in Florida. Door prizes were presented to conclude the meeting, and we should note that William Nix - our youngest new protege - really benefited!! Actually, we know that we will ultimately benefit from his input from the Palm Beach County area in the future.

Members enjoyed an early breakfast Sunday morning. Some headed home while others spent the day collecting in or around the station. Several members collected in the Lake June area and Highland Hammocks State Park in search of Hesperia meskei. None were found, although the areas contained some prime habitat for them.

A list of the species of lepidoptera collected at Archbold Biological Station during the meeting can be obtained from Dave Baggett. It was a good meeting. Hope to see you at the next one!!

MEETING ANNOUNCEMENTS

SOUTHERN LEPIDOPTERISTS' SOCIETY MEETING ANNOUNCEMENTS

The 1991 field meeting to Costa Rica has been cancelled. There was considerable interest when it was first announced, however, only three members signed up to go when the deadline came. The officer decided to cancel the meeting and hold a spring meeting in Florida. The Holbrook Travel agency still has plans to go ahead with the trip as its own venture. I you are interested in going on this exciting adventure, contact the Holbrook Travel, Inc., 3640 N.W. 13th Street, Gainesville, FL 32609, Telephone 904-377-7111.

1991 SPRING MEETING IN GAINESVILLE, FLORIDA

The Southern Lepidopterists' Society and the Association for Tropical Lepidoptera will hold a joint meeting in Gainesville, Florida at the Florida State Collection of Arthropods, Friday, April 5, to Sunday, April 7, 1991. This will be the Southern Lepidopterists' Society spring meeting and the first annual meeting of the Association for Tropical Lepidoptera. The Florida State Collection of Arthropods will be acting as host for the meetings.

All meeting sessions will held at the Doyle Conner Auditorium. Both the FSCA and the new Department of Entomology, University of Florida, building nearby will be available for visiting on Friday, April 5. Tentative plans include a Friday evening welcome session, Saturday morning and afternoon sessions divided between the Association for Tropical Lepidoptera and the Southern Lepidopterists' Society, a Saturday evening banquet, and Sunday collecting trips. The Gainesville area offers a number of interesting collecting sites nearby and others within a one hour drive.

Gainesville is a college town, home to the University of Florida, and provides ample variety of accomodations, as well as numerous areas for local collecting.

Anyone planning to attend, wanting additional information or to present a paper should notify Jeffrey Slotten, 5421 NW 69th Lane, Gainesville, Florida 32606, phone 904-338-0721 evenings, or John Heppner, Florida State Collection of Arthropods, P.O.Box 1269, Gainesville, Florida, 32602 before March 15, 1991.

This will be an informative and exciting meeting. Make your plans to attend NOW!!.....

THE LEPIDOPTERISTS' SOCIETY ANNUAL MEETING

The 42nd annual meeting of the Lepidopterists' Society will be held in Tuscon, Arizona thursday, August 1 to Sunday August 4, 1991. Sonoran Arthropod Studies, Inc. and the University of Arizona's Department of Entomology will host the meeting at the Quality Inn University in Tuscon. For additional information contact: Steve Prchal, Sonoran Arthropod Studies, Inc., P.O. Box 5624, Tuscon, Arizona 85703, Telephone 602-883-3945.

CHANGES IN THE MEMBERSHIP — TOM NEAL

NEW MEMBERS

James K. Adams, 1702 Crow Valley Rd., #214, Dalton, GA 30720 Arctiidae, insect defenses, neotropical leps., esp. Lycaenidae, Limacodidae, Noctuidae, & Sphingidae

Ms. Freddy Arthur, P.O.Box 1234, Murrells Inlet, SC 29576 Butterfly rearing. Correspondence with other butterfly and moth breeders welcome.

Jim Davis, P.O.Box 442, Viburnum, MO 65566-0442 Moths.

Jeff Hooper, 5397 Paddy Ct., Norton, Ohio 44203

Clint J.A. Murphy, 424 S. Sappington Rd., Oakland, MD 63122

Gregory V. Myers, 205 Jay Ave., Sebring, FL 33872 Papilio troilus, the Pterourus group, thoas group, homerus group, and new world Papilionidae

David K. Parshall, 4424 Rosemary, Columbus, OH 43214 Arctic & Nearctic Lepidoptera, esp. Oeneis & Hesperia.

Bill Russell, 765 Yorkshire Rd. N.E., Atlanta, GA 30306 Roph. N.A. and Amer. Tropics, coll., exch., Photogrphy, and Butterfly mechanics.

Edmund H. Sallee, P.O.Box 38, Letohatchee, AL 36047 Lep.; coll., rear, sell, trapping, & photography.

Leroy Simon, 2215 Hialeah Dr., Leesburg, FL 34748 Roph., Saturniidae, rearing, photography.

John R. Westerfield, P.O.Box 188, Nordheim Rd., TX 78141

ADDRESS CHANGES & CORRECTIONS

Richard M. Gillmore, 5724 Clear Lake Circle, Sanford, FL 32771

Tom Neal, 1705 N.W. 23rd Street, Gainesville, FL 32605

THIS-N-THAT & OTHER TIDBITS

Robert A. Belmont of Naples, Florida put together a display entitled, "Butterflies - Fantasyflies: A Unique Collection of Butterfly Specimens and Butterfly Collectibles". The display was at "The Audubon House, Museum & Garden" in Key West, Florida. The display was beautifully put together. Congratulations on an outstanding effort.

Hermann Flaschka of Decatur, Georgia is recovering from surgery at his home. We wish him a speedy recovery.

Vernon Brou, our zone coordinator from Louisiana has suffered a heart attack. We wish him a speedy recovery. I am sure he would appreciate hearing from the membership.

Irving Flinkelstein, our Georgia zone coordinator, recently returned from a collecting foray to Brazil. He wrote that the collecting was great, however, it could have been better!

Several new books of interest have recently been published:

Familiar Butterflies, North America. Audubon Society Pocket Guide. 1990. Alfred A. Knopf, New York. Soft cover: 192 pp. (80 color photos). ($5.95)

Butterflies: How to Identify and Attract Them to Your Garden by Marcus Schneck. 1990. Rodale House, 33 East Minor St., Emmaus, PA 18098. Hard cover: 160 pp. (300 color illus., photos, & maps). ($24.95)

Butterfly Gardening: Creating Summer Magic in Your Garden. 1990. A Xerces Society & Smithsonian Publication. Sierra Club Books, Random House, New York. Soft cover: 191 pp. ($18.95)

NEWSLETTER UP-DATE

The deadline for the next newsletter, Vol.13 No. 1 is March 15, 1990. All articles and zone reports must be in the hands of the Editor for inclusion in the next newsletter. The following deadlines are for the balance of Vol. 13, June 15, 1991 for No.2, Sept. 15, 1991 for No.3, and Dec.15. 1991 for No.4. Please make a note of these deadline dates.

Articles for the newsletter are needed. If you have been working on a list of lepidoptera from your area and or have a long or short article on the lepidoptera of the southern region, please send it to the Editor.

RESEARCH REQUEST & MEMBERS NOTICES

BOOKS FOR SALE. I am selling a complete library of books and journals (over 300 volumes) dealing with butterflies and moths from the United States and other countries, especially Africa. The books, old and new, are in very good condition. For a complete listing, send a self addressed stamped business sized (#10) envelope (SASE) with $0.52 postage to: Eric H. Metzler. 1241 Kildale Sq. N. Columbus, Ohio 43229-1306. Inquiries (with SASE) about individual items are welcome.

FOR SALE: Light Traps, 12 volt DC or 110 volt AC with 15 watt or 8 watt black lights. The traps are portable and easy to use. Rain drains and beetle screens protect specimens from damage. For a free brochure and price list contact; Leroy C. Koehn, 2946 N.W. 91st Ave., Coral Springs, FL 33065.

RESEARCH REQUEST: Any butterfly records from the Florida Keys, even for common species. Data for Key Largo and Big Pine Key are rather extensive. For many of the other Keys very limited information on species composition and distribution exists. Any and all information would be greatly appreciated. Contact; Marc Minno, 303-18 Diamond Village, Gainesville, FL 32603.

RESEARCH REQUEST: 1.) Need specimens to study of Megathymus cofaqui from mid to southern Florida. I have examined the "type" specimen of M.cofaqui in the Allyn Museum and need additional specimens from these localities to determine the validity of applying the name "cofaqui" to the Florida phenotype. All material will be returned in two weeks from receipt.

2.) I have been studying Incisalia irus for 15 years. I have examined the "types" of Incisalia irus arsace in the USMN and located the Incisalia irus "type specimen" in the Paris Museum. I have examined several hundred specimens of I.irus from through out its range. I need additional specimens of the recently found Clay County, Florida population. I need fresh wild caught (have worn wild caught) and reared specimens. Can offer in exchange reared specimens of both I.i.arsace and I.i.hadros. Write to: Ron Gatrelle, 126 Wells Rd., Goose Creek, SC 29445.

CURRENT ZONE REPORTS

ZONE I TEXAS; Coordinator, Ed Knudson, 808 Woodstock, Bellaire, TX 77401

No report!

ZONE II ALABAMA, LOUISIANA, MISSISSIPPI, & TENNESSEE; Vernon Brou, 137 Jack Loyd Rd., Abita Springs, LA 70420; Bryant Mather 213 Mt. Salus Dr., Clinton, MS 39056; Mecky Furr, 7926 Cross Pike, Germantown, TN 38138.

No report!

ZONE 111 GEORGIA; Irving Finkelstein, 425 Springdale Dr. N.E., Atlanta, GA 30305

No report!

ZONE IV FLORIDA; Dave Baggett, 403 Oleander Dr, Palatka, FL 32077

Leroy Koehn and Jack Heinrich visited the Fakahatchee Strand on Sept. 22. The water level was normal and that it was extremely lush. They found Nastra neamathla, Lerodea eufala, Problema byssus, Erynnis zarucco, Staphylus hayhursti, Euphyes ruricola metacomet, Danaus eresimus tethys, Harpesia petrius, and Electrostrymon angelia. They ran UV/MV lights for moths at Collier Seminole State Park and found mothing to be rather poor even though the conditions were near perfect. They did collect Madoryx pseudothyreus, xylophanes pluto, Eacles imperialis, Antheraea polyphemus, Cosmosoma myrodora, Pareuchaetes insulata, and Eupseudosoma involutum floridum.

Leroy Koehn and Jack Heinrich visited Sanibel Island on Nov 10, 1990. The first cold front of the season had just past through sending temperatures down to a rather chilly low 70's. The high wind made butterfly collecting difficult and moth collecting impossible.

Species of interest that were found included Hemiargus thomasi bethunebakeri, H.ceraunus antibubastus, and Electrostrymon angelia. The latter appears to be moving west and north up the gulf coast. It was present in the Fakahatchee Strand in Sept. and Koehn found it rather common on Marco Island in mid-May.

Leroy Koehn visited Jonathan Dickenson State Park on Oct 6 and found Hesperia attalus slossonae, Polites themistocles, Atrytone arogos, Atrytonopsis loammi, and Nastra neamathla. Moth collecting was excellent; Pseudocharis minima, Calidota laqueata, Holomelina laeta, Schinia sanguinea, S.fulleri, S.trifascia, and Lapara coniferarum.

Dave Baggett and Dale Schweitzer collected near Lake Delancey, Marion County on October 12 while on their way down to the fall meeting at Archbold. They were searching for butterflies and Schinia moths. They found Hesperia attalus slossonae extremely common. They also collected a single male of Atrytone arogos which confirmed a third brood in Florida. Schinia bina and Schinia arcigera were also collected.

Marc Minno is currently working on the lepidoptera listings for the Archbold Biological Station and presently has the list well over 1100 species. This is probably the largest list nearing completion for any locality in Florida. While at Archbold, Southern Lepidopterists added the following to the updated list: the pyralids Fumibotys fumalis and Pyrausta signatalis, the geometrid Iacparia zalissaria, the notodonids Nystalea indiana and Datana robusta, and the noctuids Physula albipunctilla, Papaipema stenocilis, Elaphria deltoides, Leucania infatuans, L.subpunctata, L.senescens, Iricholita signata semitropicae, and Schinia scissoides. A number of additional species not on Marc's list were taken and subsequently added after checking the original descriptions. Several of these species are known from Florida but are not in the current MONA checklist.

Dennis Profant, formerly a park ranger here in Florida, recently had his list of Lepidoptera from Blue Spring State Park in Volusia County Published in J. Res. Lepid. in an article entitled "The Lepidoptera of a central Florida sand pine scrub community", which illustrates a number of species and provides comments on 633 species. Dennis is now back in Ohio, and we miss his regular input on Florida species. However, our loss is Ohio's gain, and we are sure Eric Metzler will keep him busy! Thanks Dennis, for aiding us in our understanding of Florida leps.

John Kutis of Belleview continues his outstanding work with moths and has become an excellent preparator of micros, something he himself scoffed at undertaking only a year ago. He has already contributed in the discovery of several species new to science and has greatly added to our knowledge of numerous moths here Florida during his three years as a lepidopterists. Some of his more interesting records include:
Chytonix sensilis, Ocala National Forest, Marion County in October. Thaumatographa jonesi, several localities in Marion and Lake Counties during October. Anorthodes tarda and Iricholita signata from Lake County on 12 November. Antaeotricha unipunctella, Oncocnemis saundersana, and Schinia septentrionalis on 20 November from Holt, Santa Rosa County. Euplexia benesimilis and Mextaxaglaea australis from Lake Charles, Marion County on 13 December. Pyreferra pettiti from Santos, Marion County on 17 December. Lithophane lemmeri, Metaxaglaea violacea, and Nemopogon rileyi on 22 December near Sumatra in the Apalachicola National Forest in Liberty County.

Dave Baggett and Dale Scweitzer collected in the Fakahatchee Strand State Preserve after the Archbold meeting on October 15th. They found Cocytius antaeus, Isoparce cupressi, Epipagis forsythae, Petrophila drumalis, Phrygionis argentata, Pareuchaetes insulata, Gonodonta nutrix, and Hemeroblemma opigena. They also found a larva of the Sphingid Eumorpha labruscae feeding on Ludwigia ssp., a new host record for this moth. However, the most interesting find was a Glyphipterigid moth which is either a new species or a tropical species not yet known from the U.S.

Dave Baggett collecting at his home in Palatka keeps adding to the Putnam County list, a few of the most interesting records include: Xestia dolosa on 3 December, this is an extremely scarce moth in Florida and represents a significant range extension southward. Meropleon cosmion was common on warm December nights this year. Gnorimoschema gallaesolidaginis on 27 December.

Jeff Slotten reported finding a Papaipema speciosissima at the lights of a convenience store near Melrose, Putnam County in October.

ZONE V VIRGINIA, NORTH & SOUTH CAROLINA; Bob Cavanaugh, P.O. Box 734, Morehead City, N.C. 28557, Ron Gatrelle, 126 Wells rd., Goose Creek, S.C. 29445.

Gatrelle filed the following report for South Carolina:

June 8: Dirt Rd. OC 1421, at Bull Swamp off Hwy 172, Orangeburg County; Hesperia meskei a fresh female. New County Record.
Off HWY 394 1 mi. E. of jct. Hwy 3, Orangeburg County. Herkenclenus titus mopsus, New County Record. Near Aiken State park, Aiken County, Strymon melinus, Harkenclenus titus mopsus, Satyrium edwardsi, Hesperia meskei, and Neonympha septentrionalis ssp in open dry pine/oak woods (Neonympha areolatus is nearby in swamp.).

June 19: Mitoura hesseli male eclosed from ova collect on April 20-26 from Aiken County locality. Gatrelle noted that over the last three years that all the M.hesseli reared from ova from the Aiken County locality have taken exactly 60 days from ova to adult. None have ever over-wintered. However, of the several batches of M.hesseli reared from Hoke County, NC, approximately 50% over-wintered from the spring brood.

June 29: Colleton County, Jacksonboro, Westvaco Park of Hwy 17; A fresh female Cercyonis pegala was collected in heavily wooded swamp (unusual habitat), he also found Lethe creola and Lethe portlandia.

July 31: Spartanburg County, on Hwy 19 in New Ellenton. Several pupae and larvae of Megathymus harrisi were collected (first specimens found in SC and Aiken County since original state record male in 1976). Returned to New Ellenton on Aug. 14 and found 3 additional pupae. First individual eclosed Aug 16. Total of 7 male and 3 females, all M.harrisi phenotype. Jeff Slotten originally found this location on a trip to South Carolina to arrange for the 1990 summer field meeting with Charlie Watson. Slotten feels that a search in April or May could possibly reveal a first brood of Megathymus cofaqui in South Carolina. However, this can only be verified by collecting spring individuals.

Aug. 11: Colleton County, Edisto Island, searched island for Megathymus cofaqui without success. However, numerous tent of Megathymus yuccae were located.

Sept. 17: Spartanburg County, Hwy 29 1 mi. E. of Jct. of I-85. Eurema nicippi and Strymon melinus were the only butterflies collected. Calhoun County, Hwy 21 near I-26. Strymon melinus and Nastra lherminier were found. Berkely County, near Jct of I-26 and Hwy 17A. Cercyonis pegala and Neonympha areolatus were collected. Both were well worn.

Oct. 4: Charleston County, off Hwy 17, 2 Mi. S. of Ravenel. Papilio palamedes, Agralis vanillae nigrior, Junonia coena, Pyrisitia lisa, Eurema nicippe, Euphyus dion alabamae(Very worn), Hylephila phyleus, Strymon melinus, Urbanus proteus, Parrhasius m-album, Calycopis cercrops, Calephelis virginiensis, and Atlides halesus. Over 100 ova were obtained from a female Atlides halesus. 47 emerged to first instar, all 47 pupaed by Nov. 16.

Oct. 6: Berkeley County, Goose Creek, Phyciode tharos and Copaeodes minima were found.

Gatrelle noted that by mid-November the following species were still flying about his home in Berkeley County; Junonia coenia, Agraulis vanillae nigrior, Euptoieta claudia, Vanessa atalanta, Urbanus proteus, Pyrgus communis, Hylephila phyleus, Eurema nicippe, Prysitia lisa, Phoebis sennae, Papilio palamedes, Strymon melinus and Calycopis cercrops.

Gatrelle filed the following report from North Carolina:

July 4: Macon County, Jones Knob, Speyeria diana, S.aphrodite, Erynnis brizo(late), and Epargyreus clarus. The latter was absolutely , unbelievably, abundant, to say the least!

July 5: Macon County, Scaly Mtn. Polygonia faunus smythi(fresh), Parrhasius m-album, Speyeria aphrodite, and Thorybes bathyllus.

July 6: Macon County, Jones Knob. Cercyonis pegala carolina, and Satyrium falacer falacer. The latter was found in heavy wood, none were seen along the roads. Macon County, Scaly Mtn. Celastrina ladon ssp, and Speyeria aphrodite were found.

July 8: Macon County, Jones Knob. Polygonia faunus smythi and Strymon melinus were taken. Macon County, Scaly Mtn. A single male of Speyeria diana was taken, collecting was cut short when it began to rain. According to the locals, this was the first rain in four weeks.

July 30: Macon County, Scaly Mtn. Speyeria aphrodite (worn), and Lethe anthedon(fresh) were collected. Macon County, Jones Knob. Feniseca tarquinius, Speyeria aphrodite, S.cybele (worn), S.diana (worn), Cercyonis pegala carolina, and Lethe anthedon.

Gatrelle commented that his research to date on Speyeria aphrodite appears to indicate that S.aphrodite from northern North Carolina, Tennessee are the nominate S.aphrodite, only larger, sometimes much larger when compared to the topotypes from the New York City area. On July 30 he visited the mountains in the Rabun County, Georgia area which borders Macon County, North Carolina in search of S.aphrodite without success. S.aphrodite may only be a stray in north Georgia. However, the material from the mountains of Macon County, North Carolina appear to be a distinct phenotype due to their isolation in the higher elevation above and about 4,000 feet in disturbed areas.

ZONE VI ARKANSAS; Mack Shotts, MD, 514 W. Main St., Paragould, AR 72450.

No report!

PARTING SHOTS

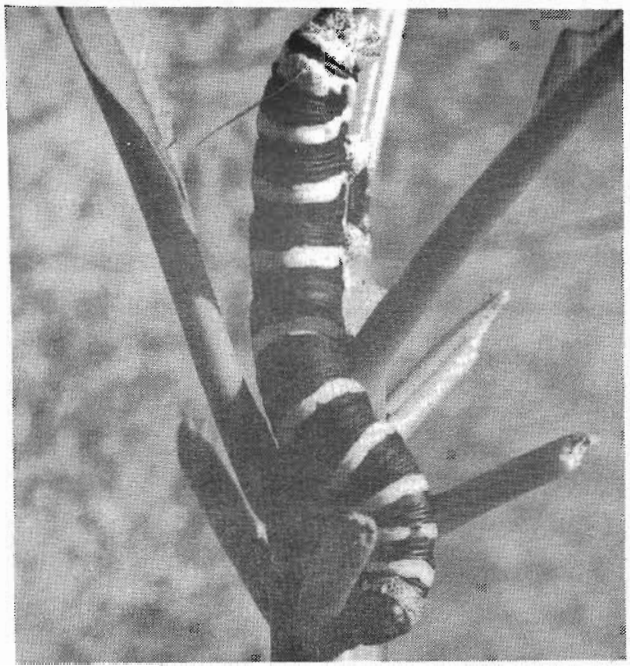


Fig.#1 Last instar larva of Pseudophinx tetrio on Fangipani(Plumeria rubra)Lee County Naples, Florida. The larva is purplish black and yellowish white stripes with a bright red head and a hair like horn. Photograph by Bob Belmont.

Fig.#2 The last instar larva of Incisalia irus arce feeding on Lupinus perennis, Clay County, Middleburg, Florida. Photograph by Jeffrey Slotten.

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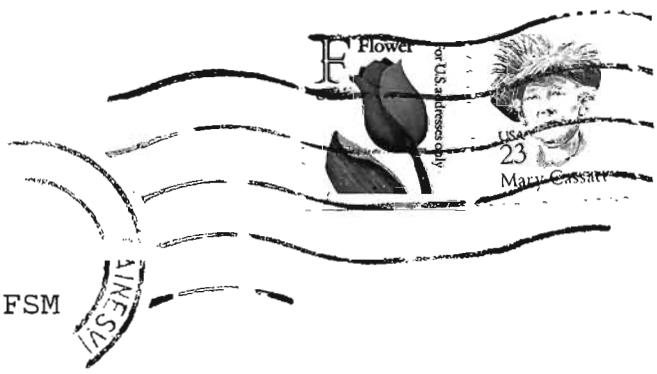

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