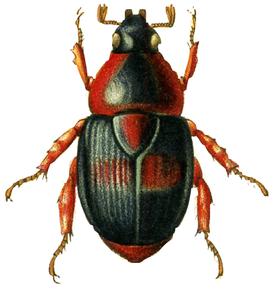

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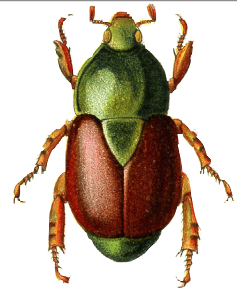

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A Remarkable *Golofa* Hope, 1837 from Peru (Coleoptera: Dynastinae: Dynastini)

by Brett C. Ratcliffe

Systematics Research Collections
University of Nebraska State Museum
Lincoln, NE, USA
bratcliffe1@unl.edu

and Jochen-P. Saltin

Idsteiner Str. 32
D-65527 Niedernhausen/Taunus
GERMANY
jochen.saltin@gmx.de

There are about 28 species in the Neotropical genus *Golofa*, depending on which authority one uses (Endrödi 1977, 1985; Lachaume 1985; Dechambre 1983; Morón 1995). Species are found from central Mexico to northern Argentina and Chile. Thirteen species are found in Central America, and 14 species are found in South America.

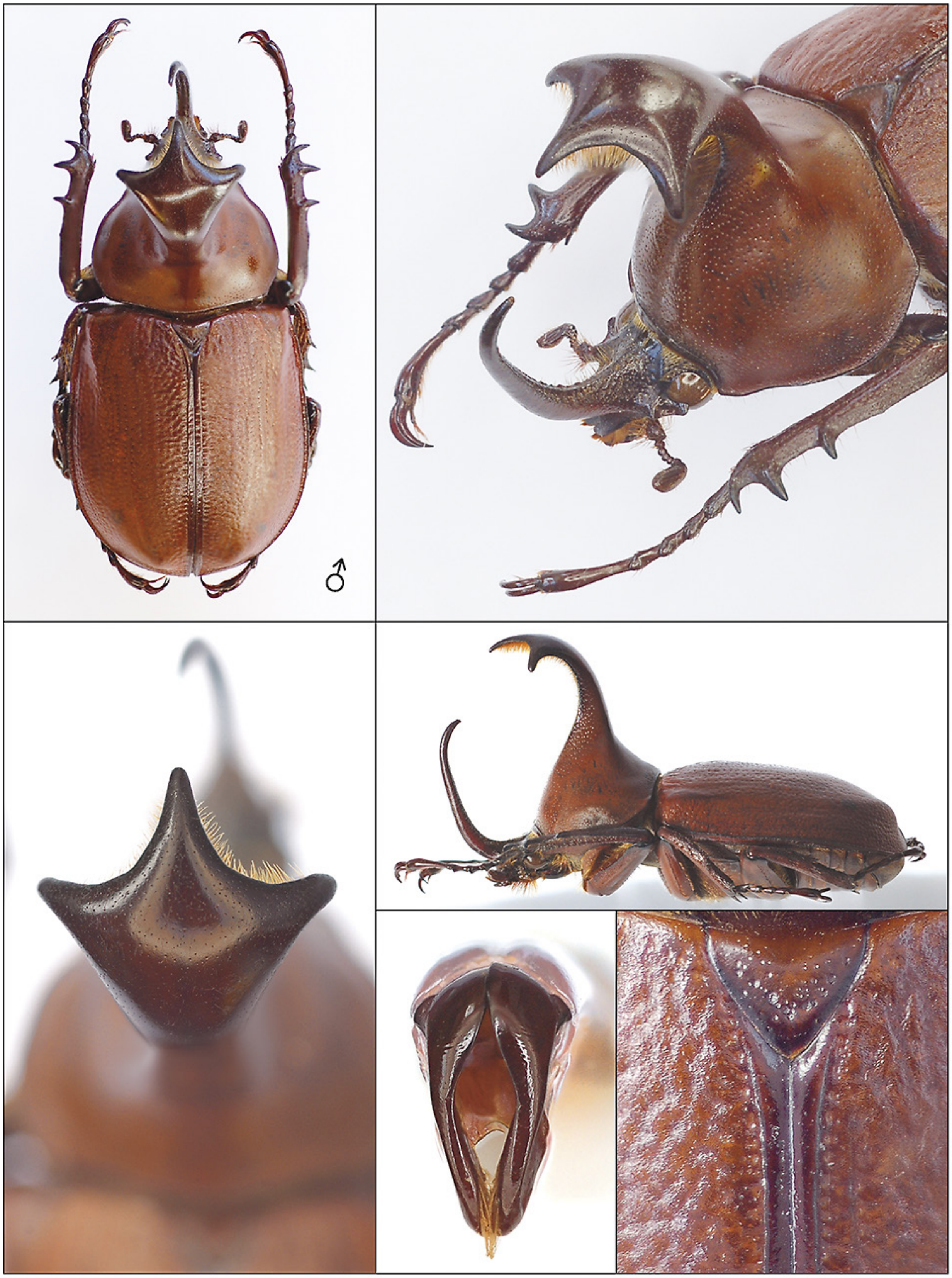
Adult males of most species may be recognized by their brownish yellow to dark reddish brown color (three species are black or nearly so); presence in the males of most species of a short to long, upright, slender head horn and presence of a short to long, erect or obliquely oriented pronotal horn or prominent tubercle. *Golofa* females are dark yellowish brown to more commonly black, and they lack armature.

Even after the modern synopses of those authors cited above, identification of many species of *Golofa* remains a difficult and often exasperating task. Why is this? First and foremost is the significant morphological variation in male secondary sexual characters combined with an unusual (for dynastines) lack of differentiation of the male genitalia. Most authors have based their concepts of *Golofa* species on the characters of male armature, and, since these vary so much within a species due to allometric growth, it has always been difficult to incorporate all of the variation in a workable key, description, or in photographs. In many cases, females can be identified only by being collected with the males. So, male characters vary considerably, the usually diagnostic parameres are not reliable, other characters seem to vary in their expression, and most of the females all seem to look alike. The result is difficulty in identification of some species.

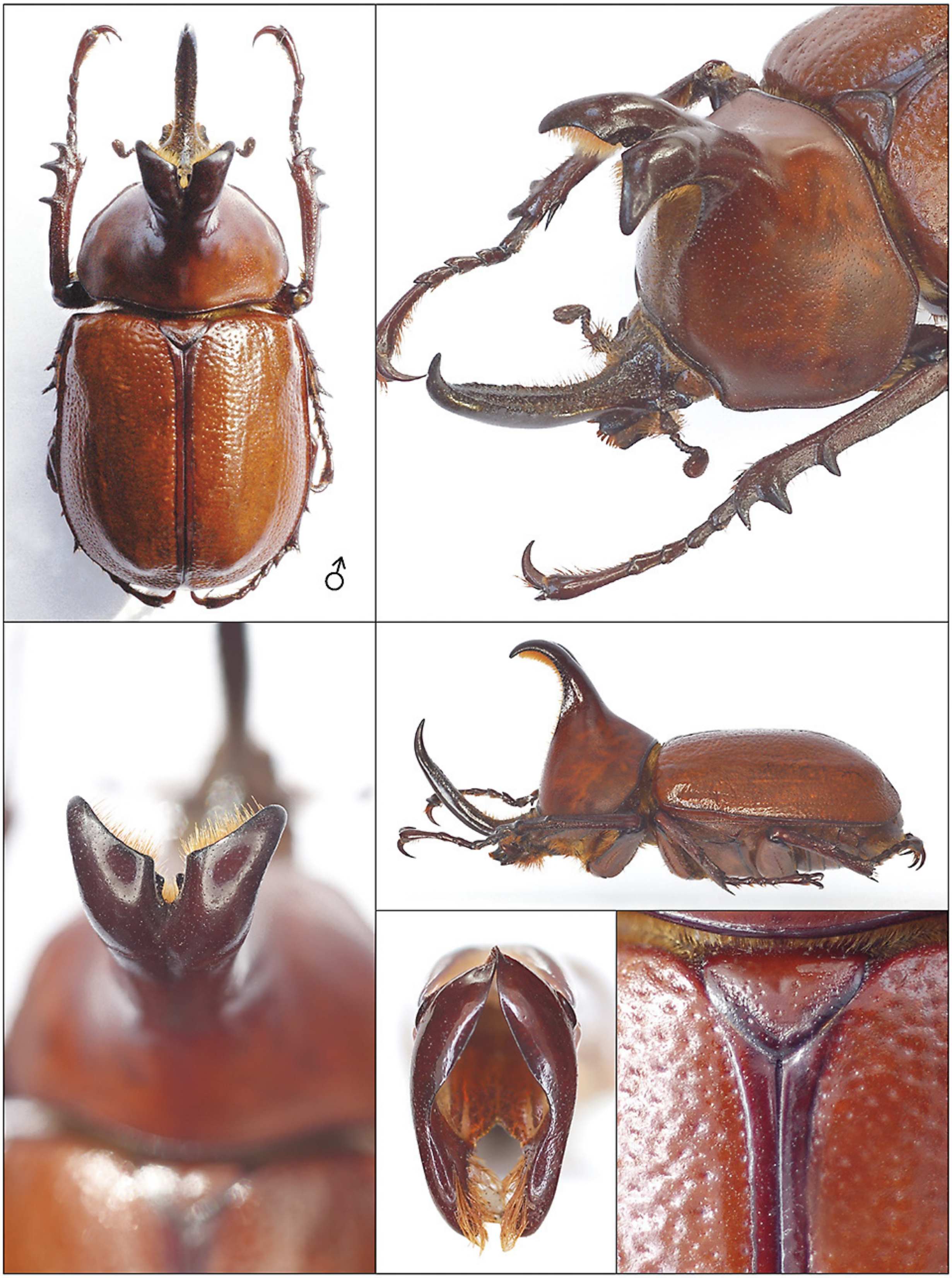
Here we report a presumably unique specimen from the Department of Cuzco, Peru that is unlike any other *Golofa* species known to us. It resembles *G. claviger* (L.) which is known from Peru, Ecuador, and Colombia, but its pronotal armature is truly remarkable, and its parameres differ somewhat. It is either a new species or the result of an irregular combination of genes resulting in a morphological oddity. Additional specimens with the same body structures would confirm a new species, but in the meantime we suspect that it is a unique monstrosity or malformed specimen. Note particularly the pronotal horn where the apex is deeply bifurcate with visible setae on the anterior edge and a thickened head horn. The sympatrically occurring *Golofa claviger*, by contrast, has a pronotal horn with an apex that is strongly and triangularly dilated with setae on the concave surface beneath the dilation and a more slender head horn. The parameres of the monstrosity are also more arcuate rather than subtriangular as in *G. claviger* and are also slightly more robust. These parameres do not seem to match any species of *Golofa*.

So, while it might be tempting to describe this specimen as a new

A map of Peru showing the Departments of Junin and Cuzco, where the range of *Golofa claviger* overlaps with that of the unique specimen describe here.



Golofa claviger **Linnaeus, 1771. Peru, Department of Junin, La Merced, I-2012. Length 46mm.**



***Golofa* sp. Peru, Department of Cuzco, 1,100 meters elevation, II-2012. Length 50 mm.**

species, we believe it serves our science better if we could confirm that there are additional specimens with the same body structure before rushing to judgment.

We thank Michael Bueche (Tingo Maria, Peru; michael_buche@yahoo.es) for providing the specimen for study, and it has been returned to him. This project was supported by a National Science Foundation Biotic Surveys and Inventory grant (DEB 0716899) to Brett Ratcliffe and Ronald Cave.

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Peru Scarabs III - *Golofa eacus* Burmeister 1847

from the Camera of Rob Westerduijn

Iquitos, Peru
rob_westerduijn@yahoo.com

In the beginning of 2009 I made a collecting trip with my Peruvian girlfriend to northern of Peru. This trip included a five-day stay at Abra Patricia, Progresso, on the eastern slopes on the Andes in Amazonas. This area is at 2,600 meters elevation and is mostly covered with cloudforest. It was the rainy season and it rained a lot of the time we were there. We did manage to find a lot of chrysomelids though and also quite a few other big, showy beetles.

Near the place we stayed there was a pasture with cows. Relative to the forest, this was poor habitat of secondary scrub with fewer species of chrysomelids.

On 30 January 2009, along the road through this pasture, I discovered a big rhinoceros beetle which I was happy to photograph. It turned out to be a minor male *Golofa eacus* Burmeister, 1847. (Figure 1). The male was resting on a wooden fencepost along the road above a rotting log. Closer inspection of the log later showed it had five big rhinoceros beetle grubs under and inside it as well (Figures 2 and 3).

Minor male of *Golofa eacus* Burmeister, 1847.

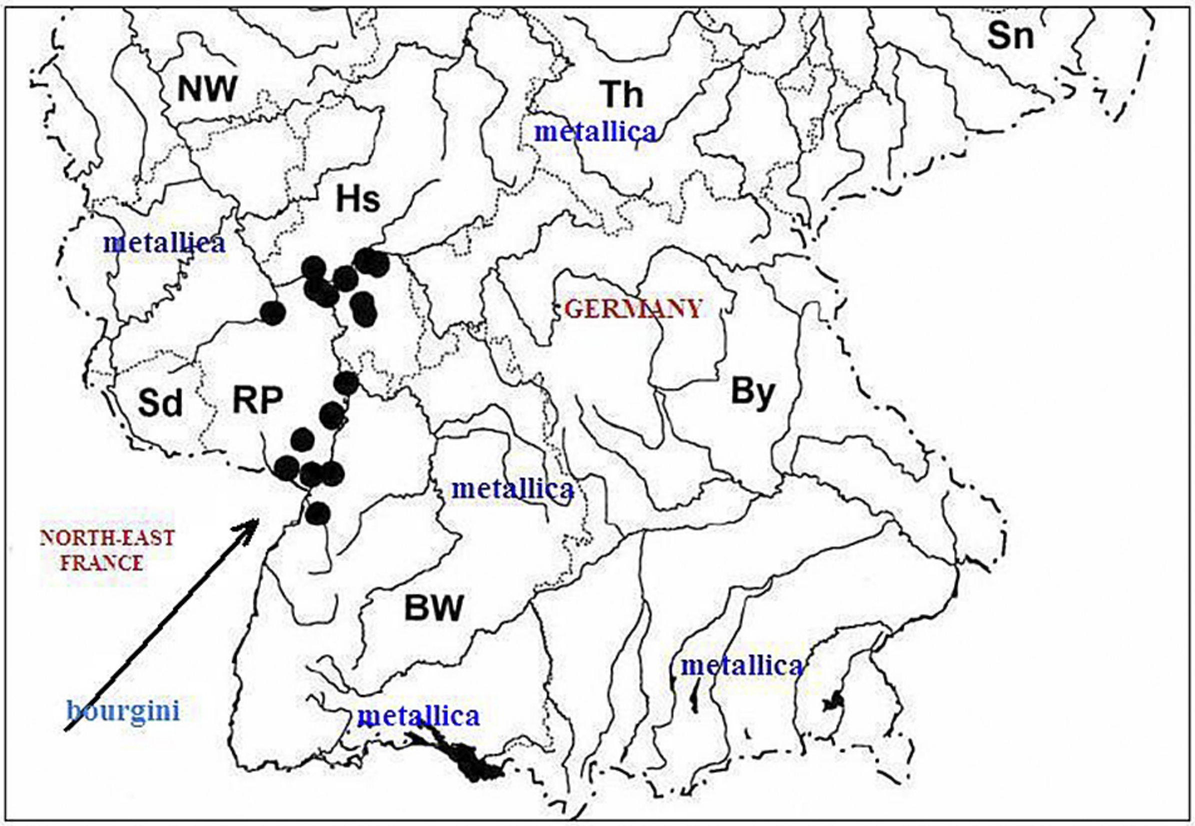
Five large scarab grubs in the decomposing log found beneath the adult male.

A closeup of one of the grubs.

The *Protaetia cuprea* Complex from France to the Orient (Coleoptera: Cetoniidae) – Part II

By Olivier Décobert & Pascal Stéfani

After our first article in March, 2011 (*Scarabs* #61), Eckehard Rössner contacted us from Germany. Thanks to him for providing this map to help to depict the distribution of the subspecies *P. bourgini* (Ruter) outside of France.



Map 1 – Northeast of France and Germany – Black spots show the limited extension of the subspecies *bourgini* in Germany.

Eckehard explained that the subspecies *metallica* (Herbst) is everywhere in Germany, except in a restricted area, close to northeastern France, where *bourgini* is present (black dots on Map 1).

He also informed us that *Protaetia cuprea obscura* (Andersch) does not occur in Germany. He knows this subspecies is from Czech Republic, Slovakia, Austria (lowlands), Hungary, Italy (Lido near Venezia), Bosnia Herzegovina, Croatia, Romania, Bulgaria and Greece.

In Slovakia, *obscura* sometimes meets *metallica* and hybrids are known. This phenomenon also happens in some parts of Romania.

In Spain, *bourgini* is replaced by the very closely-related *brancoi* (Baraud). The two subspecies are separated by the natural frontier of the Pyrenees Mountains. One remembers that our first article also examined the distributions of the subspecies *olivacea* Mulsant (South of France) and *cuprea* Fabricius (Corsica and Italia). On the island of Sicilia the subspecies *incerta* (Costa, 1852) can be found, and sometimes considered as a different species: *P. hypocrita* (Ragusa, 1905).

***Protaetia cuprea brancoi* (Baraud)**

***Protaetia cuprea incerta* (Costa)**

In this second part, we will see how the *Protaetia cuprea* complex is distributed in all parts of Europe, particularly towards the Orient. In the northeast direction, one can observe an extension of the subspecies *metallica* (boreo-alpine lineage) as far as Russia. In northern Europe, *metallica* exists in Norway and Sweden, and at least one specimen was once found (1939) in Great Britain. More recently, this subspecies was confirmed in northern England and in southern Scotland.

Towards the southeast, as far as Turkey and Caucasus, new subspecies are found: *obscura* (Andersch), *cuprina* (Motschulsky), *ignicollis* (Gory & Percheron), *caucasica* (Kolenati), and *hieroglyphica* (Ménétriès), all shown on Map 2. *P. cuprea ikonomovi* (Miksic) is not figured on the map. It is found on the island of Cyprus, south of Turkey.

P. cuprea obscura (Andersch)

P. cuprea cuprina (Motschulsky)

P. cuprea ignicollis (G & P)

Protaetia cuprea caucasica (Kolenati)

P. cuprea hieroglyphica (Ménétriès)

Protaetia cuprea ikonomovi (Miksic)

Protaetia splendidula (Faldermann)

We think that *Protaetia splendidula* (Faldermann) is a valid species because of its geographic extension in southeastern Turkey, widely mixing with *P. cuprea ignicollis*, and also with *cuprina* coming from the west. In this area are beautiful forms of *Protaetia cuprea* called *phoebe*, *edda*, and *ino* (Reitter).

Protaetia cuprea phoebe* (red form and green form)** ***Protaetia cuprea edda

Hybridization and introgression between two close species in this part of the world could explain (at least partially) these particular forms. But without breeding experiments, one must presently consider them to be regional *P. cuprea* forms.

We hope that our two articles will help to understand this complex cetonid, and give the idea to readers having a good knowledge of *Protaetia cuprea* in Asia to help write a Part III. Indeed, this species exists in more eastern countries such as Iran, Pakistan and Nepal and is represented by other subspecies in these areas.

Map 2 – The *Protaetia cuprea* complex in Europe. *Protaetia splendidula* (in blue) is a valid species, close to *Protaetia cuprea* (in red).

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Rearing Scarabs

from the Scarabs Discussion Group

SCARABS-L@listserv.unl.edu

Editors' Note: This interesting thread was started by Auke Hielkema. It contains valuable contributions from several scarabaeologists. We thought it was worth archiving this discussion in Scarabs so that it can be referenced at a later date. It is presented here in its original chronological sequence.

Because the comments were not originally written with the intention of being included in this newsletter, a few passages of text have been edited to improve their readability.

The photos are courtesy of Auke.

Auke Hielkema
auke_hielkema@yahoo.co.uk
Suriname

Dear All,

Here in Suriname I regularly find larvae of scarabs in rotting wood. I often collect them with the intention of raising them and always take as much of the substrate (mulch and crumbling wood) as possible, but this is limited by the available amount and the size of my backback. Since various species appear to be cannibalistic when kept together in a limited space, I now usually put each larva in its own cup with substrate. Often, the amount of substrate appears too little to provide each larva with sufficient substrate of its own, necessitating recycling of the substrate used by larvae that have already pupated. This is somewhat inconvenient and may possibly transfer harmful microorganisms from one larva to another. I'm now thinking of asking a carpenter for a quantity of coarse sawdust to supplement each larva's natural substrate. The question: which wood or tree species (or tree genus) would be best for this purpose? There are various species of wood here which are without oils or toxic chemicals and are not overly hard, but I can imagine scarabs select on more criteria for oviposition. When I find logs with larvae in the forest, it is usually not possible for me to identify the tree species. I prefer a 'one wood feeds all' solution. Any additional info on this topic is also appreciated.

The beetles I raised from wood so far belong to *Macraspis* and *Rutela* (Rutelidae), *Marmarina* (Cetoniidae), *Stenocrates*, *Strategus* and Phileurini (Dynastidae).

Thanks in advance for your help!

Brett Ratcliffe
bratcliffe1@unl.edu
Nebraska, U.S.A.

In my limited experience of doing this in the tropics, you might just try going to a lumber mill where there will be plenty of sawdust that they would probably give you. They might also have some knowledge of WHICH trees they are cutting have the least oils or resins in them. But see below.

It is my understanding that a LOT of the vegetation from the Guyana Shield region of South America is loaded with secondary compounds consisting of humic acids (tannins and phenolics) that are either toxic or repellent to many insects. This is why we often see lower diversity and numbers of insects in black water drainages (tea colored water due to the secondary compounds) versus the white water drainages of, for example, the Amazon, Tapajos, or Madeira. The presence of these compounds may be more of a problem with fresh-cut lumber than with that which has been laying on the forest floor for some amount of time. So, perhaps another strategy might be to simply find a rotting log of suitable moisture content close to where you live or can drive, and then collect bags of the rotting wood... a separate expedition just for wood as it were.

Margarethe Brummermann
mbrummermann@comcast.net
Arizona, U.S.A.

Dry cow dung works as a supplement (heat sterilized!!) and dog food (dry kibbles) fish food (flakes). There will be mold - don't worry about it.

Jason F. Mate
jfmate@hotmail.com
jfmate@gmail.com
United Kingdom

I have been breeding scarabs for years and have found that for rutelines and dynastines a mixture of 60-70% rotten wood sawdust to 30-40% leaf mold works best. For some species the addition of a pellet or two (depending on the size of the larva) of dry dog food will result in faster development but this is a treat. As for volume, you can develop most species in communal tanks if you provide ample room. For *Xyloryctes* I allowed for 1 L per larva with few if any losses. I usually find cetonines to be more aggressive (they require a higher leaf mold content, up to 100% plus some vegetable scraps as a top-up) but species like *Cotinis* can be kept in groups of 6 in 2 liter cans full of medium. Phileurini might be more aggressive, but I have had limited experience with them so I can´t really say how they will behave.

I use large buckets used for cured meat as holding containers. In those buckets, I place smaller containers with substrate and larvae. Just below the rim on the inside of each bucket, I smear some acid-free vaseline to keep small ants from entering. Except for the old substrate sticking to it, it's almost invisible in these photos. Each breeding container has location, date and substrate written on its side.

The 150 ml containers in this picture (probably all with *Macraspis*, one per unit) are apparently just large enough, since several larvae have started pupating. I would like to use somewhat larger containers though, but I don't have sufficient substrate to fill those yet.

These cups of about 500 ml contain what I assume to be larvae of Phileurini. I think these cups are sufficiently large. Two days before making this photo I put a small amount of fresh rotten wood into each of them. The larva in the container on the right-hand side is, although still looking healthy, apparently not too impressed since the fresh wood isn't disturbed.

In regards to wood, fresh sawdust is very indigestible to grubs. I have used white-rot oak or ash for all kinds of larvae so your best bet would be any log in which the wood can be easily torn apart with your hands into fibers (there is a good picture in Wikipedia).

However, if you need lots of it, the appropriate sawdust can be stored in rubbish bins mixed with the leafmold and some rotten wood and left to ripen for a few weeks/months.

Auke Hielkema

Dear All,

Thank you all very much for both your public and private replies!

I often find tree trunks which seem to be perfect for scarab larvae but which contain not a single grub.
I think this might (sometimes) be because of something in the wood which repels insects and I think it would be risky to use this wood as a substrate. Also, I've found there is a risk of taking termites or ants with naturally rotting wood, both of which I don't want to have in my breeding containers. It often takes quite some time to get all of the ants out of the substrate I take with me, but I don't want them to start munching on my larvae.
Because of this, and based on your other comments and the available options, I think I will proceed as follows:

1) Go to a wood trader and get some scrap wood of a light colored, not too hard, non-toxic and oil free kind.

2) Ask the trader to reduce this wood to intermediately coarse sawdust.

3) Submerse the sawdust in a large bucket with tap water and leave it stand for a day or two.

4) Drain the sawdust and repeat this procedure for another one or two times to get rid of most of the tannins, resins and other solubles.

5) Keep the sawdust moist and mix it with a bit of material from a trunk containing larvae to start the decomposing process. Leave it like this for at least about three weeks.

6) Once I find larvae, I mix their original substrate 1:1 with the moldy sawdust. If needed, more sawdust can be added later.

Since I'm not doing this for commercial purposes but solely as part of my inventory of Surinamese scarabs, I'm not interested in speeding up development with extra nutritious foodstuffs or getting beetles to breed. Also, I intend to use the above procedure only for larvae which I find in rotting wood. Many species may be rather flexible with their food, but I think it makes sense to stay close to the substrate in which I find the larvae.

Does anyone of you have comments on this proposed procedure?

Ken Miwa
kmiwa@huskers.unl.edu
Nebraska, U.S.A.

I am sorry this is a very long email, and it is not even complete to cover everything about rearing.

I have reared a lot of scarabs in the U.S. and Japan. After seeing your and other emails, it may still not be a bad idea to use substrate that mostly consists of materials found in the wild if you can sterilize them. Making fresh sawdust decay in the way acceptable to scarab larvae may not always be easy if you have not done so before. As Jason mentioned, scarab larvae do not develop on fresh sawdust.

My largest containers (1,1 liter, see last paragraph of this discussion) contain some larger (finger-sized) larvae, probably of some Dynastidae, one per unit. The lids of these containers have small holes for aeration. In the smaller cups, I just put the lids on very loosely although I may change this in the future.

When I lived in Japan, I just bought commercially available substrate, which often allowed larvae to grow faster and larger than ones found in the wild. Even though you are not trying to accelerate larval development, having the right substrate is still important because it decreases mortality of your insects. In the U.S., I often collect materials from wooded areas to feed my larvae. When I collect rotten wood, compost, and dead leaves in the wild, I usually sterilize them in an autoclave, microwave, or oven. If you do not have access to any of them, you can soak them in water for a few

Close-up of a breeding container showing a larva. I prefer transparent containers so I can keep an eye on the development of the larvae without disturbing them too much.

days to kill unwanted organisms and then place them on a tarp under the sun to dry them out.

For many dynastines and cetoniines, you can use compost-type material found in tree holes (a fallen tree with a hole may be a good place to collect it). Leaf mold works for them as well. You can also add crushed rotten wood to it; rotten wood should be so decayed that you can crush it with your hands. Multiple larvae can be kept in the same container as long as there is enough food and space. Many species of Phileurini can be reared similarly although they usually develop faster. Newly emerged adults of phileurines can kill other individuals that are still pupae and larvae in the same container. You may want to remove new adults from the container as soon as they emerge if multiple individuals are kept together.

Many rutelines do well in crushed rotten wood or pieces of rotten wood. Jameson 1997 (Phylogenetic analysis of the subtribe Rutelina and revision of the *Rutela* generic groups) mentions that *Rutela* species have been reported from rotten wood of *Artocarpus* sp., *Bursera* sp., *Conocarpos* sp., *Ficus* sp., *Inga* sp., *Mangifera* sp., *Metopium* sp., *Simarouba* sp., and *Tabebuia* sp. *Macraspis* species may have similar hosts. For *Rutela* and *Macraspis*, rotten wood can be crushed, and compressing the substrate sometimes helps. Also, you could simply place larvae in pieces of wood (e.g. 10cm x 10cm x 10cm of wood for a larva) instead of placing them in crushed flakes of wood (you can soak wood in water if it is too dry). Since a number of *Macraspis* adults, pupae, and larvae can be found in a single tree stump or log, overcrowding may not be a serious issue for the genus although it might be the case for *Rutela*.

In addition to using rotten wood, dead leaves and compost-type material found in the wild, I make a homemade diet similar to the kinds sold at beetle specialty shops and supermarkets in Japan. If you can obtain a large amount of coarse sawdust from a wood trader, you can use it to make a diet. I want to emphasize again that fresh sawdust cannot be used as a larval diet. In addition, you want to avoid sawdust from trees that contain substances potentially harmful to your larvae. In the U.S., I usually use fresh sawdust of oak and mix it with wheat flour and water. I then place it in a large container in a warm area to let it ferment. The fermentation process is usually complete in a month or two if the temperature is constantly over 25 degrees Celsius. This is a great way to make a larval diet for scarabs (and lucanids) if you are trying to make them grow faster and larger. However, it may not be as easy as collecting and mixing materials from the wild. Some people struggle to successfully make the diet when they try it for the first time. As you mentioned in your last email, you can mix fresh sawdust with materials from the wild and will probably have a smaller chance of complete failure. However, I think it will take longer than three weeks for the sawdust to decay unless only the smaller portion of the mix is fresh sawdust.

Moreover, in my opinion, the species of trees used to make a diet are not too critical as long as completely unpalatable tree species are avoided. Hundreds of scarab and stag species from all over the world can be purchased legally in Japan, and they can be easily reared using commercially available substrate, which is made mainly from two Japanese oak species. Therefore, the stage of decay of (or the species of fungi infesting) the substrate may be more important than the treespecies used, at least for many dynastines and cetoniines.

I am sorry for writing so much here. In my opinion, it may be easier for you to use rotten wood, leaf mold, and compost found in the wild, and to try to sterilize them.

Here in Suriname, it can become really warm in my room when it's closed for a day. Because of this, I put the buckets outside on the roofed terrace. Still under a zinc roof, but out of the sun and at the place where they catch most wind, so the heat from the roof hopefully has not too much effect.

When I find materials in the wild for my larvae, a lot of suitable ones do not have any insects I am trying to collect or rear. If you can identify the right kind of rotten wood, I do not think you need to worry about not finding any insects in it. I think we just find a lot of appropriate rotten wood that has not been occupied by scarabs yet.

Angel Solís
asolis@inbio.ac.cr
Costa Rica

In some cases sawdust from carpenter machines or chainsaws is mixed with oil residuals that cannot be good for larvae.

Brett Ratcliffe

This certainly seems worth trying. You might also consider adding to the final product some crumbled up, dry cow or horse manure.

Auke Hielkema

Thank you all again for the new replies! Below, I'll discuss the points you've mentioned.

Ken, your reply was definitely not too long! Very useful info indeed, especially Jameson's list of wood genera. *Mangifera* (I may have found something in that one, I'm not sure it was a *Mangifera* or something else) and *Ficus* have very sticky juice when fresh. Apparently that's no objection to scarabs. I think *Simarouba* has quite bitter wood (at least some species), so that may also not be too much of an objection. I once found a passalid under the bark of a dead *Hura crepitans* which is called posentri here (derived from poison tree), so apparently many nasty chemicals decay pretty quick once a tree is dead. I suppose the main prohibitors will than be the hardness of the wood and possibly the oil content. There is a very common species of *Inga* here in the coastal area (*Inga ingoides*) which I think is not commercially used (lousy wood quality I suppose); its leaves and fruits are eaten by several vertebrate species, so it probably has soft and non-poisonous wood. I'll try to find a good log of it to make some substrate (yes, that will take me at least a month). I now also remember a trunk in the middle of a small pasture which contained both rutelines and cetoniines. I think some large chunks of slightly decayed wood may still be there. I'll try to get back to that place too to get some material.

I'll see to it that any sawing machine is not leaking oil on the wood (they don't use the biodegradable kinds here I think).

Regarding the addition of dried horse or cow dung to the substrate: the substrate has to be moist, and so will thus become the dung. Moist dung has a stronger odour than moist wood. Given the fact that I'm renting a room and my landlord's office is right next door, I think this specific supplement would not be the best idea in my situation...

On the topic of multiple larvae in a single container, I've noticed that scarab larvae are sometimes found in a very narrow space in a tree trunk, surrounded by decaying but not yet pulverised wood (white rot I suppose) and an amount of pulp of their own production. These larvae seems to be quite immobile in their small self-made living space. Once I put such larvae in a container containing wood pulp, they become quite mobile and keep tunneling through the substrate. I don’t think this is because of bad conditions, because they still pupate and become nice imagos. I think this tunneling behaviour might trigger cannibalism though, since such active larvae have a much greater chance of finding their siblings when kept together in a container. I’ve noticed on several occasions that there were only three larvae in a container which should contain four (although I’m not sure anymore if those larvae also were found in narrow living spaces). I’m positive that larvae of Phileurini are cannibalistic, but then again, what is sufficient space? I now put only very small larvae together and put the largest ones all separate or (cetoniines) in pairs.

I don’t think I’ll try to get wood pulp from tree hollows I happen to come across. The forests here are home to several poisonous snakes, scorpions, the giant centipede *Scolopendra gigantea*, bullet ants and multiple small rodents and opossums with sharp teeth, so putting my hand in a dark hollow to grab a handful of woodpulp seems somewhat too risky. When opening tree trunks or lifting dead bark I always use a large dagger because of this, and I only collect wood which I’ve disturbed sufficiently to get all larger species to become active and move away.

So far, I’ve only once ‘sterilized’ substrate (fresh termite nest for some Phileurini). When collecting, I make sure the woodpulp is free of bigger animals like spiders and scorpions. My main concerns though are termites (dry-wood species may attack the house) and ants (which may attack the larvae). The presence of large quantities of these two are the main reason I usually have to leave part of the substrate behind. Are there other small organisms which may attack the larvae or render the substrate useless? I know about mites, but I’ve never seen those in large quantities.

Latest news: I found (and took) larvae of two species and a decent quantity of substrate for them (I think). Tomorrow I’ll try to get some containers for them and check if they all survived the bumpy voyage. Is there any knowledge on the (dis)advantages of square vs round containers and the diameter/height ratio? So far, I’ve used round containers with a more or less 1/1 ratio which I place in large buckets with lids to improve protection, microclimate and handling.

A dish of the perfect wood for rearing scarabs.

Auke Hielkema - Follow-up

I thought you deserved a follow-up because of your efforts to answer my questions, so here it is.

Of the 13 pretty large larvae I found recently (see last lines of my previous letter) 12 seem to do fine and one died after getting back home. I had no replies on my question regarding container shapes, but since the only available containers of sufficient size were rectangular that problem solved itself. The four smaller larvae I found in a different trunk are all pupating now.

A couple of days ago I was able to return to the old trunk with rutelines and cetoniines I also mentioned in my last letter. The field it lay in was burned and the lying trunk badly scorched. However, I could now reach a part of the trunk that was covered with weed before. To my surprise and delight, almost all of the inside of that part consisted of slightly moist, spongy, crumbly, termite free and light coloured wood!

Needless to say I took quite a bunch of this perfect substrate. I've now crumbled up part of it and placed some of the crumbles in containers with larvae. Although I found almost no animals inside it except for very small millipedes, I'm now soaking the rest of the crumbles in a bucket with water to get rid of any remaining beasties. I think I keep part of it moist afterwards to get it further decomposed and may thoroughly dry another part for long-time storage.

Materials and Methods for Collecting and Preserving Aphodiines

by Giovanni Dellacasa

Via Talamone 31/19
16127 – Genova ITALY
dellacasag@alice.it

1. Materials and collecting methods

Such methods in aphodiines noticeably evolved mainly due to the fact that today's systematics require remarkable sets of specimens. The commonest methods are dung, earth and sand washing, traps both traditional or automatic and catches at light.

Dung washing requires the following tools:
- an obconical plastic pail of average dimension (Fig. 6);
- a diaphragm of metallic grid with meshes of about one centimetre; the diaphragm has to have a diameter corresponding to the one taken at about the middle of the pail;
- a hook extractor;
- a tea strainer of small diameter (metallic net) (Fig. 4);
- a plastic can of about twenty litres capacity (water) which will suffice for three washings (Fig. 3);

Three generations of Dellacasa scarabaeologists: son Marco is on the left, and Giovanni is holding grandson Giovanni Gabriele. This photo was taken at Giovanni's home around Christmas in 2010.

- some large jars with a quite large mouth with a screw cap such to ensure high closure (Fig. 6);
- several rather robust nylon bags.

In the field a bag of dung, at the correct stage of maturation, is taken from the pasture. The content of the bag is thrown into the pail. The diaphragm is forced into the container. To avoid opacisation, some water, not cold, is poured in the pail until water level reaches about five centimetres above the diaphragm. After about from five to ten minutes the aphodiines, and obviously any other coprophagous beetles, will be floating on surface of the water and may be collected with the strainer. The content of the strainer is emptied into the large-mouth jars into which, at the end of the job, some ethyl acetate is poured as a killing agent.

The materials so collected are selected when back in the lab. When catches are distributed during a trip of several days, after a few hours after the addition of ethyl acetate the said jars are filled with water and filtered in the strainer whose content is transferred directly into small nylon bags whose opening must be such as to allow the introduction of the strainer, of course adding to each bag a label with all collecting data written by means of indelible ink or pencil. The bags must be sealed with a stapler.

The following day all bags must be put into a container with 70° alcohol so that they remain sunk into the liquid. Some very small holes should be pierced on the bags to facilitate their sinking. This method of preservation is long lasting so that selection of the material can be carried out at the end of the trip. This method is quite efficient for sheep pellets; it does not seem to be fit for cow or horse dung.

Earth and **sand washing** is quite useful not only for catching those species which are sabulicolous (mainly psammodiines and rhyssemiines) even when the ground below the dung patches has to be investigated. After having screened with the sieve (Fig. 10) the coarsest detritus the remaining material screened in small meshes sieve (Fig. 7). This operation shall be carried out near a stream or a water pool; the sieve is merged in the water eliminating sand and minute detritus. The sieve is left in the water so that only its edges are emerging; the floating detritus is carefully examined and with the strainer the specimens remaining afloat are collected; specimens are then put into the normal collecting jars.

It is also possible to gather the whole mass of detritus for its later examination back in the lab, throwing it in a white plastic saucer (Fig. 8) filled with the water. Obviously not much time can elapse between the collection of the detritus and its examination to avoid the rotting of the whole material.

Another important niche which must not be disregarded are the nests and burrows of rodents (i. e.: *Citellus*, *Marmota*, etc.) due to the peculiar species association living in such microhabitats (i. e.: *Chilothorax*, *Heptaulacus*, *Osmanius*, *Paracoptochirus*, *Plagiogonus*). The earth at the entrance of the main tunnel must be examined.

The most useful tools are:
- bulb extractor (Fig. 12);
- a robust kitchen spoon modified as in Fig. 14;
- a fine meshed sieve (Fig. 7).

With the bulb extractor the earth at the mouth and inside the main tunnel is removed and collected by the spoon. Sieving the whole, foleophilous insects can be collected.

Traps are quite often giving surprising results for the collection of aphodines. The two following kinds of traps are especially efficient when is utilized cow or horse dung.

Automatic traps require a plastic container with a very tight closure (Fig. 23) and another container made out with metallic grid with meshes of about one centimetre (Fig. 22). Such container has to be such as to allow its introduction into the plastic container and must be provided with some short supports to keep it two or three centimetres higher with respect to the bottom. The dung is put into the metallic net container after having filled the bottom of the plastic container with about one centimetre thick layer of sand or earth. Tight closure of the plastic container brings the dung to rapid fermentation and the gases developed therefore compel the insects to run out of the dung and to find refuge in the sand on the bottom of the container. This device can be operated for about 24 hours in normal temperature conditions (15-20°C).

The literature dealing with "traditional" traps, (pitfall traps, etc.) is extremely conspicuous. Nevertheless the types with a suspended basket or with a metallic grid placed at the level of the ground have been succesfully tested for collecting aphodiines, and result in specimens particularly fit for ecological and faunistic studies of a given niche due to its objective characteristic needed for a statistical survey. Both types need a cover protection prevent the bait from being altered by rain.

The following materials are needed:
- a small plastic pail (Fig. 21);

- a funnel cut at about three centimetres above the connection of its tube (Fig. 20);
- a strip of window antifreeze gasket;
- a basket (Fig. 17) or a sheet of metallic grid (Fig. 15);

For preparing the cover:
- two iron wires bent as per Fig. 16;
- a square sheet of plastic material of appropriate dimensions (Fig. 19).

The small plastic pail is filled with a mixture of 40° ethanol, odourless liquid soap, acetylsalicylic acid (acting as preservant) and, in winter, pure glycerin acting as antifreeze. The bait put directly on the grid or in the suspended basket can be of any kind of dung; the most productive is cow and horse dung.

Catches at light using the traps, well known to lepidopterologists, are particular efficient in tropical environments though not all species of aphodiines are attracted to light.

2. Collection

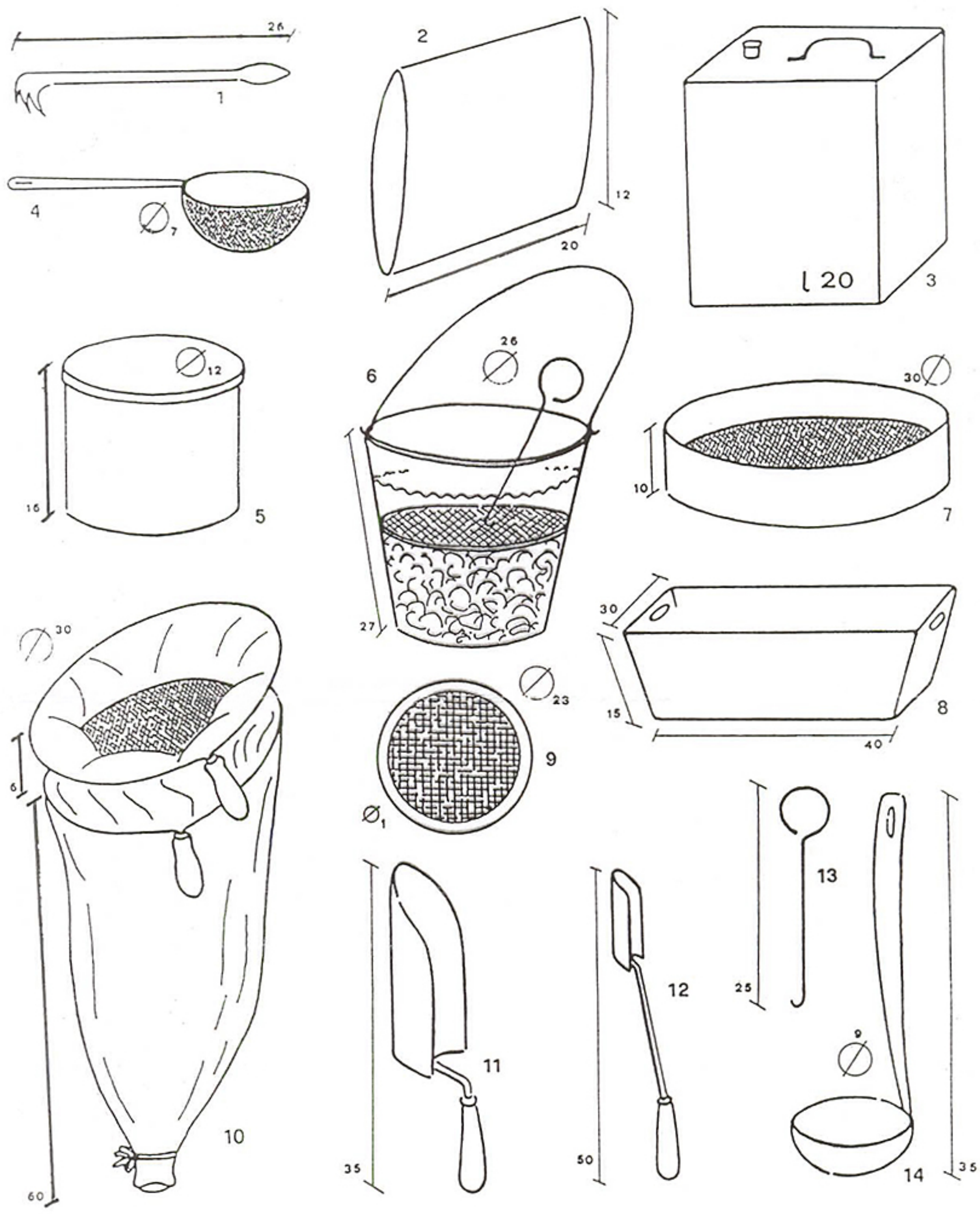
Today's systematics needs had an impact on the collecting techniques, bringing about radical changes both to said techniques as well to the preservation criteria of the collection.

In the old collections, species were often represented by voucher specimens. Today's research requires a large set of specimens of the various populations existing in the distribution area of a given taxon and such as to give a representative picture of its phaenology. At least as far as aphodines, the number of specimens sufficient as a base for a careful study of the population is about hundred for each locality and date of collection.

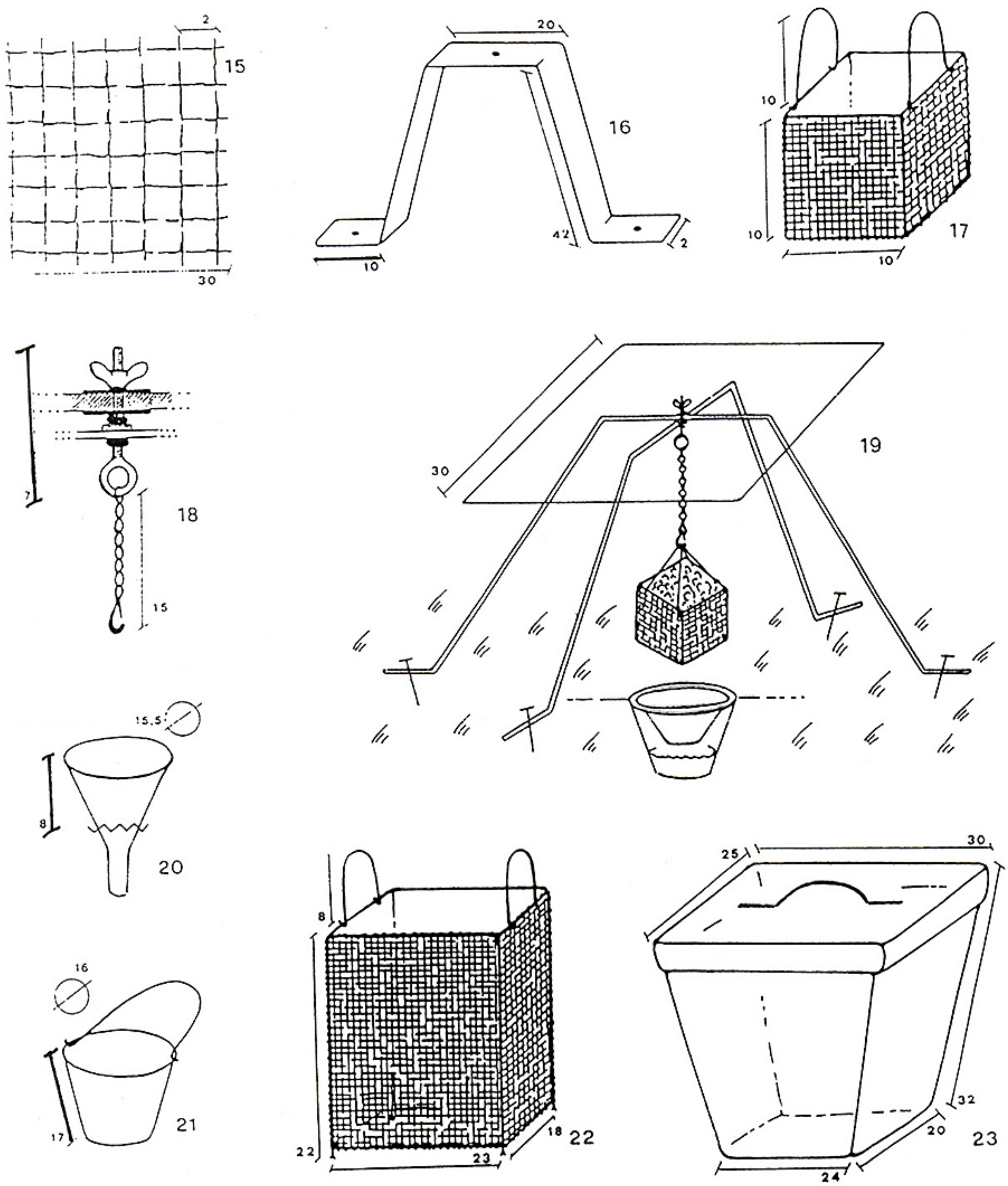
The problem of arranging material in the collection, meeting both the taxonomical needs and the today's wider systematic needs, can be solved by dividing the collection in two parts:

- **voucher collection**, whose taxa shall be represented by a few individuals so that a panoramic systematic view of the various groups be available;

- **population collection**, with numerous series of specimens by species and, within the species, keeping separate by more and more selective geographical criteria, depending upon the amount of the material available and, if possible, with further chronological subdivisions. The whole will allow the management of the material in view of further studies beyond the systematic level, i.e. ecological, geonemical and geographical distribution level.



Figs. 1-14: 1. – gardening hoe; 2. – small nylon bag; 3. – plastic can; 4. – strainer; 5. – large mouth jar; 6. – obconical plastic pail; 7. – small meshes sieve; 8. – plastic saucer; 9. – metallic grid diaphragm; 10. – sieve; 11. – gardening shovel; 12. – bulb extractor; 13. – hook extractor; 14. – robust kitchen spoon modified. (Dimensions in centimetres).



Figs. 15-23: 15. – metallic grid; 16. – iron wire; 17. – metallic grid basket; 18. – supporting-screw detail; 19. – mounted trap with basket; 20. – funnel; 21. – small plastic pail; 22. – metallic grid container for automatic trap; 23. – plastic container for automatic trap. (Dimensions in centimetres).

A New (and Old) Publication on *Phanaeus*

by Dave Edmonds

wdedmonds@sbcglobal.net

Jiri Zidek and I are pleased to announce the publication of “Taxonomy of *Phanaeus* revisited: Revised keys to and comments on species of the New World dung beetle genus *Phanaeus* MacLeay, 1819 (Coleoptera: Scarabaeidae: Scarabaeinae: Phanaeini).” (*Insecta Mundi* 0274: 1-108).

A high resolution pdf of the paper (595 MB - download time 15 minutes to several hours, depending on connection speed) is available now for download and printing at the following URL:

http://centerforsystematicentomology.org/insectamundi/0274EdmondsandZidek-full.pdf

A low resolution (53 MB) “reading copy” is available for download at the following URL:

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The reduced version suffers from anomalies in some the plates (spurious lines) resulting from the reduction of the original file. It is, therefore, not an ideal candidate for printing at this time. Work to rectify these anomalies is ongoing.

We have arranged for a one-time, limited professional printing of multiple paper copies (separata; reprints) for our colleagues who prefer not to print a shelf copy on their own. Price per copy: US $28 (Soft cover; 108 pp, 21.5 x 27.5cm; 83 plates, 410 figures in full color) Availability is very limited.

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This publication includes a supplemental reprint of W. D. Edmonds (1994). Revision of *Phanaeus*, a New World genus of scarabaeine dung beetles (Coleoptera: Scarabaeidae: Scarabaeinae), Natural History Museum of Los Angeles County, *Contributions in Science* 443: 1-105. The 1994 paper, which is an important but long out-of-print complement to the new taxonomic treatment, is available for download at the following URLs:

Large version: http://centerforsystematicentomology.org/supplemental/Edmonds-1994-large.pdf (66.5 mb).

Reduced version: http://centerforsystematicentomology.org/supplemental/Edmonds-1994-small.pdf (15.1 mb).

***Scarabs* proofreader Jennifer displaying an original of the much sought after 1994 revision - long out of print.**