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Practical advice on the rearing of Saturniid caterpillars with notes on specimen preservation and parasitoid research

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Abstract
This article offers information for the successful rearing of Saturniid caterpillars. Several different techniques are described, including Sleeving, closed container and vented container. The importance of proper preparation and preservation of well hydrated food sources is discussed. Larval and ova preservation techniques for research and DNA analysis are discussed, as well as the contingencies required when larval parasitoids are detected.

Keywords: Saturniidae, rearing techniques, caterpillars, pupae, parasitoids, larva

1. Introduction
There are many different ways to raise saturniid caterpillars in captivity. In order to maximize the opportunities for success, one must ensure the presence of high quality, well hydrated food plants, proper environmental conditions and enclosure strategies suited to the species being reared. Maintaining species specific environmental conditions to foster the growth of the caterpillars through their various instars is crucial to minimize mortality rates and optimize successful pupation.

2. Materials and Methods
Specimen collection: The most common technique for the collection of ova for rearing is to capture fertilized, gravid females and place them into paper bags to allow them the opportunity to oviposit. Once oviposition has completed, the resulting ova can be counted, characterized, and placed into enclosures to await hatching. It is very important to capture data at all stages of the rearing process and to record these data in a fashion that allows them to be compared across subsequent rearing projects. Relative to ova, document the shape, color, size and manner of placement. You may need to provide some suitable host plant in order to encourage the females to oviposit. Some species, such as the Hemileucinae, prefer to lay their eggs in rings around twigs – typically on the host plant. Others will simply scatter eggs in small groups on the inside of the bag. It is important that you understand the species preference prior to confining the female.

In the event that no fertilized females can be located you can go into the field specifically looking for ova on the anticipated host plants. This technique is typically best when the species host plant is easily identified and visually accessible by those hunting. Knowing the host plant biological specifics will make the task of locating ova significantly easier.

One other technique that is commonly used is to go looking for larvae in the field on the host plants. This technique can be very successful, especially with species whose larvae are gregarious. When searching for caterpillars, one should carefully watch for telltale signs of feeding activity on the target host plants. This technique will have the greatest chance of yielding specimens that have been naturally parasitized. Understanding the parasitoids of the species that we rear is extremely important.

Several books cited in our references are invaluable in the identification and initial research into life cycle of captured caterpillars. We recommend the works by David L. Wagner [10, 11], Tuskes, P. M., Tuttle, J. P., & Collins [8], Triplehorn, Charles A., and Norman F. Johnson [7], and Gardiner, Brian [2].

When ova or caterpillars have been located, record the location where they were found, the date, the host plant on which they were located, and a good general description of the

circumstances. Recently, several GPS enabled smart phone applications have been created that allow for photography and metadata capture with GPS tags while in the field. The use of these cooperative natural cataloging applications allows data to be made available to others with similar interests. The authors favor, and are active reporting members of a community which uses the application – iNaturalist. This free application, which is available for download for a variety of different mobile devices, and is accessible from desktop computers (http://www.inaturalist.org), allows the capture and sharing of geo-tagged field data with the ability to add photographs, audio clips, and notes. Participants can receive ID and range confirmations, participate in community research projects and request identification assistance from the community in general. The use of this "social media" style reporting application is a great way to get early information published and should be considered a critical step if you do not intend to otherwise publish your results in a peer reviewed journal. The growing importance of the citizen scientist and the emerging community based resource sharing applications are important new trends in the area of research.

Once ova or caterpillars have been collected and identified the next step is the task of researching the species and its larval requirements. This can be made easier if the livestock was obtained on an identified host plant. If the supply of the natural host plant in your area is problematic, then experimentation with alternative food sources that are closely related to the natural host plant can be worthwhile. There are resources that list the known host plants for each species along with identified alternative host plants which can be used in the rearing process, the authors recommend Stephen Stone's "Foodplants of world Saturniidae" [6]. Alternative food sources are frequently successful; however, in some instances due to the differences from the naturally occurring host plants, you may have higher-than-expected mortality, or adult moths which are noticeably smaller than naturally reared specimens. In the references section of this paper we list several resources that are available to help determine host plants and any known alternates.

Rearing Containers: We start batches of ova in small vented plastic containers; following the rule that all ova from a single batch are reared together in their initial instars. (Fig.1A) Once our containers are set up we do our best to mimic the natural temperature of the original location where the ova were collected. If we are collecting locally we will typically house the ova outside in a sheltered area where the temperature is similar to the temperature in the environment where the ova were obtained. If our local weather conditions are warmer than the location where the ova were obtained we will use minimally vented holding containers housed within a small dormitory style refrigerator to re-create those temperatures (Fig.1B). These holding containers are vented with a small hole in the top and on all four sides to allow marginal airflow over the ova. Regular misting with non-chlorinated water will help keep the ova viable. We use a small mister to get moisture into these containers on a weekly basis. Refrigerators naturally desiccate living organisms so adding moisture is important. The key point is to maintain the temperature within the species' anticipated requirements. We typically remove the containers when the average temperature in their native environment begins to approximate our rearing location average temperatures. All holding containers with ova are monitored for the first emergence of caterpillars while in refrigeration. If caterpillars are seen within the holding container, they are removed from refrigeration, placed into a rearing container and well hydrated food plant materials will be added.

Fig 1A: Vented rearing containers of varying sizes

Fig 1B: Minimally vented refrigeration holding containers of varying sizes for ova and pupae

Some species of Saturniid caterpillars will consume all or part of the eggshell from which they hatched. If the species is known to practice this behavior, it is extremely important that you do not separate them from the eggshells until they have transitioned onto the host plant. Never put unhatched ova into the same containers with cut food plants. The cut leaves generate carbon dioxide and in a small closed container you could suffocate the ova or first instar hatchlings with a lethal carbon dioxide level. (Villiard, page 8) [9]

Food plant preparation: There are several important steps in preparing food plant for consumption by the caterpillars. You want to harvest your food plant in small enough quantities to just meet your caterpillar's requirements for a single day. In early instars this could be as simple as harvesting one small branch and in later instars and particularly with large batches one will need significantly larger quantities of host plant on a daily basis. After obtaining the host plant you should carefully inspect it for the presence of other invertebrates that might cause harm to your caterpillars, paying close attention to spiders, ants and other insects known to feed on caterpillars. The plant material should be thoroughly washed before introduction. The cut ends of stems should be re-trimmed underwater, typically in a small bowl or bucket, with the stem cut at an angle (Fig. 2). Cutting the host plant while underwater allows the plant's natural vascular system to continue to draw water by preventing air from blocking the vessels, and thus maintains well hydrated leaves for a longer period of time. After the stems have been cut underwater, they should be placed into florist's cut flower vials (Fig. 3) which

have been topped off with clean water. Keeping the provided food plants hydrated will prolong the life of the food plant and provide better nutrition and hydration for the caterpillars. (R. S. Peigler, personal communication, July 2014)

Fig 2: Sharply angled cut on stem of food plant

Fig 3: Using florist cut flower vials for food plant hydration

Fresh food plant should be added to each rearing container as necessary. You should replace the food source when there is very little host plant available within the container or if the host plant appears to be drying out or degraded. Keeping well hydrated high-quality food available to your caterpillars will typically lessen your mortality rate and give you better quality reared adults. "The most successful breeders known to me completely change the fodder every two days (three days at most). The containers used for breeding or thoroughly cleaned and the water used for sprinkling fodder is changed. Every larva is examined. Wilted leaves lead to intestinal problems." (Meister, page 38) [4] Some caterpillars require younger, tenderer leaves in their early instars. This is particularly true in the Hemileucinae that hatch in early spring. The early instar caterpillars require young leaves that are not yet sclerotized. Older leaves, which have become hardened, are described as sclerotized. For some caterpillars the ideal food source will be young leaves or even the host plant's reproductive structures such as catkins. If you're having difficulty in getting your caterpillars to eat the host plant you should try alternatives such as younger leaves, catkins (if available), or even an alternative food source from a closely related food plant.

When adding food plant to the enclosures you should not manually detach caterpillars from older materials, especially if they are in a dormant – non-feeding stage just prior to molting. "The caterpillar spins a pad of silk on a leaf or other support. This pad is very hard to distinguish when you are a beginner because it is so tiny. The caterpillar attaches his claspers to the pad so that he has purchase from which to pull out of his old skin. This is one of the main reasons why, when transferring caterpillars to fresh food, you should not pick them up with the brush or pull them off their leaf. If you cut around them, enough of the pad will be left to afford a purchase and the molt will continue normally." (Villiard, page 8) [9]

When raising caterpillars is important to keep the enclosures clean on a daily basis. Depending upon the size of the batch and the size of the species the accumulation of frass can be significant. We typically clean each enclosure on a daily basis which allows the opportunity to observe the caterpillars, record our observations, and look for unwanted predators or sickly or dead specimens. In order to avoid the spread of disease among your caterpillars, any sickly, excessively dormant or limp specimens should be removed from the batch and housed in quarantine containers with fresh food plant and an appropriate moisture level. We typically house quarantined specimens in solitary to avoid passing any disease on to the larger brood. Caterpillars can also exhibit behavior such as noted above if they have been parasitized. If this is the case, solitary confinement will ensure that any adult parasites do not emerge to a full buffet of uninfected caterpillars within your batch. After several days in quarantine if the caterpillar begins feeding normally or in general becomes active and appears to be in good health it can be returned to the batch from which it was originally removed. Any specimens which fail to thrive in quarantine can either be dispatched or preserved as a part of your larval collection.

If you encounter dead caterpillars within the container it is extremely important that you remove all caterpillars, their food plant and clean the container thoroughly. We will typically wash the container out with soap and water and then wipe it down with a 25% solution of Clorox bleach. After treating it with bleach, we will rinse the container again to be sure that the bleach has been removed. Keeping containers clean will help to avoid the spread of diseases within your broods. We will typically replace all the food plants within a container that has dead caterpillars in it. This is particularly true if the caterpillars are found lying dead at the bottom of the container surrounded by a watery exudate. Some caterpillars will die while attached to limbs and these should be removed and the cleaning protocol engaged to avoid transferring pathogens to healthy caterpillars within the enclosure.

Sleeving: One way to avoid many of these issues is to sleeve your caterpillars. (Fig. 4) Sleeving is the process of confining caterpillars to a host plant in the wild by using a fine mesh sleeve surrounding one or more limbs of a host plant. Sleeving is a particularly good strategy especially in the early instars for most caterpillars. Creating the sleeve is actually quite simple; you purchase a fine mesh fabric similar to the mesh used on most insect nets and create a long hollow cylinder sewn together at the edges along its length. Locate a suitable host plant and use string or wire ties to group several limbs together. Place the sleeve over the limbs and secure one end to the limbs with cord or wire ties. Place the caterpillars or ova into the enclosure and then use a piece of string or wire tie to close the other end. You will then need to monitor the sleeve on a daily basis to ensure that fire ants or other predators have not discovered your caterpillars. If you notice fire ant mounds near the base of the tree where you have sleeved your caterpillars it is advised that you use a commercial fire ant treatment to eliminate the colonies as they may begin to prey

upon your caterpillars. Monitor the availability of food plant within the sleeve and if the caterpillars begin to run out of leaves you can remove the sleeve, capture the caterpillars and then move them to a new set of limbs.

Fig 4A: Sleeve in place on *Quercus nigra* L

Fig 4B: *Anisota fuscosa* Ferguson 1971 caterpillars in sleeve.

Rearing caterpillars in this fashion requires significantly less work on a daily basis but places your caterpillars at risk if the environment is not entirely under your control. You should not consider sleeving caterpillars unless the location is remote, private, and you have permission from the property owner.

You should never consider sleeving caterpillars in an environment where they are not native. "One next should take precautions that foreign species, species that don't belong there, do not expand into the local environment." (Meister, page 37) [4] If caterpillars were to escape and become established in a new environment they may cause significant environmental and economic damage. The introduction of species into new environments where they are not traditionally found should be avoided at all costs. We frequently rear caterpillars to hand out to local elementary and middle schools but only do so using species that are found in the area where the schools are located. This allows the teachers and their students to release the adults into the wild without fear of introducing a new species into the local environment.

Batch management: Avoid overcrowding when rearing caterpillars. Many species are gregarious and will cluster together for protection; however, in a non-natural environment such as a rearing container you increase the risk of losing the entire batch to disease or parasitoids if you keep them together. We typically follow a guideline that suggests the batches should be split in half at each instar occurrence. You could easily tell when your caterpillars are changing from one instar to another. There will be head capsules in the frass at the bottom of the container and you will see the shed skins in the container as well. Many species also change appearance between instars, not just an increase in overall size or length but an actual physical difference. Caterpillars of *Citheronia regalis* (Fabricius, 1793), for instance, are completely different in sixth instar. The ultimate instar is bright green in general skin color but the five previous instars are brown. At the time of each transition from one instar to another it is a good practice to divide the batch in half. We also increase the size of the container in which we rear the caterpillars at the instar changes. We use a small sealed container for the first and second instars. We use a new container typically twice the size for the third and fourth instars, and transition to our largest container for the two final instars. We have recently begun to use screened cages for our caterpillars that are third instar or later (Fig 5A). These wire cages provide a tremendous amount of ventilation and are constructed with the simplest of materials. We use aluminum window screening and cookie tin. In the simplest of fashions, a roll of aluminum window screening is cut with 1 – 2 inches of overlap to fit within the top and bottom of the cookie tin. We then secure the screen to itself by cutting and attaching both ends with screws to a thin strip of wood, typically a wooden measuring stick that is 2 inches wide by 18 inches long (Fig. 5B). We can then place the top of the cookie tin on top of the roll of screening and then insert the bottom of the cookie tin as well. This arrangement is easy to clean, and can typically be created for less than ten dollars. Overall, these large screened enclosures provide plenty of air for circulation, and are superior to closed containers for rearing. We still use smaller plastic containers for the early instars as they can easily crawl through the screening in our rearing cages. If we are concerned about the humidity level of the caterpillars then we will typically raise them in successively larger sealed rearing enclosures or if they are in a screened enclosure we will be sure to mist regularly to keep the foliage moist and the surrounding atmosphere sufficiently humid.

Fig 5A: Screened rearing container for mature larvae.

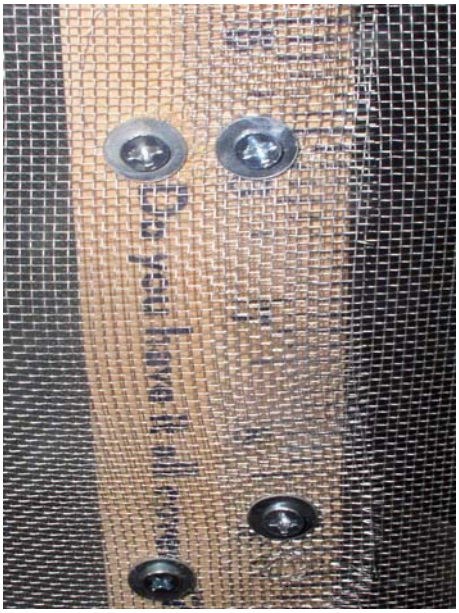

Fig 5B: Detail of the screen attachment assembly.

Maintaining caterpillars in an environment that closely matches their natural environment during the rearing process will generate the best results. Careful study of the weather patterns, humidity and other environmental factors in the caterpillar's home range will give you much needed clues as to how best to raise your larvae. In order to reduce the incidence of mortality from pathogenic diseases, caterpillars (especially species from the western United States) being reared in cages should be exposed to sunlight for at least one hour per day. [13]

Pupation: When caterpillars have reached the end of the larval phase they will typically cease feeding and began to wander about or gather in groups on the floor of their enclosure. This is typically the first sign that they are about to pupate. Some species pupate underground with a minimal or non-existent silken cocoon. Some species will instead create silk cocoons in the branches of their host plant. It is important that in either case that you do not handle them until the pupae have had a chance to fully mature and harden their exoskeletons. Handling a freshly pupated specimen can easily cause damage or even death. Ground pupators should be kept in minimally ventilated holding containers with a small amount of non-chemically treated potting soil in the bottom. The potting soil should be baked in an oven at 250 degrees Fahrenheit for an hour to ensure a relatively sterile substrate with no bacterial or fungal issues for the pupa. This heat treatment should eliminate most fungi and fungal spores. These containers should be misted regularly to maintain the humidity level and may need to be kept in a refrigerator for a period of time to allow the pupa to complete metamorphosis.

In temperate climates where there is typically a winter or cooler season between the pupation and eclosion events, refrigeration is important (Fig. 6a). In either case the container should be ventilated and should have vertical surfaces for the newly enclosed moths to climb up so that they can complete the process of expanding their wings. Be sure to label the enclosures with all the necessary data to link the pupae to their rearing data (Fig. 6 B). We will typically use sticks or strips of cloth to assist them in gaining a perch to allow gravity to assist them in the expansion and hardening of their wings. If the species spins a cocoon upon pupation we will typically attach that cocoon to a small tree branch located inside a wire enclosure (see Fig 7A & 7B). When the adults emerge they can either cling to the branch where the cocoon was attached or they will be able to climb up the screened sides of the enclosure. Keeping a watchful eye on your pupae particularly as you approach expected eclosure periods is very important.

Some pupae will skip a year and emerge in a different calendar period as a survival technique. This is a population maintenance strategy that guards against all the pupae from particular species emerging at the same time and encountering circumstance where food plant is in limited supply and therefore the survival rates of any caterpillars will be greatly reduced. Having some pupae eclose at a different time is simply a way to hedge the bet and ensure survival of the species. You should be prepared that some of your specimens may enter an extended diapause and take significantly longer to eclose than others.

Fig 6A: Minimally vented holding containers in the refrigerator**.**

Fig 6B: Data tags on the containers in refrigeration**.**

Fig 7A: *Callosamia promethea* (Drury, 1773) cocoons in wire screened eclosure container also see Fig. 5A.

Fig 7B: *Callosamia promethea* cocoons detail – attached to branches with wire ties.

3. Observation and results

Data keeping: It is extremely important that you keep track on a daily basis of all the statistics about the batches that you are rearing. We maintain an account of hatched versus unhatched ova, total number of caterpillars, any incidents of mortality or parasitism, or pupation events. We have developed an Excel spreadsheet to help us keep track of our batches. We keep the same spreadsheet for every rearing project and file them on our hard drive in folders organized by species. This allows us to quickly refer to previous rearing efforts in the event that we have questions about current projects.

These spreadsheets are available via download on the East Texas Natural History Collection website [http://www.etnhc.org/in-print/jezs-articles/]. You may use these to track your own rearing efforts. The spreadsheet is an extremely easy way to keep track of a batch of caterpillars and our observations. When raising a species for the first time, we will also keep a written log of daily events. This log contains detailed observations about the caterpillars and their behavior or anything unusual or unique that we see occurring. The spreadsheet and the log when used together provide significant information that could prove invaluable in subsequent rearing efforts (Chart 1).

Rearing History Sheet — Copyright © 2004 by rjnjr.

Mother Data:				Summary Data		
				Description	Date	Count
Order:	Lepidoptera			Ova Count:	9/16/2004	118
Family	Saturniidae			Days to first Hatch:	9/25/2004	9 days
Genus	Anisota			Days to last Hatch:	9/27/2004	11
Species	discolor	Day	Time	% hatch	116/118	98%
Capture Date	9/12/2004	Sunday	9:00 PM	Start 1st Instar	9/25/2004	116
Locale	USA			Start 2nd Instar	9/29/2004	113
Nearest City	Montgomery	Montgomery County		Start 3rd Instar		
Leg	Christopher Lowery			Start 4th Instar		
How	Attracted to Porch Light			Start 5th Instar		
Habitat	Rural Residential area with greenbelt			Start 6th Instar		
Confinement	9/12/2004	Mason Jar - Paper towel		1st Pupation Date		
	9/13/2004	Brown Paper bag		End Pupation Date		
Results	9/12/2004	No Ova		% Pupated		
	9/13/2004	No Ova		First Eclosion		
	9/14/2004	Approx. 100 Ova		Last Eclosion		
Date of Death	9/15/2004	Disposition:	Pin for ID	% Eclosed		
Food Plant Used	White Oak - Quercus alba					

Days	Date	Stage	Count	Fresh Food	Cleaning	Comments
1	09/15/04	Ova	118	None Needed	N/A	Still in container
2	09/16/04	Ova	118	None Needed	N/A	Still in container
3	09/17/04	Ova	118	None Needed	N/A	Still in container
4	09/18/04	Ova	118	None Needed	N/A	Still in container
5	09/19/04	Ova	118	None Needed	N/A	Still in container
6	09/20/04	Ova	118	None Needed	N/A	Still in container
7	09/21/04	Ova	118	None Needed	N/A	Still in container
8	09/22/04	Ova	118	None Needed	N/A	Still in container
9	09/23/04	Ova	118	None Needed	N/A	Still in container
10	09/24/04	Ova	118	None Needed	N/A	Still in container
11	09/25/04	Hatching Ova	98	White Oak	N/A	Starting to hatch
12	09/26/04	Hatching Ova	116	N/A	N/A	Very small on leaves
13	09/27/04	First Instar	116	N/A	N/A	Very smal on leaves
14	09/28/04	First Instar	116	N/A	N/A	On leaves in large groups

Chart 1: Rearing data sheet for *Anisota discolor* Ferguson, 1971

4. Discussion

Preserving caterpillars: If you wish to keep voucher specimens of the caterpillars that you are rearing (Fig. 7A), particularly if you wish to keep a sample of the species at each instar, you should follow these guidelines. It is important that caterpillars be immersed in boiling water for about 1 minute before being added to alcohol. This strengthens the caterpillar's body structure and kills off any bacteria within the gut which could cause darkening to the specimen even while it is being preserved. "Proper preservation is critical because the numbers and relationships of setae and crochets are difficult or impossible to see on shriveled or darkened larvae; the use of KAAD or hot water for killing, and other solutions and procedures as outlined in the "techniques" section that distend and straighten larvae is essential." (Stehr, page 288) [5]

You may need to change the alcohol after a couple of days as many caterpillars will leech chemicals that stain the alcohol thus obscuring the coloration of the preserved specimen. The timing of when to preserve the caterpillars is extremely important. You should not preserve freshly molted caterpillars but instead wait at least 1 – 2 days to give their skin, colors, and external features a chance to mature before preserving the specimen. As with any scientifically collected specimens data labels must be created. You can use India ink, a number 2 pencil, or technical pen to write on acid-free card stock and include that label inside the glass vial with the specimen (See Fig. 8A). Caterpillars should be preserved in 70% - 80% isopropyl or ethyl alcohol in glass vials with full data. These vials should be kept out of light, in a dry environment. You should check alcohol levels on your preserved specimens and augment or replace alcohol as needed.

Fig 8A: Preserved caterpillar in 70 – 80% ethyl alcohol.

Preserving specimens for DNA analysis: Caterpillars or adult moths can be an excellent source of DNA but require special handling procedures. If you are intending to use caterpillars for DNA extraction you will need glass or plastic vials, 100% ethanol, forceps and card stock. Caterpillars for DNA extraction should not be boiled. For best results place them while living directly into the 100% ethanol. Caterpillars can be kept dormant in a refrigerator for several days, if necessary until they can be preserved in 100% ethanol. All specimen vials should contain a data label that identifies the location where the specimen was obtained, the date it was placed into the alcohol, genus species (if known), and host plant on which the caterpillar was obtained or general notes on the habitat. Each data label should also contain the name of the collector (See Fig. 8B).

Fig 8B: Preserved caterpillar typical data label.

You should not mix caterpillars of different species in the same container, and be sure to thoroughly clean your forceps between specimens to avoid cross contamination. The specimen vials with 100% ethanol in them should be maintained in a freezer until you are ready to extract the DNA. Typically 1 or 2 specimens will provide enough DNA for analysis purposes. If you are using DNA analysis to study differences between geographically distinct populations you should preserve a few caterpillars from each population in a vial of 100% ethanol – do not mix them with specimens from other populations.

Ova and pupae from Lepidoptera are typically not considered to be good sources for DNA analysis. Caterpillars and adults give the best overall DNA results. Caterpillars can be used in any instar; however, you should allow first instar caterpillars or recently molted caterpillars several days to complete the hardening of their skin before immersing them in alcohol. If you are providing adults for DNA analysis, the same overall procedures apply. Adult moths should be placed while living into a vial containing 100% ethanol. Place one adult into each vial using forceps, add a data label and maintain in a freezer until ready for analysis. Ethanol will not freeze so this method best maintains the DNA integrity for subsequent analysis.

Preserving Pupae and Ova: We also typically preserve ova and pupae as well as the caterpillars. Ova and pupae can readily be preserved by simply placing them into 70 - 80% ethyl or isopropyl alcohol in sealed vials with data labels just as described above with caterpillars. When preparing adult moths to go into Cornell drawers for permanent display we will include the shed pupal shell on the same pin using a large card stock point and white glue. This can be particularly helpful because it marries the individual in the adult phase with its pupal skin. [See Fig. 9]

Fig 9: Reared adult specimens of *Anisota pellucida* (J.E. Smith, 1797) with pupal cases included. Specimens courtesy of Dr. Richard S. Peigler.

Parasitoids: Some researchers will also pin or otherwise associate parasitoids in various stages of their life cycle with either pinned specimens or keep them separately in vials of alcohol. Research into the regional parasitoids of Saturniid caterpillars is extremely important. Many common species may have parasitoid relationships that are as yet unknown to science. The most important point with parasites is to capture as many stages in the life cycle as possible. It is sometimes difficult to gather larval phases of parasites but pupal cases and adults are typically very easy to gather. You are not likely to have the opportunity to study parasites if you do most of your rearing from ova. Your best chance to understand the parasites of a species is to collect caterpillars after they have already completed several instars in the wild. Careful preservation and documentation of parasitoid activity can lead to further enhancement of our understanding of the natural checks and balances on lepidoptera populations in the wild. [12] In conversations with Richard S. Peigler, he stressed that parasitoid activity may be one of the primary factors for the well documented peaks and valleys in Saturniid moth populations in the wild. The two populations follow each other with generational lag. As moth populations increase so do their parasitoid populations until the parasitoids cause the moth populations to crash which leads to subsequent declines in the parasitoid populations. Further understanding of the dynamics in these ecosystems will help us to better understand population stressors affecting Saturniids.

5. Acknowledgments

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6. References

1. Ferguson, Douglas C. Bombycoidea Saturniidae (Part), The moths of America north of Mexico, Fascicle 20.2 A. E.W. Classey Limited & R.B.D. Publications, London, 1971-1972.
2. Gardiner, Brian OC. ed. A silkmoth rearer's handbook. Amateur Entomologists' Society, 1982.
3. Lemaire, Claude. Saturniidae of America: Hemileucinae. Antiquariat Goecke & Evers, Keltern, 2002.
4. Meister, Frank. A Guide to the Breeding of Tropical Silk Moths (Lepidoptera: Saturniidae): Die Zucht Von Tropischen Wilden Seidenspinnern (Lepidoptera: Saturniidae). Pfeil, Munich, 2011.
5. Stehr, Frederick W. *Immature insects* [Vol. 1]. Kendall/Hunt Publishing Co., Dubuque, Iowa, 1987.
6. Stone, Stephen E. Foodplants of world Saturniidae. The Lepidopterist's Society, 1991.
7. Triplehorn, Charles A, Norman Johnson F. Borror and DeLong's Introduction to the Study of Insects. Thomson Brooks/Cole. Belmont, California, 2005.
8. Tuskes PM, Tuttle JP, Collins MM. The wild silk moths of North America: a natural history of the Saturniidae of the United States and Canada. Cornell University Press, Ithaca, New York, 1996.
9. Villiard P. Moths and how to rear them. New York, New York: Funk and Wagnalls, 1969.
10. Wagner, David L. Caterpillars of eastern North America: a guide to identification and natural history. Princeton University Press, Princeton, New Jersey, 2005.
11. Wagner, David L, Schweitzer, Dale F, Sullivan J. Bolling and Rearsdon, Richard C. Owlet Caterpillars of Eastern North America. Princeton University Press, Princeton, New Jersey, 2011
12. Peigler, Richard Steven. Catalog of parasitoids of Saturniidae of the world. Lepidoptera Research Foundation, 1996.
13. Steele TJ, Peigler RS. Biology of four species of Hemileuca (Saturniidae) in Texas with special reference to their parasitoids. Lepidoptera Science 2015; 66(1):1-7.