The Very Handy Manual: How to Catch and Identify Bees and Manage a Collection

A Collective and Ongoing Effort by Those Who Love to Study Bees in North America

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This manual is a compilation of the wisdom and experience of many individuals, some of who are directly acknowledged here and others not. We thank all of you. The bulk of the text was compiled by Sam Droege at the USGS Native Bee Inventory and Monitoring Lab over several years from 2004-2008. We regularly update the manual with new information, so, if you have a new technique, some additional ideas for sections, corrections or additions we would like to hear from you. Please email those to Sam Droege (sdroege@usgs.gov). You can also email Sam if you are interested in joining the group's discussion group on bee monitoring and identification.

> *"They've got this steamroller going, and they won't stop until there's nobody fishing. What are they going to do then, save some bees?"* - Mike Russo (Massachusetts fisherman who has fished cod for 18 years, on environmentalists) Provided by Matthew Shepherd

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Nets - Almost any sort of insect net will catch bees. However, bee collectors do have preferences. Most people now use aluminum handled nets rather than wood. Some prefer the flexible strap metal netting hoops as these work well when slapping nets against the ground to capture low flying or ground resting bees. Others prefer the more traditional solid wire hoops. Hoop size varies from about 12" to 18." The larger the hoop the larger the area of capture, but the more difficult it is to swing quickly due to air resistance and the more netting there is to snag on branches.

BioQuip makes a net that has a pole that disconnects into 3 small sections with a hoop that can be folded into itself to form a very useful net/pole combination for travel or backpacking, additional sections can be added to reach into out of the way places. Telescoping poles are also available but must be treated with care or they locking mechanisms will jam. An inexpensive long pole can be rigged by attaching a net hoop to a section of bamboo with hose clamps. Aerial nets rather than beating or sweep nets are normally used around the hoops. A fine mesh net bag rather the traditional aerial net bag will keep the smallest Perdita from escaping.

Netting Technique - Netting strategies vary with the target species, habitat, and the preference of the collector. Most bees fly very quickly and when in the air can readily detect and avoid a slow moving net. Consequently, to net bees in flight you must swing the net rapidly and without hesitation. If you

hesitate for even a fraction of a second the opportunity will may be gone. Thus if there is any possibility that what you see is a bee it is best to net it to confirm your suspicion and release it if it is not a bee. For some quick moving Megachilids and Eucerines you will often find them so fast that it is often best to simply wait in a section with few flowers and swing through the flowers when any motion in the vicinity is detected hoping that you intersect with the bee.

When netting off of flowers, ideally it is best to swing through the flower with the bee centered in the middle of the net. Again, a quick swing is demanded here and although this will often result in the beheading of the flower, bees readily detect the presence of a slow moving net and will flee. If there are prickly plants in the area then care must be taken before swinging to make sure there are no pricker bushes, beggar's ticks, or hitchhikers nearby or your net may tear or become full of difficult to remove seed heads as you swing. When in the field usually one hand will hold the tip of the net bag against the pole and clear of the ground and vegetation until you make your swing.

If a bee is flying low to the ground, resting on the ground or on a flower that lies close to the ground then the best approach to capture is to slap the net quickly and firmly over the bee. Once done you must lift the net bag vertically to encourage the bee to follow its natural tendency to fly or crawl upward rather than try to escape under the rim. In some cases you must wait a few moments for the bee to start crawling up the net. Once the bee is half the way up or higher you can pull the net out and back and snap the bee to the bottom of the net for retrieval.

The most efficient way to collect bees is to not remove them immediately after you catch one, but, rather to fill your net with numerous specimens and remove them after you have accumulated a number of specimens. You can keep bees in your net while collecting by either holding the net closed above the end of the net where the specimens are accumulating or by turning the net sideways or flipping the net bag over the rim so the bees have a difficult time getting out. It pays to periodically shake the bees back down to the end of the net bag while collecting as they can eventually work their way out of the bag if you wait too long. Use of a soapy water or alcohol vial to remove bees from a full net is very efficient as once they go in they don't go back out and you can better control the situation of removing so many bees at once.

A video about how to net collect bees is viewable at: http://www.youtube.com/watch?v=n6ZFlz3uA7E

Removing Bees From the Net - Once you have a bee or bees in the net there are several ways to remove them. In all cases it is best to vigorously snap the net to drive all the insects to the bottom. You can then safely grab the bag just above where the insects are resting. Even the larger and more aggressive bees can't get at the hand that is closing off the net due to the bunching of the netting. If you are timid, are worried about the specimen escaping, or have numerous insects in the net, you can kill or at least pacify your catch by stuffing the specimens and the netting into your killing jar and loosely closing the lid over the specimens and the netting. In these cases it pays to have your jars well charged with cyanide or ethyl acetate so that the specimens quickly quiet down or you will waste a lot of time waiting. Once your specimens are quiet you can open up the net and drop them directly into the kill jar without worrying if they will fly away.

Most take a more direct approach and bring the open kill jar into the net and trap the bee against the netting. Slapping the hand on top of the bottle or test tube through the netting is at times useful to drive the bee to the bottom so the bottle can be safely capped without the bee escaping. More than one bee at a time can be put into a bottle this way, but at some point more escape than are captured. Because seeing the bees through the netting can be difficult (hint: use your body to shade the netting to better see the bees) some have taken to hanging the net on the top of their head using one hand to hold the net out and up and then using the other hand to reach in and collect the specimen with the kill jar. It is important in this situation to keep holding the net out so the bees move away from your head and to use small collecting jars or large test tubes that can be handled with one hand. Despite having your hand (and sometimes your head) in the net with the bees most collectors are rarely stung.

In general, bare hands are recommended when removing bees from nets. Bees and wasps will almost never sting you in a net if you don't trap them in your hands or against the net. Use of a centrifuge tube filled with soapy water makes removal easy as you can keep well away from the bees.

Some people will use gloves, but the larger wasps and social bees can drill through some of these. People have used handball gloves, welder gloves, latex gloves used to protect your hands when washing dishes (they can't get a purchase on these glove, but can sting through them), and goatskin beekeeper gloves.

A video that demonstrates how to remove bees from a net is viewable at:
http://www.youtube.com/watch?v=n6ZFlz3uA7E

General How-to Videos on how to work with insect collections are available at:
http://www.bugs.nau.edu/learning_modules.html

Using Ice and Dry Ice - If it is important to keep bees alive or very fresh, then you can bring along a cooler of ice or dry ice. You can then continuously net bees with one net and once the net is full you can place the entire net end into the cooler. If the cooler is filled with ice the bees will remain alive but inactive, if the cooler is filled with dry ice they will freeze. You then continue collecting with a second net and once that one is full the bees in the first net have been chilled or have perished and you can transfer them to jars in the cooler for further storage.

Baggie Catcher - A second useful capture system, particularly when working with individual specimens, is to use large baggies and pop the open end over flowers with bees on them. The bees can then be sealed in the bag and placed in a cooler of dry or regular ice to preserve them until taken back to the lab.

Bee Vacuum – The first section on converting a Leaf Blower was contributed by Julianna K. Tuell another technique for converting a portable "dustbuster" vacuum is available in pdf format at our Listserv web site (http://tech.groups.yahoo.com/group/beemonitoring/files/) and was written by Glenn Hall.

Sam Droege asked me to send out a detailed description of the modified leafblower that was used in Michigan to collect flower visitors, because it may be of interest to members of this list. Every method used to collect insects has certain biases, but we found that vacuum sampling ended up collecting similar numbers of both large and small bees to those recorded during timed observations at the same flowering plots by trained individuals. One obvious advantage of vacuum sampling is that it can be conducted by someone with very little training.

A Stihl leafblower and vacuum converter kit were purchased from a certified Stihl dealer. My colleague, Anna Fiedler, who purchased the components and conducted most of the sampling said it was very easy to assemble and use. She added two screws a couple inches from the end of the intake tube (not sure if this was part of the kit or if this was something extra she did on her own) so that she could use rubber bands to hold a handmade mesh bag (made of no-see-um mesh) over the end for collecting the insects. She vacuumed each 1m^2 plot's flowers for 30 seconds and then while the leafblower was still on, she would quickly remove the mesh bag, close it and then place it in a cooler to immobilize the insects in the bag so that they could be transferred to a ziplock bag without losing any individuals. In this way she could reuse the mesh bag for another sample on the same day and she only needed to carry 4 mesh bags.

Here is the link to the actual model leafblower that was used:
http://www.stihlusa.com/blowers/BG55.html

You can find out more details on the natural enemies part of the project via these two references:

Fiedler, A. and Landis, D.A. 2007. Attractiveness of Michigan native plants to arthropod natural enemies and herbivores. Environmental Entomology 36: 751-765.

Fiedler, A. and Landis, D.A. 2007. Plant characteristics associated with natural enemy abundance at Michigan native flowering plants. Environmental Entomology 36: 878-886.

The manuscript for the bee part of the project has been published:

Tuell, J.T. and Isaacs, R. (2008) Visitation by wild and managed bees (Hymenoptera: Apoidea) to eastern U.S. native plants for use in conservation programs. Environmental Entomology 37: 707-718.

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Smaller, Pocket Sized Bee Vacuum - This section was contributed by Cheryl Fimbel based on the initial suggestion by Priya Shihani.

I am writing to pass along a tip that was provided to me by Priya Shihani. She recommended a small hand-held vacuum unit – the Dirt Devil Detailer (model CV 2000) for vacuuming up small bees and other insects off of flowers. This small vacuum does a fantastic job of scooping up all sizes of bee from the smallest to big'uns (Bombus, but perhaps not the queens). It is especially useful for the tiny bees that would get lost in a net, or are foraging among flower parts that preclude capture with a net. It is ready to use right off the shelf, as it has a small flap that comes down to prevent escape by insects when the vacuum motor turns off. It is small enough to fit in a pocket, and one charge of the battery lasts for weeks. I like carrying two of them in a 'holster' I devised, like a pair of six-guns…. ever at the ready to scoop up a flower visitor, with each vacuum dedicated to a specific flower species. It is the most fun I have ever had vacuuming (I just hope my house guests don't notice that my flowers are cleaner than my carpet!).

Bees Through Binoculars - For those investigators who do observations of bees on flowers or around nest sites the Pentax Papilio 8.5X21 binoculars are ideal. They have high magnification and focus down to 0.5m permitting sight identifications and detailed behavioral observations (once you have learned specimens identification under the microscope).

Kill Jars - Several companies make kill jars. Traditional jars are made of glass with a layer of plaster of paris at the bottom. At the start of the collecting day, enough ethyl acetate is added to the plaster so that it soaks in but leaves no liquid on top. If you are using the jar regularly then the ethyl acetate will need to be recharged every couple of hours as it evaporates. Ethyl acetate has the advantages of not being as toxic as the most common alternative chemical (potassium cyanide), not a controlled substance, and relaxes the specimen a bit more than cyanide (useful if the genitalia are being pulled). It has the disadvantage of needing to be recharged often thus requiring that the ethyl acetate be brought into the field or that several charged kill jars remain available. Additionally ethyl acetate adds moisture to the killing jar that can end up matting a specimen's hairs and significantly degrades DNA unlike cyanide and other killing agents.

Most collectors eventually end up using a cyanide based kill jar. BioQuip makes kill jars with a hollow plaster top underneath the lid that can be charged with potassium cyanide crystals. However,

cyanide jars can be made from any glass or plastic container. A layer of cyanide crystals are placed in the bottom of the container and then a layer of sawdust and then wet plaster of paris is poured over that. The jars are left open for a few hours outside or in a hood and then closed. Alternatively, a combination of cotton balls and tightly rolled paper towels can be used in place of the plaster and sawdust. Such jars can remain effective for over a year.

Cyanide jars usually work immediately in the field, but if they don't knock down specimens right away a drop of water or a bit of spit (don't lick!) will cause the crystals to begin giving off gas. Many collectors use test tubes or narrow vials with a cork top as collecting vials. These are useful when there is a need to keep collections separated in the field, such as when collecting off of different plants species. Tubes can also be handled easily with one hand while in the net. Vests, aprons, hip packs, and carpenter belts are handy ways to keep a number of collecting vials handy.

Most people will wrap the bottom of glass jars and vials with duct tape to reduce the chance of the glass breaking or at least shattering in a fall. Additionally, it is handy to place a bit of paper towel in the bottom of each jar to absorb the extra moisture from the bees collected and the nectar they regurgitate.

After bees have been captured and placed in a kill jar they usually quiet down in just a few seconds (depending on how charged the jar is), but if the specimens are taken out of the jar too soon and pinned some may "wake" back up and begin to move again, albeit usually only very slowly. Usually half and hour or so in the jar will prevent this.

An alternative to chemical based killing jars are jars, vials, or bottles filled with soapy water (a mix of water with a little dishwashing detergent is best) or alcohol. These are particularly useful for those of you who store specimens in alcohol or wash them prior to pinning. The best jars/vials have a tight fitting lid, are large enough to take a fair number of bees, fit in your pants pocket or are easy to hold in one hand along with the lid. Fill the vial about half full or a little less with soapy water. The jar quickly forms a constant head of suds while riding around in your pants pocket. When using it in the net it has the great advantage of immediately trapping any insect in the suds thus permitting you to clean out the net of as many specimens as you wish. With a normal killing jar you can accumulate 2-4 specimens with some effort but at some point more would be leaving than going in. The soapy jar is particularly nice when dealing with large nasty specimens.

You have to be a bit more aware of how you carry the jar when open (water seeking its own level and all that) but such jars can easily be used to directly collect off of flowers without a net.

Like specimens caught in bowl traps, specimens can be readily left in the soapy water for 24-hours and, while a bit soggy, will last for 48 without too much degradation. Afterwards specimens can be dried and pinned, drained and put in alcohol for long-term storage, or drained and wrapped with a piece of cloth (to soak up excess moisture and to prevent breakage) and frozen in a plastic bag.

The **advantages** of the Soap Jar appear to be:

- Don't have to lug toxic chemicals around
- Soap and water are readily available
- Restrains specimens immediately
- Can collect all specimens in a net at one time
- Inconspicuous to the general public
- Pollen and gunk are washed off while in the vial
- Cheap

Disadvantages:

- No pollen analysis
- Specimens are wet
- Jar needs to be held a bit more upright when open than a normal killing jar
- If cap not on correctly the water can leak

- Specimens have to be dried prior to pinning

Chlorocresol Humidor (Contributed by Rob Jean) - For those of us that enjoy net collecting but do not have the time to prepare and pin up our day' s catch the same evening, here is a technique for preserving specimens in a pliable state for extended periods of time (6 months-1yr or longer if moisture conditions are kept right). This is a simple technique I learned from Mike Arduser, Natural History Biologist, Missouri Department of Conservation, who uses it exclusively and rarely pins anything until he runs it through a chlorocresol humidor. The technique requires: 1.) a pint or quart-sized plastic container with a tight seal (I use a 4 cup or 1 qt Ziploc Twist N Seal container, but I have used on occasion up to 1/2 gallon sized containers) 2.) paper towels, 3.) chlorocresol (an antifungal crystalline substance with a sugar-like consistency available from Bioquip-item #1182B - $18.45/100 grams) (chemically = p-chloro-m-cresol or 4-chloro-3-methylphenol), 4.) a few strips of duct tape or its equivalent, and 5) a few drops of water.

To make the humidor, start by putting one rounded teaspoon of chlorocresol in the middle of one heavy paper towel or two lightweight paper towels. Then fold the paper towel(s) around the chlorocresol so that the chlorocresol is enclosed in the paper towel(s), and so that the folded paper towel(s) can fit into the bottom of the plastic container. Tape the loose edges of the paper towel(s) with narrow strips of heavy (duct) tape, using as little tape as possible. Thus, the container will have a securely sealed, but porous, clorcresol "packet" at the bottom.. You should do this under a fume hood or outdoors as chlorocresol has a strong smell and it can be harmful if inhaled or swallowed. Once the chlorocresol packet is in the container you simply have to play with the moisture level to get it perfect. In most cases keeping the paper towel damp (not soaked) is enough to keep the specimens moist and pliable enough to spread mandibles and pull genitalia, sternites, etc., but you will probably have to experiment a bit with this before you get it right. Specimens will dry up and become brittle if there is not enough moisture (but can be rehydrated in a few days usually). If there is too much moisture hairs will become matted on specimens and make them harder to identify later. Again, you may have to play around with the exact moisture conditions for the container/humidor you are using. One good thing is that the chlorocresol goes a long way (10 years or longer according to Mike A.). I have been using this method for two years and I am still on my original doses of chlorocresol in my humidors (I carry two with me at all times when collecting).

After I have the humidor I can catch specimens on flowers without an immediate need to pin. I can keep each collection event (different flower species, times of the day, etc.) in separate glassine envelopes or paper triangles within the humidor. Glassine envelopes and paper triangles are great to use in this situation because they are easy to write data on and because they allow the moisture in the humidor to get to the specimens. With periodical checking on the moisture levels in the container (I have to check mine every week or two) specimens can last several months to a year according to Mike Arduser. The specimens stay fresh as the chlorocresol wards off fungal agents. The chlorocresol also seems to relax specimens somehow which makes mandible spreading and genitalia pulling a little easier in bees. One caution: pollen loads (particularly apines and panurgines) can become soupy in the humidor and may inadvertently get stuck or plastered onto other bees. Also, specimens will smell like chlorocresol for some time after they come out of the humidor. Good luck and I hope this method saves some preparation time.

Types of Insect Pins to Use - Bees are usually pinned using pin sizes 1-3 with size 2 the most common. Pin size 1 is prone to bending when pressed into traditional hardboard lined trays and boxes but does nicely in foam units. Pin sizes below 1 should not be used as they are delicate, don't hold labels well, and end up bending if the specimen is moved or viewed often. Size 4 is generally too large for anything other than bumblebees. In humid environments stainless steel pins should be used to prevent rusting. Student pins should be avoided as they are cheaply made, the tips bend and the balls come off. Pins can be expensive and the least expensive way to purchase pins is to order them in bulk directly from Czechoslovakia where apparently most insect pins are made. Some newer inexpensive (same price as European steel pins) stainless steel pins are now available from China that appear to be of high quality.

Pinning - Bees can be pinned directly from the killing jar into boxes, or they can be washed first. If the bees are dry and not matted down then pinning directly to a collecting box is best as it preserves

the pollen load for future analysis and speeds up the entire process. However, if the bees are matted from too much moisture and regurgitate, then washing and drying them using the protocols listed in that section are advantageous as they result in better looking and easier to identify specimens. If the pollen load is not going to be analyzed then washing the specimens also has the advantage of eliminating the pollen from the scopal hairs and diminishing the "dustiness" of the specimens.

Each person develops their own process when pinning bees. Some pin under the microscope which usually results in very accurate placement of the pin, but many pin by eye. One technique is to hold larger specimens between the thumb and forefinger with the pin ready in the other hand. Another finger from the hand holding the pin can be used help hold the specimen steady and help in inserting the pin accurately into the bee's scutum. Others pin larger bees using a pair of forceps or tweezers, trapping the specimen on a foam pad. Expanded polyethylene foam (often referred to as Ethafoam) or cross-linked polyethylene foam (our preferred foam) is better than polystyrene foam (usually referred to as Styrofoam) for pinning purposes. Styrofoam is not supportive enough; both labels and specimens will bend too much when pinned upon Styrofoam.

Specimens are best pinned through the scutum between the tegula. If at all possible the pin should be to one side or the other of the mid-line. The midline of the scutum often contains features that are very useful in identification and these can be destroyed by the pin. Most museums prefer that specimens be pinned on the right side. The height of the specimen should be such that there is plenty of pin left above the top of the specimen. This will permit someone to pick up the specimen without breaking off an antenna but the specimen should not be so low as there isn't room for two or more labels below plus room for the pin to go into the foam. Height can be set with purchased insect pinning blocks, or with pieces of foam of the correct height, but be aware that large bumblebees need to be adjusted downward to make sure there is room at the top of the pin for grasping. After a few uses of the pinning block it is best to adjust specimen height by eye as that will be the quickest.

Gluing Small Specimens - If specimens are too small to be pinned they can either be placed on a point, glued to the side of a pin, or attached as minuten double mounts. Glues that are reversible should be used and include Elmer's Glue Gel, white glues, clear nail polish, shellac, hide glue, and others. The use of points is traditional. Points are very small, acute triangles of stiff paper that are cut out using a special punch which can be ordered from entomological supply houses. The pin is placed through the base of the point, the pointed elevated on the pin to the same height as pinned specimens, and the small bee is glued to the tip, usually on its underside.

When pinning a specimen directly to the pin, rather than to a point, the specimen is glued on its side or the underside between the thorax and abdomen. Most museums prefer that specimens be glued on their right sides.

Gluing specimens to the side of the pin has the advantage of speed, better prevention of glue hiding useful characters, and a specimen that is easier to view under the microscope because its axis of rotation is minimized and the point is no longer there to hide the view or block the light. Specimens should be glued to the pin at the same height as those that are pinned. We use tacky glue in our lab, it's a thicker white glue than the more common Elmer's and school glues and permits specimens to be glued immediately and set up right in the box. Use of regular white glue or glue gel requires leaving the pin resting on the specimen for 5-10 minutes prior to picking up the pin. From our limited investigations Aleene's Original Tacky Glue in the gold bottle appears to be the best gripping tacky glue. Others have had good success using clear nail polish and Elmer's Glue Gel.

While unorthodox our current process for pinning involves washing and drying specimens in the machines listed in this document, placing them in open labeled petri dishes and then letting them completely dry for a week or more prior to pinning. If time doesn't permit pinning right away then after a week or so of drying the petri cover is replaced and taped on and the specimens stored in their dishes. When ready to pin all the specimens are laid out on a large foam pinning board and a pin glued to the side or underside of each ….including the largest specimens…. using gel glues. Large specimens require larger amounts of glue and all specimens need to have pin and glue attached to the body of the specimen rather than to a wing or leg.

We use a magnetic pin holder that attaches to the wrist. These are available in hardware stores, online, or in sewing shops. A sawn off section of bolt (we use 2 of them) is handy to have on the wrist holder the threads will separate the pins for easier pick up. We then run a small line of glue on the side of our thumb on the same hand that has the wrist holder. The glue is kept in a small quiller's glue bottle inverted in a canister so that glue is always ready and does not dry in the tip. A reverse set of tweezers is use to pick up a pin by the head or the tip, it is dipped into the glue line on the thumb at the proper specimen height and then placed on the specimen on the pinning board. Because the specimens are so dry care must be taken to place the pin gently, but has the advantage in that the specimen can then be easily be picked up and moved to other places on the board (except for the biggest specimens). With a little practice it is easy to achieve pinning rates of 250+ per hour.

After the bees have dried for a couple of hours they are then transferred to boxes. In some instances that transfer can be accomplished with the attachment of labels.

Parchment paper is to have around when pinning bees. It is a silicone impregnated piece of paper that can withstand the heat of an oven but is super slick. When we are pinning bees, we now like to use glue gels which have a longer work time and dry crystal clear (and also are easily reversible). However this requires laying the pin with glue on it on the insect while it is lying down and at times glue gets on the substrate they are lying on and the specimen pulls off when you pick it up. Parchment paper alleviates much of that problem as glue doesn't stick well to it at all. Another nice thing is that you can put dried specimens on the paper and they will easily slide around without sticking and breaking, so they are easy to position. We now dump dried specimens onto the paper, pull up the sides, which causes the specimens to slide into the center, and then use the fact that all the specimens are in a line to more rapidly pin them. The paper is placed or pined to a large foam board so you can press the tip of the pin and cause the specimen to rise up so you can pick it up.

Minuten double mounts are not used very often, but do create the best looking mounts. A tiny bit of crosslinked polyethylene foam is pinned to the same height as a regular specimen. A minuten pin is added to the right side of the specimen and then inserted into the foam block. On the down side this takes a lot of time to accomplish.

General Videos on how to mount and work with insect collections is available at:
http://www.bugs.nau.edu/learning_modules.html

A videos for how to pin bees is under development at: http://www.youtube.com/user/swdroege

Bee Boxes - There are a variety of drawers, cabinets, and boxes available to hold specimens. We prefer to use the simple cardboard specimen box with a completely detachable lid and an Ethafoam bottom for everything except for housing our synoptic collections. These boxes are stackable, the date and location can be written on the outside in pencil and then erased when reused, relatively inexpensive, and, unlike hinged lid boxes, are convenient to use in cramped spaces on a desk or worktable. Such boxes can be made from scratch and we provide in a separate document directions on how to make specimen boxes from pizza boxes.

After a batch of specimens is washed dried and pinned we place them in a cardboard specimen box. At the upper left hand corner of the box, a tag with the date, place, site or batch number is pinned. This tag is usually the same tag that was placed in a batch of specimens stored in alcohol when the specimens were originally captured. A line of specimens is pinned to the right of the tag and continues running from top to bottom and left to right like a book until complete. The next tag is placed immediately thereafter and so forth until the box is filled. In general it helps if each box contains specimens from only one region. We label the year across the top of the box, then the month and then the locality, so that we can quickly pick out the box we want.

Control of Pests – Simple cardboard boxes are not pest proof. Dermestid beetles are the primary pest of insect collections. Fortunately, infestations are usually small usually one beetle larvae in a box scattered here and there. An infected specimen is usually easy to spot as small black droppings and shed skin are visible below the specimen. Control and prevention take place according to the literature by freezing the box at -20C (about zero degrees Fahrenheit) for 3 days, thawing for a day

and then freezing for another 3. In a pinch kitchen freezers appear to work too. Mothballs and pest strips can be effective but carry some apparent health risks with long-term exposure. Spring is a good time to freeze your entire collection as that is when dermestids appear to be most active. An excellent means of keeping your collection pest free (particularly if using cardboard boxes) is to keep each box in a large zip lock bag. Note that you should have let the specimens dry out thoroughly after pinning (one month or so) before enclosing them in the bag.

In humid conditions (such as July and August in Maryland) unprotected specimens, particularly those just caught, can turn into balls of mold. Either take them into an air-conditioned space or put them in plastic bags or tightly closed bins that contain active desiccants. Keeping specimens in a refrigerator or cooler without moisture control will ultimately lead to mold too.

Labels – Following pinning labels are produced for each batch of specimens. We use a label generating program available on the www.discoverlife.org web site. Each batch or site is given a unique site number and each specimen is also given a unique specimen number. On each label the specimen number and site number are listed as well as the country, state, county, latitude, longitude, date of collection, and collector. A small data matrix is present on the label that encodes the specimen number and permits the specimen to be scanned with a hand-held scanner directly to a database while remaining specimens remain in the box. These data matrices are included automatically in the free Discoverlife system (http://www.discoverlife.org/label/) or can be added using commercial software such as BarTender (http://www.seagullscientific.com/).

Dan Kjar has generalized the Discoverlife label program so it will print out on a laser printer. You can use his simple web based form (http://bio2.elmira.edu/fieldbio/) by following the link at the bottom of the page for insect labels. Each label is unique based on the specimen number.
Depending on how many labels you are making and your Internet speed it will take a little time to build the label page. 50 labels take about 20 seconds to assemble. The system will be integrated into Discoverlife soon and when that occurs Dan will announce that on his site.

In a good museum cabinet specimens deteriorate only very slowly and can last for well over 100 years. That is not true of the paper used in making labels. Paper that is not archival or acid free gradually deteriorates. Fortunately, archival paper is readily available in office supply stores. A heavier weight paper is also important to use so that the label stands up to handling and the pinning process. A 35- 65 pound paper is good label stock.

Specimen labels are quickly added to specimen pins by laying them across a piece of Ethafoam, the thickness the desired height of the label on the pin. To increase the durability of the Ethafoam it can be glued to a piece of plywood to form a sturdy pinning surface. To manufacture a pinning board, smear white or wood glue across both surfaces, rub together, and then place another (unglued) board on top of the foam and pile books or other heavy objects on that board to clamp the foam and board tightly together. Let dry overnight. It can then be used as is or the edges can be trimmed with a saw for a nice and tidy look. Labels are oriented along the same axis as the specimen. Prior to putting labels on specimens do a quick check to make sure the label information matches the row tag.

Gretchen LeBuhn has a system for making labels in Word which is explained below.

1) Open up a new Word document and just type the label like you want to see it, i.e.,

   CALIFORNIA: Napa Co.
   Rector Reservoir, 60m
   3.2 km NE Yountville
   38º26'13"N,122º20'57"W
   17 March 2002, ex: Vicia sativa
   G.LeBuhn, R.Brooks #2002001

   As a numbering system I make the bees collected at a single species of plant an individual

collection record. For example, the bees that I collected on *Vicia sativa* at Rector Dam were collection #1 and those collected on *Lupinus bicolor* were collection #. 2. I recommend just keeping this system going or some similar system so that you can identify and talk about each collection separately each year. You use #2002001 for this year and then start over next year with collection #2003001, etc. The point is to adopt some system by which you can talk about any particular collection event in a multi-year study and that it has a numerical identifier.

Now back to making labels

I make a label log which I actually type directly into my data base and then extract and put into Word. I cut and paste a copy of each collection event the number of times needed to label the bees in each lot. I do this one in long continuous roll. When I am finished I put it into column format to fit more per page.

2) Now I have all of my labels duplicated like this

CALIFORNIA: Napa Co.
Rector Reservoir, 60m
3.2 km NE Yountville
38º26'13"N,122º20'57"W
17 March 2002, ex: Vicia sativa
G.LeBuhn, R.Brooks #2002001
CALIFORNIA: Napa Co.
Rector Reservoir, 60m
3.2 km NE Yountville
38º26'13"N,122º20'57"W
17 March 2002, ex: Vicia sativa
G.LeBuhn, R.Brooks #2002001
CALIFORNIA: Napa Co.
Rector Reservoir, 60m
3.2 km NE Yountville
38º26'13"N,122º20'57"W
17 March 2002, ex: Vicia sativa
G.LeBuhn, R.Brooks #2002001
etc. - the above was for 3 bees collected in Collection #1 and then leave a blank line between collection events so that I can see where each separate collection event starts.

3) I now go up to pull down the "Edit" menu and then select "select all"
I go up to pull down the "Format" menu and select "Font" and type into the "Size" window the number 3 (for 3 point font) and click okay
I go up to pull down the "Format" menu and select "Paragraph" and select under "Line Spacing" the word "Exactly" and under "At" select "3 pt." (this sets the leading or space between lines)
I go up to pull down the "Format" menu and select "Columns" and under "Number of Columns" I start with 8 and then under "Width and Spacing" is set the "Space" (that is space between columns) to 0.00. I check with Print Preview which is selected after pulling down the "File" menu. The trick here is to get the columns as close as possible to each other without any lines wrapping around. Sometimes I can get 9 columns at others when the label lines are longer I can only get 7 columns. 8 columns is my usual maximum column width.

You are done and can now print onto your acid free or archival, 100% linen ledger #36 white paper. Cut the labels out neatly not leaving white around the edges and place the labels on the specimens with the top of the label on the right with the specimen's head going away from you.

You can email Sam Droege (sdroege@usgs.gov) for Excel spreadsheets that will print out blank determination labels (labels that give the species name) that you can modify with your name and date.

Pens – When writing locality or determination labels by hand archival ink should be used. Rapidographs were most commonly used in the past, but they have almost entirely been replaced by certain technical pens as Rapidographs tend to clog when left unused for any length of time. Technical pens in sizes 05 and 001 are the best and are available in art supply stores and from entomological supply stores. Be sure that they state that they are using archival ink.

Organizing Specimens for Identification - After the specimens are labeled and those labels checked against the original row labels in the box, the specimens can freely be moved about for identification. We usually sort and identify only those specimens in a single box rather than try to merge specimens across many boxes. Others color code their projects with colored pieces of paper placed under the locality label so that projects can be tracked visually in large groups of specimens. In this way multiple projects in multiple states of completion can be tracked and are less likely to become entangled.

When identifying specimens we make a first pass through the box and identify everything identifiable without using a guide. These are taken out of the box and pinned to a separate foam board. This board is set at a 45 degree angle next to our microscope. As new species are detected a determination label is created (available as a modifiable Excel file from us). The determination label is pinned to the board separately from the specimens so that it can be easily viewed. All subsequent specimens of that species are then placed to the right of the label. Bees that cannot be immediately identified are kept separate and identified at the end using computer and paper guides.

Once Bees are all identified they are then placed back into the original box. Bees are placed in the box in rows starting at the upper left corner and going from left to right top to bottom with determination labels interspersed at the beginning of a new group of species. Females are placed so their label is positioned vertically and males positioned so that their labels are horizontal. Positioning the sexes this way permits those who enter the data to quickly ascertain and check the sex without having the check the label.

Entering Specimen Data - In the system that we use each specimen has a scannable matrix on its label and data entry consists of scanning each specimen directly from the box into an Access database. The scanner has a feature that sends a linefeed character at the end of scanning in the number, thus moving the cursor down one line to the next cell where the next specimen can be scanned…and so forth until that species is completely entered. Access has a nice feature that permits default values for database fields. Thus genus and species field defaults can be set to the current species being processed and as the scanner enters a number and drops down a line the data for the other fields are automatically entered. Thus data entry becomes simply a matter of pulling the scanner trigger and periodically resetting species and sex information either by hand or by changing the defaults. Access has another nice feature which sets off an alarm or sound if a number is entered twice, something that can easily happen in a crowded box of specimens.

After the data are entered by one person another person cross-checks the specimens and the database. After that final check the bees are dispersed to final resting spots in museums, sent to other colleagues, or their pins are recycled for reuse.

Shipping Specimens – When shipping pinned specimens (see section in this manual for traveling and shipping alcohol specimens) whatever type of box you send them in should have the following characteristics. The specimens should be firmly pinned to the foam. There should be pinned specimens or pins in all four corners of the box to support the cardboard that will go on top. Some people will also pin loose cotton wadding in the corners of the box so that if a specimen comes loose it will be trapped by the cotton. Cut a piece of cardboard that will fit snuggly inside of the box and rest on top of the specimens. Do not use foam rather than cardboard for this layer as it will engulf the tops of the pins and cause problems when removed. Two pieces of tape can be affixed to the top of the cardboard in a way that they will form handles that will help remove the cardboard without upsetting the specimens below. Simply press one end of the tape to the cardboard and then fold the other end back on itself so the sticky sides meet. If there space between the top of the cardboard and the lid of the box put in some bubble wrap or packing peanuts so that when the lid is closed it slightly compresses the cardboard to the tops of the pins keeping them in place during travel. Tape the lid of the box closed. Put the box of specimens into a larger box with at least 2 inches of free space on all

sides. Fill the box with packing peanuts, bubble wrap, etc. and ship. In the U.S. we have found parcel post to work fine, albeit not as fast as Fed Ex or UPS. For valuable specimens all companies provide tracking and confirmation of receipt services.

Choosing and Purchasing a Microscopes - Using bowls or nets it is easy to quickly amass a large collection of bee specimens. Unfortunately, unlike most butterflies, bees (even the bumblebees) need to be viewed under a stereo or dissecting microscope to see the small features that differentiate among the species. While even inexpensive microscopes and lights can be of some use, in the long run they lead to frustration. Inexpensive microscopes usually have poor optics, very low power, small fields of view, difficult to set or fixed heights, and their stands are usually lightweight and often designed such to make specimens difficult to manipulate.

Unfortunately, a good microscope is not cheap. New, our experience is that an adequate microscope costs over $1000 and good ones run over $2000. That said, microscopes with even moderate care can be seen as a one time investment. Additionally, because a good microscope has optics that can be adjusted and cleaned (unlike most inexpensive ones) it is usually safe to buy a used or reconditioned microscope from an online dealer (buying off of E-Bay or Craig's List is more risky as the seller has less of a reputation to risk). There are many used microscope sites and we have purchased microscopes from several of them and have never had a bad experience. In two cases the purchased microscopes had a problem and in both cases they were repaired for free. Usually, used prices are about half the cost of new.

Good stereoscope brands to consider that we have experience with include Leica, Zeiss, Olympus, Wild, Wild-Heerbrug, Nikon, and Meiji. We can supply you with some model numbers from our collection or you can send us web sites with the microscopes you are considering and we will be glad to give you our impressions. Of special consideration are the Bausch and Lomb StereoZoom series. These microscopes have been around for years and often form the core of college biology and entomology department teaching labs. These are adequate to good scopes and we have about 5 in our lab. They are readily available used from $500 -$900 online. Their negatives include a view that is not as good as the better scopes and I my personal preference for zoom magnification on the side rather than on top as is the case with student scopes. Finally, be aware that many of these scopes only go up to 30X power with the standard 10X oculars, though higher powered models exist and higher power replacement oculars are readily available.

Magnification - Magnification power needs some mention here. Any adequate to good scope will have variable power settings. We have never seen any instance where the lowest magnification was an issue, but a useful scope should go up to about 60X power or higher, something that many good scopes do not achieve with the standard 10X ocular. If the scope does not go to that high a power it is a simple matter to change the magnification by purchasing a higher power set of ocular pieces (these are the eyepieces that you look into). Oculars simply slide into tubes on top of the scope and readily removed (as some of you who have turned a microscope upside down have found out) although sometimes there is a set screw that needs to be released first. That said, replacement oculars, while almost always available for every model and brand, can be expensive to purchase. Magnification is determined by multiplying the magnification of the ocular lens (this number is listed usually on the side of each ocular piece but sometimes is found on the top and is most commonly 10X) by the zoom or magnification level which is listed on the zoom knob. Note that some manufacturers list the zoom levels multiplied out with the assumption that you are using 10X oculars.

Most higher-end microscopes come with a zoom magnification where all powers are available in any increment. In some scopes, powers are available only in steps. I haven't found the scopes that move in increments to be any major hindrance. I have found, however, that scopes that have the magnification/zoom feature available on the sides of the scope in the form of a small knob are the easiest and quickest ones to use. The ones with the knob on top or located as a movable ring around the base of the scope head take more time to change. Often the magnification is changed several times when viewing a specimen.

Measuring Reticule - Some microscopes come with a measuring reticule in one of the oculars but most do not. A measuring reticule is a very small ruler etched into a piece of glass. These are useful for taking precise measurements or, more often the case, taking relative measurements. This piece

of glass is inserted into the bottom side of one ocular. All or almost all oculars are built in a way that they can be taken apart for cleaning. Often there is a threaded tube inside the body of the ocular that holds the lenses in place. If taking one apart be gentle as the threads can be delicate. Measuring reticules can be ordered online or some microscope dealers will custom make one for you.

Adjusting, Cleaning, and Storing Microscopes – Most good scopes are fairly sturdy and don't go out of adjustment without suffer some sort of blow. In our experience we have come across two primary adjustment issues. The oculars don't focus in the same plane or image the oculars are processing are out of alignment. If no matter how much you play with the width of adjust of the eyepieces the images don't completely align then the scope has significant problems and will have to be repaired professionally.

Differential focus is usually something you can fix. Small differences in the focal distance of the oculars can be accommodated by your eyes, but at some point they eyestrain will become apparent and uncomfortable. In most scopes one or both of the tubes that the oculars slide into are adjustable. These focusing eyepieces are easy to determine as there are zero, plus, minus, and tick marks to align. To adjust the focus so that both eyepieces are in the same focal plane, place a piece of graph paper or something similar on the base of the scope and shine a good light on it. Adjust any adjustable eyepieces to zero. If there is one eyepiece that is fixed then open that eye and close the other. Change the focus of the microscope so that the grid is in sharp focus. Now close that eye and open the other. If the grid is not in alignment then adjust the focus of that eyepiece until it is. If, as it sometimes rarely happens, after adjusting in both directions you still cannot get the eyepiece in focus then try sliding the eyepiece up slightly to see if that works. If that doesn't work then likely the other eyepiece is the one that has to be adjusted upwards. If the microscope has set screws you can use them to fix the height, if not then you will have to work out some other mechanical means. Usually, however, such and extreme situation indicates that something is generally wrong with the scope or the oculars and you might check the oculars to see if the lens are loose or you have mismatched oculars from some other scope.

The objective lens of a microscope almost never needs to be cleaned. However, the top lens of the oculars often do, particularly if the person using the scope likes to press their eyes close and wears make-up (mascara is the worst). After trying a number of cleaning methods we use lens paper and window cleaner as needed. To keep the dust out of the oculars we found it simplest to just put a baggie over both lenses.

Holding Specimens and General Microscope Setup - Most people when viewing specimens under the microscope naturally place them on a piece of clay, foam, or some sort of stand. We try to avoid this as it is far faster to view specimens when held in the hands of the observer. To hold specimens, pick up the head of the pin using the thumb and forefinger of your dominant hand. This allows you to easily spin the specimen around the axis of the pin. The point of the pin is then either lightly pressed against the middle or forefinger of the other hand or some hold it between their thumb and forefinger.

It is important to place the bottom sides your hands on the base of the microscope, this stabilizes the hand so the specimen is held steadily even under high magnification. With hands in place, the specimen can be quickly and efficiently rotated in all directions while the observer looks into the microscope. To take full advantage of this the focal plane of the microscope should be raised such that the specimen is roughly in focus (usually about 3 inches above the base of the microscope) when the hands are in place. Once this focus is set on the microscope it is never moved again as any change in focus is accomplished by moving the specimen rather than moving the focus knob. If the magnification level needs to be changed then the hand holding the head of the pin can retain the specimen while the other hand changes the magnification without having the eyes leave the oculars.

The final part of microscope setup is to adjust your chair or the table holding the microscope such that you do not have to bend or strain your body to look into the microscope.

Acknowledgements: John Ascher, Harold Ikerd, Gretchen LeBuhn, Jack Neff, and Karen Wetherill had valuable additions to this section.

The Bee Bowl - Bee Bowls are small colored plastic bowls or cups that are filled with soapy water. Bees are attracted to these colors, fly into the water, and drown. Originally meat trays (a.k.a. pan trap) and 12 oz. salad bowl were used. Later field experience and experiments demonstrated that size of bowl was not necessarily correlated with capture rate (see http://online.sfsu.edu/~beeplot/ for several reports that document those results or contact Sam Droege and Gretchen LeBuhn for a unpublished experiments on such).

Currently, the USGS lab is no longer using bowls at all but items that are listed in plastic manufacturer's catalogs as soufflé or portion cups, the type of small plastic cup that potato salad might come in at the deli. Several manufacturers make such cups, but Solo is the line that most people have experience with. These cups usually come in their native translucent plastic color, which is not at all attractive to bees. However the Solo 3.25 oz. cup, which is steep sided and stable on the ground, does come in white (model number: p325w-0001). This particular model is attractive in that plain white is highly attractive to bees and it also provide a nice base color when painting blue or yellow.

For travel the Solo 0.75 oz. and perhaps the 2 oz. cups are nice sizes to carry in your luggage as they minimize water use, but will lose too much water too quickly in hot, low humidity areas. The 1 oz. cups are steeper-sided and narrow (and therefore more unstable), however, this model may be worth investigating for use in desert areas. Surprisingly, loss or upsetting by the wind is rarely an issue with bee bowls.

The white cups usually need to be ordered by the case from a local Solo distributor (that means about 2500 cups) while the other sizes are widely available and very inexpensive online. Solo distributors can be located by calling 1-800-FOR-CUPS. The solo product line catalog is online and can be viewed at: www.solocups.com. The price for a case of the white bowls is usually in the range of $50 to $85. Do a Google search on the model number and see what you can find. If you are in a bind email me (sdroege@usgs.gov) and I will send you some at cost. Party stores are also a possible place to find white bowls and certainly have plenty of non-fluorescent colored bowls to investigate.

Painting Bowls - Originally, when bowls rather than soufflé cups were being used, colored plastic bowls from party stores or other sources were used to capture bees. The usual colors were yellow, white, light blue, and dark blue. Those worked well, but fluorescent yellow and fluorescent blue were found to be much more effective in the East (and field experience indicates the same to be true in the West). However note that Laurence Packer has found that cactus bees, especially *Macrotera*, seem to be attracted to dark blue and even red colored bowls (red bowls attracted absolutely zero bees in the East). He didn't compare these with fluorescent colors, but both those colors collected more M. texana than did either white or yellow. A literature is accumulating that indicates that there are individual species preferences in bowl color and that these preferences appear to shift regionally and perhaps even seasonally.

Commercial fluorescent spray and brush paints vary in their color characteristics and availability by brand and location. In 2003 we experimented with creating a standardized fluorescent yellow and blue paint from scratch and have done so with the help of the West Coast Risk Reactor Company (800-803-5281) (http://www.riskreactor.com/) who can be contacted both for price and ordering information as well as a complete spec sheets that list this paint's components . In 2004 we experimented with some different formulations and found a fluorescent combination from the East Coast Guerra Paint and Pigment that works better than the system we had earlier tried. The liquid pigments mix much more readily than the dry pigments do and the base paint they supply sticks well to plastic. When ordering from Guerra (212-529-0628) specify:

Silica Flat
Yellow Fluorescent
Blue Fluorescent

Jody has been the person we have worked with.

You can order online at:

http://www.guerrapaint.com/tandc.html

To get to the fluorescent pigments click on “Search By Group” and run the scroll bar down to the bottom and click on “Flourescent.” Choose “Flourescent Blue” or Flourescent Yellow” from the list of the size and quantity you desire.

To get to the silica flat click on “Search By Type” and choose “Binder” from the list choose the size and amount of “Silica Flat” you need.

The ratio is 16 oz. of dye to 1 gallon of Silica Flat Paint. You can mix it with a stick without difficulty.

For future reference their Fluorescent (water-dispersed pigments) formula is:

Water	47.5%
Methocel – KMS – Thickener – Methyl Cellulose	0.45%
Defoamer – Drew -647	0.80%
Tamol 731 – Dispersant (soap)	1.25%
Flourescent Pigment	50.0%

The formula for the Silica Flat Acrylic Latex Paint is:

Acrylic-Latex
Calcium Carbonate
Kaolin – Clay
Tanium Dioxide (I think this should be Titanium Dioxide)

No percentages were given and these are only listed as the major components, there are likely to be surfactants and other things in here as well. The carrier of the dye is not as important as the dye itself.

Most standard types of paint and paint primers do not stick well to plastic (The primer from Guerra does, however). Many commercial spray paints have added compounds that do help with adhesion to plastic. However, if many bowls are being painted cans of spray paint becomes both expensive and wasteful. We have experimented with using liquid paint in compressed air spray guns but the paint inevitably clogged the sprayer (even when thinned) and it was difficult to coat the sides and bottom of bowls uniformly. That said, when it was working spraying is fast, so if you figure out a good spray system please let us know. Oil-based primers seem to work the best on plastic but primers that have a shellac base or are formulated for glossy surfaces may do equally as well. There is a nice spray can primer by Krylon that is specially formulated for use on plastic called Krylon Fusion Dover White Paint that can come in handy. If priming prior to painting the color coat a white primer provides a good base color for fluorescent yellow and a gray primer (a paint shop will tint your primer for free) works best for the fluorescent blue. Sanding the bowls also allows paint to stick, but takes a great deal of time.

In 2003 we completed a series of small experiments that indicated that the amount of surface area painted on a bowl did influence the number of bees captured. When completely painted and partially painted bowls were placed adjacent to on another the completely painted bowl (about 50% more) caught significantly more bees. It is possible that this effect may diminish if bowls are spaced apart rather than adjacent to one another.

If using a commercial spray paint, the Krylon brands seems to be composed of the same colors as those from the paint specialty shops, but this brand can be hard to find in many areas (particularly the fluorescent blue).

In 2004 a student placed a set of 3.25 oz. plastic bowls painted fluorescent yellow and blue from 2 separate pigment suppliers out in the sun (empty) for 6 months starting in early September. Over that time the blues became slightly faded and the yellows quite a bit more so, but both remained pigmented. She mentioned that the fading was only noticeable after 3.5 months of being out in the

sun and then only in comparison with bowls that had been kept indoors. Consequently, it looks like the pigments from Guerra and Risk Reactor are quite long-lived and will essentially outlast the life of the plastic bowl - which became quite brittle towards their end. It would be interesting to try this same experiment in some mid-summer southern hemisphere desert and see what the life expectancy is under those conditions.

How to Set a Bowl Trap - A bowl trap is set when it is filled with soapy water and left outside. The soap decreases the surface tension permitting even small insects to sink beneath the surface. Most insects thus trapped stop moving within 60 seconds of hitting the water. However, we have found that if pinned right away after being trapped some will begin to do a slow crawl if not either placed either in alcohol for several hours or put in the freezer prior to pinning. Unpinned insects that do begin to move after being in a bowl never regain full functionality and usually simply stand or move around only very slowly.

We have found that the amount of water in a bowl does not affect the capture probability of bowls. However, in hot and arid climates bowls can dry out if not completely filled or if the bowl is too shallow. In terms of the type of soap to use in the bowls we suggest that people use Blue Dawn Dishwashing soap it readily available and appears to function similar to other brands. Be aware that citrus-scented detergents and ammonia mixed with water will decrease the bee catch compared to other detergents. Laundry soaps have been tried and do work, but contain so many fragrance chemicals that we fear that changes in formulation could easily affect the capture rate. We have tried adding salts, floral oils, sugars, honey, and other compounds to bowl trap water but found that captures were either the same or lower than those with dawn dishwashing liquid. While some bee bowlers add detergent directly to each bowl, we have found it easiest to add a big squirt of dishwashing liquid directly to a gallon jug of water and pour it from there.

When using bowls in a collecting rather than an inventory or monitoring situation, it is often convenient to leave bowls out for longer than a day given that the water doesn't completely evaporate. Specimens appear to not suffer any substantial deterioration for at least 48 hours, perhaps more. Laurence Packer has found that propylene glycol can be left in bowls for at least 3 weeks without substantial loss even in early summer in the low rainfall southern Atacama Desert. A bit of formalin in the bowls decreases the attraction to vertebrates. Digging the bowl into the substrate may be necessary when bowls are left out this long. When bowls are placed near the level of the surface tenebrionids, scorpions, and the occasional lizard may also be collected in some circumstances.

Matthew Somers ran some experiments in Ontario that indicated that there wasn't a significant difference in the number of bees captured between yellow bowls filled with soapy water versus those filled with propylene glycol. Interestingly, he found that about 33% of the bees that landed in either fluid would escape the bowl and that rate apparently varies with species. He also noted that a high proportion of insects were attracted to the bowls but either only flew low over them or simply landed on the rim. This was a small pilot study, worth repeating and expanding upon.

Propylene glycol is often difficult to find. However places to look are RV centers, swimming pool supply stores, livestock supply stores, and heating and cooling supply houses. Heating and cooling suppliers are often the best as you can order the raw materials directly with no added colorants rather than getting a preparation that has been diluted and may have additional chemicals added to it. One common supply company for the basic material is Comstar (http://www.comstarproducts.com/).

Several people have tried using urine instead of water in the bowls (bees in tropical areas are often attracted to urine soaked soils) but no great increase in catch was noted of the few who tried.

Sunny days are best when setting out bee bowls. The effects of temperature are often unclear but catch appears to be reduced in the spring if temperatures are in the 50's F or below during the day. However, in the fall temperature seems to have less impact. Cloudy days catch few bees, and rainy ones never catch bees.

Where to Set a Bowl Trap - The best places to put bee bowls are exposed open settings (e.g., fields, roadsides, grassy areas, barrens, sand), places where bees are likely to see them. In North America

this also extends to deciduous woodlands prior to leaf out. Within these habitats bowls left under any dense vegetation (e.g., thick cool season grasses, leafy shrubs), will catch few bees. Open warm season grasslands often have good capture rates of bees if the grass overstory is not too thick. The general rule of thumb is that if you can easily see the bowl then bees can too. Flowers need not be apparent in an area in order for catches to be quite high. However, the presence of a superabundant nectar and pollen source (e.g., creosote bush, mesquite, a field of blooming mustard) often appears to lead to low bowl capture rates.

Bowls seem to work in open habitats around the world (e.g., Fiji, Taiwan, Thailand, South Africa, Central America, and South America). The bycatch in bee bowls can be very interesting with parasitic hymenoptera, sphecids, vespids, skippers, thrips, flys, and other things that often come to flowers often more abundant than bees.

In tropical Central and South America Dave Roubik (Panama), Steve Javorek (Belize), and Gordon Frankie (Costa Rica) have all noticed that soapy water bowls capture almost no bees in closed canopy or canopy top situations (however, more extensive tests are warranted here), but are successful in open habitats. Roubik also has had good success with capturing stingless bees using a honey solution (or sucrose when honey is not available) either in bowls or sprayed on vegetation.

Laurence Packer writes about strategies for collecting bees in bowls: "When attempting to collect Xeromelissinae, some of which are oligolectic, I have often put pans out by suitable looking flowers en route to a different collecting spot. The success rate has been remarkably high and I have found males of species only collected by net as females, and females of species only collected by net as males using this method.

By placing pans adjacent to the flowers visited by oligolectic species. I have managed to collect samples directly into buffered formalin and absolute alcohol for histology and DNA respectively - though capture rates were not high, in a couple of hours a couple of pans of each collected enough for my needs."

In general small bees are sampled well in bowls, but larger bees often need to be netted.

Most researchers put bowls out in strings rather than as single bowls. Capture rate per unit of field time is much higher this way. Once a location has been chosen in which to place bowls it takes relatively little additional time to place many bowls as compared to just one, particularly when compared to the cost of traveling to a new place. An internal study available from Sam Droege (sdroege@usgs.gov) indicated that the variances for characterizing the species richness of a single site may level out around 15- 30 bowls.

Bowls placed immediately adjacent one another other have been shown to have reduced individual per/bowl capture rates. In studies in Maryland using 3 separate trapping webs in open fields a distance of 3-4 meters was calculated to be the threshold below which bowls competed with one another for captures, but did not compete above that level. In Brazil additional species were captured when bowls were elevated off the ground, but in the Eastern United States no additional species were captured and capture rates were much lower than bowls placed on the ground. In the East, when large black circles were added to the bottom of cups catch was decreased, while adding small Andrena-sized markings to a bowl did not change capture rates.

A procedure has been developed for monitoring bees on plots and is available at Gretchen LeBuhn's bee monitoring web site (http://online.sfsu.edu/~beeplot/). A scheme for replicable monitoring or inventory of bees over larger landscapes is being developed by Sam Droege (sdroege@usgs.gov). That design will likely include the superimposing of a grid over the landscape with numerous individual transects of 15-30 bowls taken within each grid in appropriate habitats and used as replicates. Because a completely random or systematic sampling scheme is impossible in most public/private landscapes, an effort will be made to GPS all points and collect habitat co-variates to permit counts to be adjusted over time.

How to Collect the Bees Once Trapped - At each bowl it is usually best to remove all moths,

butterflies, and skippers as well as slugs, and very large bodied non-hymenoptera (e.g., grasshoppers and crickets). These groups tend to contaminate the other specimens when placed in alcohol. Following their removal the remaining specimens can be dumped along with the water in the bowl into an aquarium net, sieve, or tea strainer. The key matter when choosing a particular type of bee strainer is that their mesh be fine enough to catch the smallest of bees, some of which may only be 2-4mm. If using an aquarium net, look specifically for brine shrimp rather than regular nets. In general, most kitchen sieves are too coarse while most tea strainers have nice fine mesh. Brine shrimp nets are our favorites.

Usually researchers pool all the bowls from one transect or plot rather than keeping individual trap data separate, as handling time increases greatly when collecting from individual bowls. Many researchers also wash the soap from their catch in the field using a squirt bottle, however, we have found that not to be necessary. Most researchers store their catch in 70% alcohol in whirlpaks. We usually use a plastic spoon to gather the specimens from the brine shrimp net and then transfer them to the whirlpaks. This works with the strainer, but not as easily. Alternatively, you can pick out the mass of insects in the net or strainer with your fingers and dump it into an individual whirlpak. However, Frank Parker uses a larger sized whirlpak along with a small tea strainer and then gives the strainer a sharp rap when in the bag to dislodge all the insects at once. Others dump specimens directly into mason jars, or baggies.

Isopropyl, ethyl, or denatured alcohols are all appropriate for storing insects, but isopropyl should never be mixed with the other alcohols. You can go to the pharmacy and almost always find small pint bottles of ethyl alcohol, ethanol, or denatured alcohol (they aren't very consistent in what they call them), if not, they will readily order it for you all of these products are altered (i.e., denatured) so that they aren't drinkable. At the hardware store they have gallon and pint cans of denatured alcohol (these are 100% alcohol and almost always contain a mix of 50% ethanol and 50% methanol). The drug store alcohol appears easier to work with as it is made with a smaller amount of methanol.

Often the purchased alcohol needs to be diluted to achieve the right percentage (70%). All hardware store alcohol should be considered to be 95% alcohol. Drug store alcohol can be close to 100% but usually is something less, thus you will have to read the bottle's label to check. Note that most cheap dollar type stores sell isopropyl that is only 50% alcohol. To add confusion to the matter drugstores often label the percent alcohol in terms of "proof." Proof is a simple doubling of the percentage. Therefore 100 proof is 50% alcohol and 190 proof is 95% alcohol. To dilute from 100% alcohol to 70%, choose a convenient sized container, such as a pint bottle, then fill it ~70% full with alcohol and the rest with tap water. This measurement doesn't need to be exact.

Miriam Richards from Brock University has found that specimens stored and processed as above retain high quality DNA for at least several years.

The process for washing bees after they have been in alcohol is illustrated later in this document. The difference between a good bee collector/researcher and a poor one can be told by how well they wash and dry their bees, so don't skip this step!

Bob Minckley has found that when he does collections from individual bowls it is useful to use clear plastic fishing lure boxes. The compartments can be numbered and individual bees picked out of bowls by hand and placed in the appropriate compartment. Afterwards, he freezes the entire container for at least 10 minutes to keep anything from re-awakening and then pins them straight from the box. Sometimes these specimens are more matted than ones that have been properly washed, but most of the time they area readily identifiable to species.

Another alternative to whirpaks is to dump the catch into small numbered squares of cloth which are then rolled up and rubber banded together. Once back from the field these are collected, put into Ziploc bags and frozen until ready for pinning.

In all cases each bag, fishing box, or cloth should have a tag listing the sample location and date written on paper with pencil. Do not trust any kind of writing to stay on the outside of a Whirlpak bag as they inevitably get wet with alcohol or water and run.

The following figure was clipped from the EPA's Volunteer Monitoring Newsletter and should be handy for removing individual bees from individual bowls.

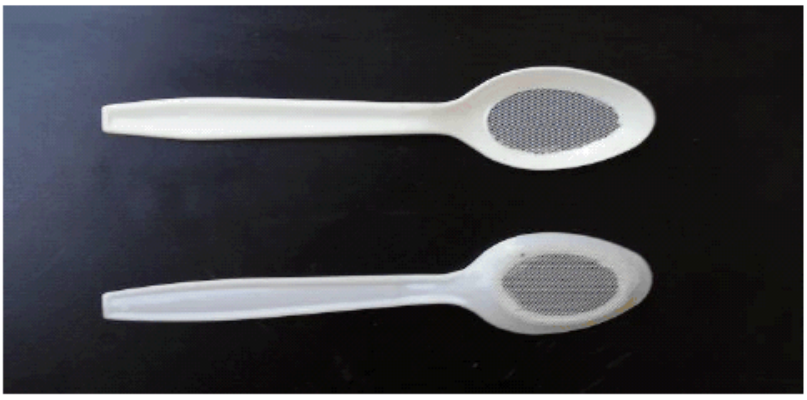
Net Spoon

by David Wilson

Sorting benthic macroinvertebrates from the tray with forceps is a pain, and often results in rather badly mauled specimens. With a net spoon, it's much easier to catch the critters and they are virtually never damaged.

To make a net spoon, use an electric grinder to grind off the bottom of a plastic picnic spoon, leaving only the rim. Be sure to wear safety glasses while grinding. Use waterproof cement to glue a small piece of fine mesh netting to the back of the rim. After the cement has dried, trim excess netting with small sharp scissors.

DAVID WILSON



David Wilson is a volunteer monitor with the Huron River Watershed Council in Ann Arbor, Michigan.

A Few Little Efficiency Tips - When getting ready for a day of setting out bowls we have found that it is helpful to create your sets of bowls the day before rather than have to do them as you set them out. In particular, it is very handy to have an empty divided flat like those found holding bedding plant starts at your local nursery as this holds the separate sets of bowls quite nicely. Wire flags (very useful for refinding your transects when driving at 60 mph) can be set in the passenger foot well. If working in a 4-door car we have found it fastest to keep the jug of soapy water on the back seat or on the floor of the back seat behind the driver. When the driver gets out they can grab a set of bowls and a flag in their right hand, open the door with their left hand, leap out of the car, pivot and grab the jug through the back window and then sprint off to put out bowls. By GPSing your transects as you put out bowls you can use the GOTO feature of your GPS unit to track back to your transect locations that evening or the next day. This is particularly useful when working in an area with few landmarks.

We have learned the hard way that getting into and out of the car many times a day while putting out bee bowls can be hard on the human body. In particular it is hard on the left leg as it levers you into and out of the car and that action can lead to some slow healing muscle strains. The best way to get in is to sit down on the seat first and then swing both legs over. Getting out is the reverse operation swinging both legs out and then standing up.

New Pan Trap Design for Arid, Low Humid Regions - As part of a native pollinator inventory, I have been looking for a reliable pan traps method. I initially used two-ounce Solo cups with soapy water at five sites along an elevational gradient north of Flagstaff, Arizona. In order to increase the opportunity to sample a higher diversity of species, I decided to leave the traps out for a week. I substituted recreational vehicle antifreeze grade propylene glycol in 12 ounce plastic bowls since it was obvious soapy water would not last long enough. Unfortunately, summer temperatures and low humidity caused the bowls to dry up and blow away before a week had expired (I can make someone a killer deal on a couple thousand 12 ounce plastic bowls).

I changed from 12 ounce bowls to 12 ounce heavy plastic “stadium cups”. Each cup, painted either fluorescent blue or yellow, or left white is attached to one-half inch PVC pipe with a five- inch hoop cut from a plastic culvert pipe (see photo). The stadium cups are very sturdy and are likely to hold up for a long season or two (or three) of sampling. The hoop is attached to the PVC pipe with a bolt and lock nut. The pipe slips over a piece of rebar set into the ground. The hoop is set high enough so the cup rests about three inches above the ground. If desired, a trap number can be written on the white plastic hoop. In order not to confuse the issue by having insects attracted to the white PVC end up in the blue or yellow cup, I painted the tops of holders the appropriate color for the cup it would hold. The top of the pipe is plugged to prevent bees from crawling down inside and getting trapped. The cups, filled halfway with the 50% industrial grade propylene glycol (50 Water:50 propylene glycol) easily lasts for a week. The cups do not sit on the hot ground (air passes under them) and deeper cup lip and deeper fluid level slows down evaporation. I remove the cup from the hoop and dump the sample into a sieve. Leftover propylene glycol is collected in a bucket when the sample is poured through the sieve. The sieve is dumped into a plastic jar with alcohol. I use a funnel used when you add oil to your car (the opening is large enough not to get clogged with the sample, I get a lot of big Tachinids) to dump the sample finally into a labeled Whirl-Pak. Funnels with wide opening can be found at Auto Parts Stores. I also find that collecting samples goes much faster if the labels are pre-cut and placed in the Whirl-Pak beforehand.

Field Trip Checklist

- Bowls
- Plastic Spoon
- Brine Shrimp Net
- Dawn Dishwashing Liquid
- Alcohol
- Whirl Paks
- Ziplock Bags
- Gallon Jugs
- 5 gallon Water Jug
- Aerial Net
- Replacement Nets
- Killing Jars
- Ethyl acetate
- Eyedropper
- Replacement Net Bag
- Location Log
- Blank Paper
- Sharpie
- Pencils
- Clipboard
- Maps
- GPS Unit
- Batteries
- Charger
- Scissors
- Tweezers
- Det Labels
- Paper Triangles
- Humidors
- Hand lens
- Reading Glasses
- Two-Way Radios
- Sun glasses
- Hat
- Toilet Paper
- Matches
- Cell Phone
- Collecting Permits
- Plant ID Material
- Technical Pens
- Enamel Sorting Pan
- Hair Dryer
- Pinning Board
- Bee Washer Jar
- Reading Glasses
- Empty Bee Boxes
- Pins
- Glue
- Boots
- Sun Screen
- DEET
- Drinking Water Bottle
- Backpack
- Hip Pack
- Camera
- Collecting Vials
- Watch

Bee Monitoring Discussion List and Announcements - If you are interested in bee monitoring or identification issues you might want to sign up for the bee monitoring listserv. It is a good way to alert you to interesting developments.

Email Sam Droege (sdroege@usgs.gov) to sign up.

Archives can be read at:
http://tech.groups.yahoo.com/group/beemonitoring/

Quick Bee Survey Protocol

What follows is the USGS Native Bee Inventory and Monitoring Lab's standard protocol for an individual site.

Setting Out Bowls

1. Place bowls level on the ground.
2. Put one heavy squirt of dish washing liquid in 1 gallon jug of water (Blue Dawn is the standard, others are fine as long as they are NOT citrus-based or scented). Any soap will do in a pinch
3. Fill each bowl with soapy water about ¾ or more full

4. Bowls can be left out for the middle part of the day or for 24 hours
5. Set bowls out in transects with 30 bowls spaced 5 meters apart (pacing is fine) alternating blue, yellow, and white
6. Avoid putting bowls in any heavy shade as few to no bees will come to those bowls
7. There don't have to be flowers nearby to have bees come to bowls as often there are bees scouting over flowerless areas and these individuals are highly attracted to bowls

Straining Bowls

1. Strain insects from bowls by dumping water from bowls through the brine shrimp net
2. After all bowls are strained, scoop out specimens with a spoon or your fingers, put insects in Whirlpak; fill enough to cover with alcohol. Any type of alcohol will do in a pinch. I usually pick up a small bottle at the pharmacy…it should be 70% or better. Best kind is ethanol but isopropyl will also work. Hardware store alcohol should be considered 95% alcohol, dilute to 70%
3. Add in with Whirlpak a contents label written IN PENCIL on a scrap of paper saying who collected, DATE (with month spelled out), location. It would be useful to show where you collected on a map, but not absolutely critical.
4. Remove the air from the whirlpak with your fingers, then roll the top down to the level of the alcohol, bend the ends forward and twist the wires together. Tuck the ends of the wires in to the center of the bag so they don't poke other bags.
5. Write down the time and location on another piece of paper so there is a log of what you have done.

Airplane Travel and Shipping Alcohol Specimens

When traveling with or shipping Whirlpaks of specimens you should partially drain the alcohol out of the bags to diminish the possibility of leaking while in transit without affecting their preservation. Be sure to properly fold and tie the Whirlpaks as outlined in the section above. Put all the Whirlpacks into a Ziplock bag and then into another larger Ziplock bag to make sure nothing leaks. Some paper towels in the outer bag will be added insurance.

Processing Bees that Have Been Stored in Alcohol - Pinning bees directly from water or alcohol usually results in matted hairs, altered colors, along with a good coating of pollen, scales, and other detritus picked up from the sample. We have found that washing and processing bees using the process listed below will result in well groomed specimens that can exceed the quality found when hand-collected.

We use one of two main approaches to wash bees, using either a strainer or a bee washer to accomplish the task, both are explained below.

Strainer Washing

- Fill your specimen Whirlpak with water and then dump contents into the strainer (tea strainers work well because of their fine mesh, brine shrimp nets also have sufficiently small mesh, but it is more difficult to remove specimens because of the flexibility of the netting)
- Either dump the specimens into a beaker or other container containing a SMALL amount of warm water and dish washing liquid or other kinds of liquid soaps and **very** vigorously swirl the specimens around with a stick or, better, put a lid on the top and shake them for 30-60 seconds.
- Place specimens back into the strainer and rinse under tap water until no more suds are present. Use your hand to break the force of the water to protect the specimens.
- Rap off loose water and use a towel to blot out as much excess water on the bottom of the strainer as possible. A cloth towel is more environmentally friendly than using a lot of paper towels.
- Either squirt 95%+ alcohol onto the specimens or drop them into a jar of alcohol and blot

again.

- Dump the specimens onto a paper towel and roll them around with your finger, pencil, or tweezers to remove the bulk of the alcohol.
- Move to a second paper towel and fold towel over specimens and blot. Often using a cloth towel or set of paper towels on top helps. Move specimens around on paper towel and repeat until no more alcohol wets the towel. Quite a bit of pressure can be used when rubbing down the bees; they are tough.
- Fold corners of the paper towel up and shake the specimens around inside to further dry them. Stop shaking once their wings are no longer stuck together or folded up on themselves and all bee hair is nice and fluffy. Note that you will likely have to hold the corners AND the towel area between the corners in your fingers or the specimens will jump out while you are shaking them.
- Note that after the specimens have been dipped in alcohol you can leave them lying on the paper towel for a bit (up to 45 minutes or so) before further fluffing if you aren't in a hurry.
- Pin as normal
- Note that the paper towels can be reused many times.

Washing Using a Magnetic Stirrer – Rather than cleaning bees by swirling them around in a jar by hand, we now use a magnetic stirrer the same as used in all chemical labs. A small magnet is turned inside a jar or cup by a magnetic plate. The water, soap, and bees are swirled around as gently or quickly as you wish and it does the best job of removing all the pollen, nectar, and gunk on specimens, simply because you can leave it washing for quite a while without a time penalty. Costs for new stirrers are about $100.00 and much cheaper on Ebay.

See video at: http://www.youtube.com/watch?v=A2y-ind12Cc

Bee Washer and Dryer - We have found that you can obtain beautifully coiffed hair on even the longest-haired of bumblebees if you spend the time shaking them around in a paper towel. Unfortunately, that can take a while and most people shake them only until their wings unfold and then pin them, leaving the specimen less than presentable. We then have to ID bedraggled specimens which, in the worst cases, can lead to errors in identification and always leads to a lessening of the aesthetic experience.

That need not be as you can use a hair dryer and the system below to speed things up.

You will need the following:

- A small clear glass pint or half pint jar (a quart will do) that has a canning jar lid of the kind with a removable central metal disk.
- A section of window screen. We use the fiberglass type, but metal might be ok, though they could be too stiff or may unravel. Note that you can buy loose fiberglass screen from the hardware store and cut it with scissors.
- A hair dryer.

Procedure:

- Fill the Whirlpak or vial holding bees with water and dump into the container.
- Even though you may have caught them in soapy water bees and other insects need to be washed again to get all the pollen and other junk out of their hair and get them squeaky clean.
- Screw on jar lid and screen.
- Drain out the liquid.
- Rinse once with plain water.
- Add dishwashing liquid or other kind of strong soap or degreaser.
- Fill ¼ full with warm water.
- Cover the top with the old metal disk or your hand and shake vigorously for 60 seconds! Don't be afraid, you can't shake too hard. Note if you don't wash them well their hair will still stick together.

- Rinse until there is no obvious soap in the water.
- Beat out as much residual water by inverting the jar and rapping it against a folded up hand towel.
- Dry the outside of the container, put it in one hand, and then whirl out as much internal water as possible to increase the speed of drying by removing as much free water as possible. It is even better if you the jar in a cloth towel to do this. Remove the lid and dry the rim and screen and put back on.
- Turn on hair dryer to dry. We use high heat though some people don't even turn on the heat, particularly if they rinsed their specimens in quick evaporating alcohol.
- Place the jar on its side on the folded hand towel and place hair dryer pointing into the jar as close as possible without causing the hair dryer to cut out (usually about 1 inch).
- Apparently, as we have found, if you put many hair dryers right up to the screen they will overheat and turn themselves off (stick them in the freezer if you want them to come back on quickly).
- While drying, shake the specimens back and forth vigorously, hitting the sides on the towel periodically to dislodge them if they stick to the glass.
- Specimens, when wet, are very flexible and tough, so they can take a moderate amount of bumping around.
- Once the specimens are all loose, shift the jar slightly downward so that the specimens slide towards the screen and whirl around in the dryer's wind, continue shaking the specimens.
- Small short-haired specimens are done once their wings are flexed away from their body and their hairs are not matted. Bumblebees and long-haired specimens take longer. Depending upon your hair dryer and your technique this may take anywhere from 1.5 minutes to 4.

Using Compressed Air – We have found that using compressed air results in the quickest drying of wet bees. When using compressed air be aware that there can be moisture in the air lines so run the air wide open for a few seconds to get rid of any loose moisture. Also be aware that at high pressure compressed air can blow apart specimens, particularly their abdomens. We initially hold the bee washer about 2 feet away and let the air dry out the free water at full blast. Once the bees start to move around in the jar, cut the pressure down and move the jar right up to the nozzle. Direct the air stream to the side of the jar and let it swirl the specimens around in a vortex (if the pressure is too high or they are bouncing violently around you can rip some abdomens off). Small specimens with short hair take less than 1 minute. Bumblebees take about 2 minutes to have all the hair on their thorax fluff up.

Salad Spinner - We have used the salad spinner operation a number of times with fair success, but email us your experiences if you try this technique, so we get a better understanding of how it works for others.

- Wash specimens thoroughly as mentioned above.
- You will have better results if you do a last rinse in alcohol.
- Line the inside of the salad spinner basket with paper towels.
- Place drained bees in basket.
- Spin like mad. (It is unclear how long you should spin, so try it several ways)
- Move specimens at least once so they are in contact with dry paper towels.
- Large batches of specimens may require changing the paper towels if they become too wet.
- As usual look for wings that spread out nicely and hair that is nice and fluffy.

Making and Using an Autobeedryer - If you are really involved in collecting and processing many specimens then you will want to invest in the creation of an autobeedryer. We have a slideshow and video up on the web on how to make such a device at:

http://www.slideshare.net/sdroege/how-to-create-an-autobeedryer

http://www.youtube.com/watch?v=935jlJep6go

Cleaning Bees that Have Gotten Moldy - Leif Richardson has put together a method of removing most of the mold on bee specimens that have gotten moldy due to storage in high humidity conditions. He writes: "First, I cut a piece of foam board (like the foam you find in a standard insect box; I got mine from Bioquip) to fit snugly in a small plastic food storage container. I wedged this into the bottom of the container, stuck pinned specimens (labels removed) into the foam, and added warm, soapy water to submerse the bees. With the top on I gently shook the container for about five minutes, then drained it and repeated. I next filled the container with 70% ethanol and shook for five minutes. I used two additional alcohol rinses, then removed the foam board from the container and used a hair dryer to dry and fluff the bees.

The bees emerged from this treatment with most of their body parts intact. Some pollen was removed from scopas. Most of the fungus was removed, but some still clung to hairy places and the tight spaces between body segments. I think you could use a soft children's watercolor paintbrush to jab away more of the fungus during one or more of the rinses. One caveat: the foam board has a tendency to break free and float, causing the specimens to get pressed up against the top of the container. I think this could easily be avoided with the right container, foam, glue, etc. Finally, the dimensions of the container will determine how many bees you can clean at one time and how much alcohol you will have to use."

Re-hydrating Bees that have Been Pinned – At times there is a need to re-hydrate bee specimens in order to remove them from the pin or to pull the tongue or genitalia (note that pulling open the jaws on specimens is difficult after they have dried, even with extensive re-hydration). To do that people will place bee in a re-hydration chamber of some sort. Any container which can contain a small amount of water at the bottom and specimens at the top will do. It can take anywhere from a few hours to several days for larger specimens to relax. To prevent mold you can add a few drops of ethyl acetate, a few moth balls, or a large dose of alcohol in the water. Thanks to Jack Neff and Jason Gibbs for their contributions on this topic.

An Inexpensive, but Powerful LED Lightsource – We have been frustrated by the cost of high quality microscope lights. Even old fashioned illuminators now cost well over $200.00 and still deliver sub par light compared to that from fiber optic lights. We discovered that the Gerber LX3.0 LED miniflashlight works extremely well as a microscope light. It is quite small (7.5" X ~1" in diameter at

the head) but produces and impressive amount of light, exceeding all our illuminators and equaling the more inexpensive fiberoptics.

We found that the flashlight fit very nicely into the standard Bausch and Lomb microscope stand's illuminator hole; a little adjusting up and down and we had all the illumination we wanted.

As the batteries drain down the light will dim, so if you are in the field then it is useful to have spare sets of batteries charging while using the flashlight.

In our office and at home we have converted these flashlights to use household current.

To convert you will need a wall cube transformer of some kind that converts 120VAC to Direct Current. You can buy a wall cube at Radio Shack or you may have one around the house. Make sure that the wall cube converts AC 120V (input) to somewhere around 4.5V DC (note, make sure it is not 4.5V AC!!!). Other flashlights will use other voltages depending on the number and type of batteries they are using. I didn't look closely the first time I tried this and I ran the light on AC, which worked OK, but the light shimmied around and was distracting. According to the experts the LED bulbs are pretty tough so you could likely run them up to maybe 6V without shaving too much off their life.

German Perilla has created a wonderful power point how-to presentation about converting flashlights into microscope lights. That presentation is available on the web (http://envstudies.org/) but be aware that it only runs under Microsoft Internet Explorer not other browsers. The file is also available from Sam Droege (sdroege@usgs.gov) or if you are a member of the bee monitoring listserv (http://tech.groups.yahoo.com/group/beemonitoring/files/)

We recommend that you use the power point presentation to convert your flashlight as the instructions are illustrated and much more detailed and permanent, but here are the basics. Take out the batteries and run a wire down to the bottom of the flashlight. We used a dowel to which a tiny screw was attached to one end and then attach a wire to that screw and tightened it. Be careful to not let any of the wire touch the wall of the flashlight or it will create a short (the body of the flashlight is the negative lead). Then tape the wire to the dowel and run the whole thing to the bottom of the flashlight. For the return, I ground off some of the outer nonconductive anodized finish on the flashlight body, and simply taped the end of another wire to the body. I cut the end of the wall cube off and attached its wires to the wires coming off of the light. If your first try doesn't work then switch the wires as the polarity may be wrong (this is not supposed to be healthy for the LED, but mine survived). You can then put a switch in the line if you want, or simply plug and unplug the wall cube.

For technical information about other, similar flashlights check out the LED forum at:

http://candlepowerforums.com/vb/index.php?

How to Make a Pizza Insect Pinning Box

(Refer to Figure at End of this Section)

Written by Rob Walker and Sam Droege

Because of the volume of insects collected in our lab we have begun using Pizza Boxes as an inexpensive alternative to traditional field boxes.

Pros: Inexpensive, saves shelf space, holds more specimens

Cons: Materials have to be purchased separately and assembled, box not as sturdy as others, pest insects have greater potential access to specimens

Blank pizza boxes can be ordered online from many sources. Pizza shops may also be willing to

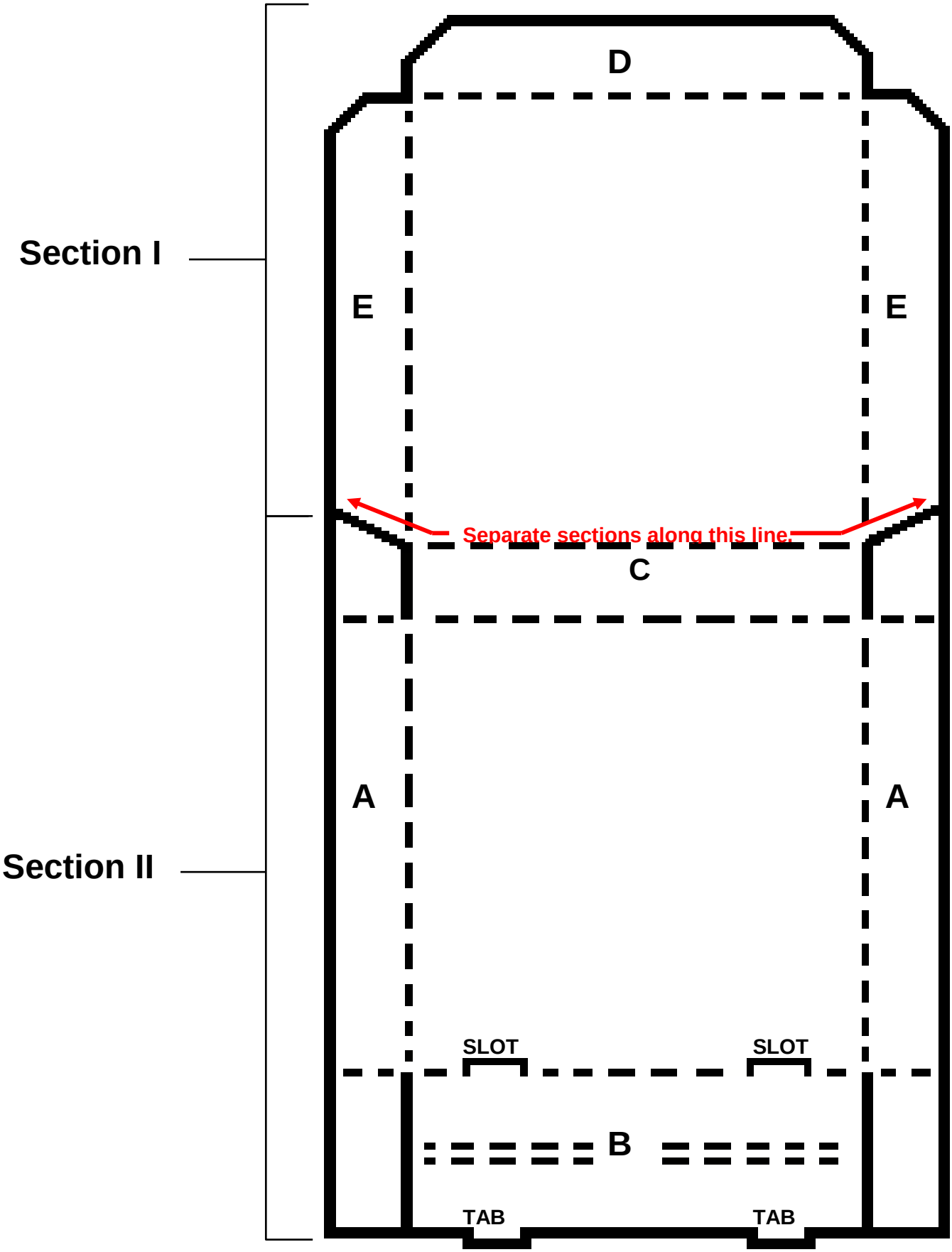
donate cartons. We use crosslinked polyethylene foam for our pinning base within the boxes as it seems to have superior pin holding properties to that of ethafoam, but either could be used. If you order in foam in bulk you will save a great deal by going directly to a manufacturer. We have had good luck with Reilly Foam 610-834-1900. We have them cut the foam to 3/8" in thickness and ship as 2'x4' sheets.

Assembly directions for a standard pizza box:

1. Use a knife, scissors, or paper cutter to cut and separate Section I from Section II, along red arrows as shown.
2. Take Section II and assemble by taking side flaps A and turning in end tips.
3. Fold flap B over end tips so that the tabs are securely in the slots provided.
4. At other end, fold end tips in and fold up flap C.
5. Staple flap C and the end tips together so that flap C stays upright. (Staple 4x's per end tip to secure them.)

1. With blade or paper cutter, remove flap D completely from Section I.
2. Fold up flaps E.

1. With blade, scissors or paper cutter, cut a square of foam large enough to fit snuggly along the box sides B and C. Leave room enough along the other two parallel sides (sides A) so that the Section I box top flaps (E) will slide in, keeping the lid edges from flipping into the specimens.
2. Hot glue the foam to the bottom of the box. We use low temperature glue guns, but have not tested higher temperature guns to see if they melt the foam. To make sure the glue does not dry before you finish applying, glue the central third of the foam first and affix it inside the box. Then lift the sides and glue. Be sure to place a glue line close to all the edges of the foam.


Section I
Section II
D
E
E
Separate sections along this line.
C
A
A
SLOT
SLOT
B
TAB
TAB

T. Mitchell's Guide to the Bees of the Eastern United States

While published in 1960 and 1962 Mitchell's 2 volume set on Bees in the Eastern United States is still a very valuable reference book and source for identification keys, illustrations, and species accounts. While now quite expensive to purchase via rare book dealers it is available as a series of pdf files for free at:

http://insectmuseum.org/easternBees.php

Note that Mitchell's taxonomy is out of date and that all identifications made with this book should be cross-referenced against the list of bees of North America available at www.discoverlife.org and within the bee identifications guides located at that same site and discussed in the next section. You can cross-reference names by either going to one of the genera guides directly or simply typing the name into the search bar on the home page of Discoverlife.

Mike Arduser's Keys

Mike Arduser has been putting together keys to several genera of bees from the Midwest. Those can be viewed at:

http://www.nbii.gov/portal/community/Communities/Ecological_Topics/Pollinators/Featured_Projects/Bee_Keys_by_Michael_Arduser/

A Guide to Identifying Bees Using the Discoverlife Bee Keys

This document provides guidance in the use of the online Discoverlife guides or keys. These instructions are designed for use with the guides for the genera and species of bees, however, these instructions will largely hold true for any of the non-bee guides also available at the site. Be sure to also see the section on the end regarding the use of already identified specimens. A set of identified specimens can be obtained at no charge from Sam Droege at the numbers above.

All of the Nature Guides are located at:

http://pick4.pick.uga.edu/mp/20q

However, the consolidated links to the bee guides and associated materials are at:

http://www.discoverlife.org/nh/tx/Insecta/Hymenoptera/Apoidea/

Discoverlife guides differ from traditional dichotomous keys in that characters that help differentiate species are evaluated and scored for all or almost all of the species. Think of it as a matrix, with species as rows and character states as columns. That matrix is employed by answering questions regarding the presence or absence of characters for a specimen. As questions are answered the list of possible species is narrowed until, in most cases, the list resolves to a single name.

Hint: If you are just beginning to learn how to identify bees we suggest that you download the glossary of terms, vocabulary, identification tips, and pronunciation materials that we have compiled at:

http://online.sfsu.edu/~beeplot

On the bee page at Discoverlife there are a series of guides listed for **Eastern North American** bees (states and provinces east of the Mississippi River). Over the coming years we will expand these guides to include the western states and provinces. Guides are listed as either implemented or draft. Implemented guides have at least the wording completed and have gone through several rounds of editing, checking, and testing. These guides will continue to evolve as new information comes in on species, names, characters, and the addition of pictures and drawings. Draft guides are potentially useful, but should be used with great caution as they are still being developed, edited, and validated.

Most guides deal with a single genus of bees, if there are a large number of species present these guides are often divided into two guides, one for each sex, as characters useful for identifying species are often gender specific.

Hint: If you are unfamiliar with the bee genera we suggest that you start your identification process by using the guide to bee genera to divide your collection into genera.

The instructions that follow apply equally to the guide to bee genera or to any of the individual bee genera guides.

Each guide has questions on the right, a species list on the left, and navigation tools across the top. The list of species and the list of questions interact with each other. Answering any question (**in any order**) narrows the list of candidate species. Similarly, one can flip the process and have the computer narrow the set of questions based on the species that remain on the list.

Clicking on any pictures present within the guide will display an enlarged or version of the picture. Many species names can also be clicked on to reveal species specific pictures and often have associated text material on the nature history or identification of that species.

Hint: Answer ANY NUMBER of questions IN ANY ORDER. You do not need to answer all questions. Initially answer ONLY questions where you are sure about your answer.

The **initial page** presents a subset of all the questions in the guide. These questions are both easiest to understand and most likely to separate out large numbers of species.

There is no need to answer the questions in the order presented.

At least initially, you will find that there are some questions that are clearer in your mind than others. These should be answered first.

Leave questions you are unsure of blank! Don't guess!

We recommend that you spend more time reading and learning about the morphological characters in the questions before providing your answer, or simply skipping the question.

Not all characters will have been scored for all species. If both sexes are present in a guide then characters that only apply to one sex will obviously not be scored for the other sex. Similarly, if we have been unable to obtain a specimen of a rare species we may not be able to score some characteristics from the available literature. The consequence of this is that any species that has not been scored for a particular question will remain on the list of possible candidate species regardless of whether it actually has that character or not, simply because it cannot be eliminated from the list of possibilities.

Hint: While using a guide, there are 2 types of species that remain on the list. 1. Those species that have the characters you have indicated. 2. Those species that have not been scored for some or all of the characters you chose in your answer. The second type of species will stay in the list simply because we do not have enough information about its characters to eliminate it.

Hint: For many characters you are given three or more choices of states. If you are not sure which of the states your specimen's character fits into don't hesitate to click on all possible correct combinations rather than trying to narrow it to the one that best fits.

At any point you can press any of the SEARCH buttons that are located throughout the page. Doing so will update the species list on the left based on the charaters you have chosen.

At any point you can also click on the SIMPLIFY button that appears in the left hand column above the species list. Doing so eliminates both questions and states within questions that do not help resolve the identity of the species remaining on the list. Clicking this button also adds those appropriate questions that were not included in the initial list of questions present when the guide was first opened. Additionally, hitting the SIMPLIFY key will also reorder the questions alphabetically.

Both the SEARCH and SIMPLIFY buttons can be clicked as often as you wish. We usually click on the SEARCH button after answering a question, just to get a sense of the questions that best help eliminate species the quickest and to make sure that we haven't made some fatal error. We suggest waiting to click on the SIMPLIFY button until you have a reasonably small list of species left or have answered most of the questions you are comfortable with on the first page. If you hit the SIMPLIFY button earlier in the process it will bring up a potentially very large list of additional questions, that may not be as useful or as easy to use as the initial ones.

***Strategy* -** Especially when you are unfamiliar with the species within a genus it is very useful to take some extra time to double check your initial identification. In many cases there will be pictures and extra information stored as a link to the species name. Those can be compared to your specimen (be aware that males and females often look quite different from one another).

The next step to verifying your species ID is to compare your specimen to the complete list of the scored characteristics of that species. To get a list of those characteristics click on the MENU link at the very top of the page, then at the top of the left hand column click on the CHARACTERS option and then the species you wish to review, then hit the SUBMIT button to get a list of scored

characteristics.

One nice feature of the Discoverlife guides is that there are many paths to the final answer of correct species identification. This feature can be exploited when checking your identifications. By hitting the SIMPLIFY button at the very beginning you will display ALL the questions for the guides. By answering a different set of initial questions a different species will remain on the list. These new questions and species may expose some flaw in your initial identification and this will become obvious if you don't return to the same species identification at the end.

Hint: These guides are easier to use than dichotomous keys. However, answering questions incorrectly will still yield WRONG IDENTIFICATIONS, so be careful and conservative in your answering.

The RESTART link, located at the top of the page in the header, restarts the guide at the beginning.

Advanced Uses of These Guides - By pressing the MENU link at the top of the page the simple species list found normally in the left hand column is replaced with a set of new options used by individuals building or editing guides. Some of these features are also useful when exploring the identity of a species. Don't worry about exploring any of the features found in the MENU page as only the guide developers have permission to make permanent changes.

The CHARACTERS option will give you the scored characters for any of the species you have checked.

The DIFFERENCES option will give you the differences in scoring among any 2 or more species you click.

Clicking the HAS key restarts the guide but brings up ALL the characters for that guide in alphabetical order. Additionally, a new set of 2-3 buttons has been added at the top of each characters section; the NOT, ONLY, and HAS buttons (sometimes the NOT button may be turned off). If you don't click any of these 3 buttons the guide acts as it normally does. If, however, you click on the HAS button along with one of the character states…. hitting the SEARCH button will generate a list of species on the left that will include only those species that have been scored as having that character. What will be missing are those species that were never scored for that character at all. Similarly the ONLY button provides a list of species that have been scored for that character alone. This means that if a species was scored as possibly having all or more than one of the possible states, it will not be displayed if the ONLY button was clicked. The NOT button provides a list of species have not been scored for the selected character state(s).

The Discoverlife website also has a HELP link, which takes you to even more details on some of the more advanced features.

If you have questions about any of the bee guides please contact Sam Droege at sdroege@usgs.gov or 301.497.5840. My lab is open to anyone who would like to come learn to process and identify their collection of bees. Most of the time we have space, computers, and microscopes available as well as access to our synoptic collection.

Final Hint: If you find any errors or can think of a better way to do anything with these guides please contact Sam.

Using Previously Identified Specimens as an Aid in Learning Your Bees - When first starting out you will learn how to identify bees far more quickly if you use pre-identified specimens than if you try to immediately key out the bees you have collected. Because you already know the identity of the specimen, you can track your progress and reflect on your errors while using the guide and the mind/eye/guide learning loop will take place more quickly. If you use unidentified specimens you may find it difficult to initially feel 100% confident that your id was correct.

There are two ways to approach the situation. One is to use the guides directly. After selecting each state of each character you believe your specimen expresses from the selections available on the computer screen click the search button. You can then watch the list of matching specimens on the left side of the screen to see if your species or genus remains on the list. If it does not then you know which state of which character you entered that lead to the incorrect match.

Alternatively, you can go to the menu section of the guide and call up the entire list of scored states/characters of the species or genus you have on hand. Once you are in the menu section, you click the radio button next to "score," then click the box next to the species you want to investigate, and finally click the submit button. All the information for that species will appear onscreen and you can compare every scored character in the guide to the characters you see on your specimen, thus familiarizing yourself will all the characters in the guide. You will also find that you can "see" certain characters easily and others may remain difficult for you to interpret or find, thus helping you decide which characters you will preferentially use when keying out that group.

Feel free to contact Sam Droege for a set of identified specimens to use.

Acknowledgements: Many thanks to Liz Sellers for the many helpful edits to this section.

Stylopized Bees – As you identify bees you will, at times, come across bees that have an infestation of mites and more rarely bees that have been parasitized by a Strepsipteran (i.e., stylopized). Strepsiptera is a mysterious order of unclear position within the holometabolous insects. They are endoparasites of various other insect orders including a diverse array of Hymenoptera. Families Andrenidae, Halictidae, and Colletidae are the most frequently parasitized bees.

One can find male puparia (MP), empty male puparia (EMP) and adult females (F) in bees. MP are usually very large spherical extrusions, however findings of these are quite rare. More frequently you can find EMP, these are sometimes hidden and difficult to recognize. In some cases, EMP appears as an obvious deformation. F cephalothoraces are most commonly encountered in bees and appear as small orange/brown plate-like extrusions that emerge from beneath the rim of the tergites of the abdomen (see figure below). Upon seeing one you will have the impression of a small head peaking out from beneath the rim. Sometimes the apical rim of the tergite covers most of the parasites body (in most Halictidae) and will appear almost invisible from the dorsal view. However, the rim of the tergite is usually lifted upwards and the Strepsipteran can be viewed when looking under the rim.

Strepsiptera can modify not just the morphological features of the site where they are attached but the morphological characteristics of the entire bee, including the sexual characters of bees. At times the characteristics of the bee are changed enough to partially disguise the species identity of the specimen. Deformations occur among all bee hosts, but they are quite rare. Sexual character changes are manipulated by the parasites and occur only in some groups - most bees of the family Andrenidae and some Hylaeus (Colletidae).

Jakub Straka, a researcher from the Czech Republic, is working on the taxonomic and ecological facets of Strepsiptera. He is very interested in collecting host records for this group, parasitism rates, and specimens for DNA analysis. If you come across any stylopized specimens in your collecting activities please contact Jakub (straka-jakub@vol.cz). This group occurs uncommonly so even single records are of great interest.

Figure – Stylopized *Andrena clarkella* – It's the pale rounded thing poking out of tergite 3.

Taking Pictures of Bees with the COOLPIX 990

Natalie Allen and Stephanie Kolski - March 2005

This document covers photography of bees using the COOLPIX 990 digital camera. These cameras are now readily available and inexpensively on E-BAY and other used equipment sites. Their recent model replacements should also work reasonably well too, although they were not evaluated. The camera settings and specimen set-ups were created using a series of 990 web sites for nature photography, discussions with Dan Kjar on lighting at Georgetown University, and plenty of trial and error. With these set-ups you can get remarkably good photographs of your bees for very little money.

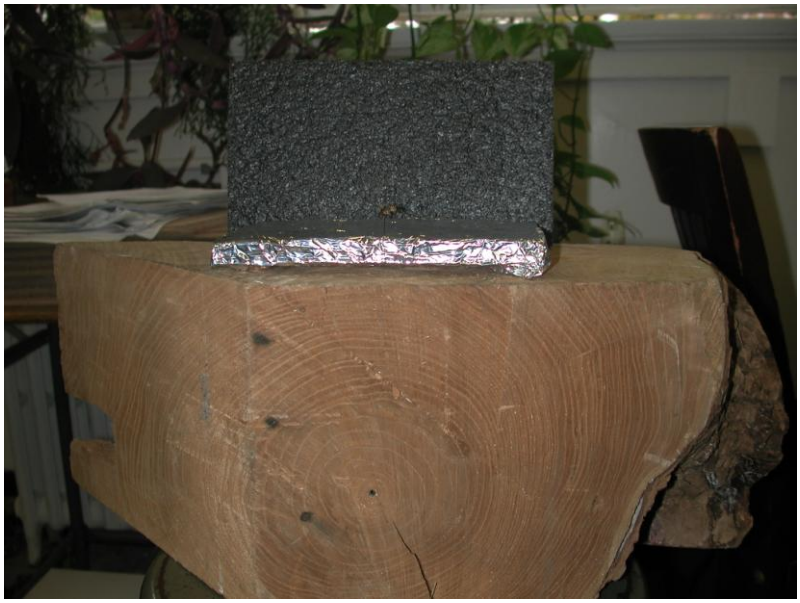
For examples of photographs take with the outside set-up see the pictures found on the Coelioxys keys at: http://pick4.pick.uga.edu/mp/20q. Click on the species name to see these pictures, the lists appears when you hit the identify or checklist buttons…..the first species, *alternata*, has some nice examples.

For examples of photographs taken under the microscope see the guides for Agapostemon. Click on the photos of the character states to enlarge them and see the listing for the authorship. Note that on this site there is a mix of pictures taken by John Pascarella and ourselves. As we progress in our picture taking more and more of the guides will be illustrated using these techniques.

Photographing Bees without a Microscope - Photography should take place outside on clear and sunny days around midday. The sun should be at a ninety degree angle to the photographer and the camera. For best lighting we use mirrors to reflect additional light onto the specimen. The person holding the mirrors should be at a 180 degree angle to the sun.

Camera Settings - The camera should be turned on to MANUAL. The ISO equivalency should be set to 100. Leave the light balance on AUTOMATIC. The metering method should be set to SPOT. The focus mode should be set at MACRO CLOSE-UP. Quality should be placed on FINE.

Display Set-up and Procedure for Side and Frontal Shots - Take a piece of foam board (approximately four by six inches) and cover it with a piece of aluminum foil that has been crumpled and then reflattened. (The aluminum foil is used to reflect the sunlight onto the underside of the bee.) The foam board should lie flat on a raised surface with the aluminum foil facing upward. A black foam board about the same size as the base foam board should be placed vertically so that the two six-inch sides touch one another to form a ninety degree angle. Place the specimen approximately one inch in from the outside edge of the six-inch long side of the aluminum-covered foam board, or about three inches from where the foam board meets the black board.



Place the camera on the raised surface directly up against the foam board so that the camera is even with the specimen. For side shots, the specimen should be placed with the body parallel to the camera lens. For frontal shots, the head of the specimen should be pointed directly at the camera

lens. The camera and the specimen should be one or two inches from one another so that when looking through the monitor, the specimen appears in the center of the screen. Move the camera closer or farther away from the specimen to make the image correctly sized and in focus. (Although it is possible to move the specimen instead of the camera, it is typically easier to move the camera when dealing with small distances such as these.) The ZOOM button can also be used to achieve greater clarity and to adjust size. While one person photographs the bee, the other should use two mirrors to reflect additional light onto the specimen. However, it is generally easier to have the bee in focus before the mirrors are added, as the mirrors create additional glare for the person looking at the monitor, making it more difficult to view the bee, and thus to focus. Furthermore, to reduce the glare from the sun on the camera monitor, a blanket may be placed over top of the camera and the photographer's head. Care should be taken to keep all shadows from the camera, the photographers, the blankets, and the mirrors off of the specimen. One mirror should be placed at a 180 degree angle to the sun and specimen. An additional mirror should be placed between the first mirror and the photographer. Adjust this mirror until the rays of the sun are directly reflected onto the specimen.

Display Set-up and Procedure for Top Shots - For top-view shots, take the black foam board used as the background in the previous set-up and lay it flat on the raised surface. The specimen should be placed in the center of the foam board.

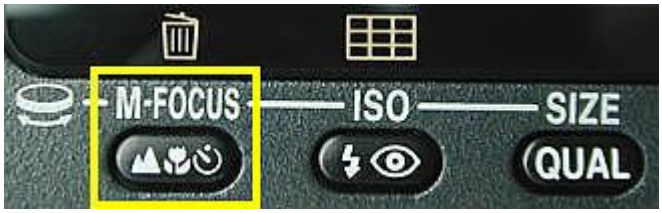
Looking down on the bee, position the camera one or two inches away from the bee. Focus in on the bee, paying particular attention to the head, using the ZOOM button for greater clarity. While one person photographs the bee, the other should use two mirrors to reflect additional light onto the specimen. Again, it is typically easier to get the specimen into focus before the mirrors are added. However, because of positioning, a blanket cannot be placed over the photographer's head to reduce the sun's glare on the monitor. Take care to keep all shadows from the camera, the photographers, and the mirrors off of the specimen. One mirror should be placed at a 180 degree angle to the sun and specimen. An additional mirror should be placed between the first mirror and the side of the foam opposite the photographer. Adjust this mirror until the rays of the sun are directly reflected onto the top of the specimen.

Taking pictures with the microscope -

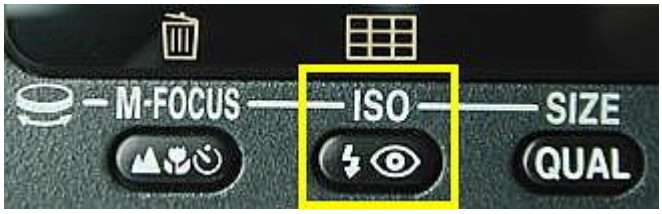
Follow the directions found on the website http://www.cdfa.ca.gov/phpps/ppd/Entomology/Diptera/digphot.htm for directions on how to assemble

a collar for your Coolpix 990 that will fit on one of the oculars of the microscope. The collar should fit somewhat snuggly on the eyepiece of you microscope while remaining easy to remove. If the fit is too loose, wrap several layers of masking tape around the eyepiece to provide increase the diameter. The pictures that follow came from the above web site.

Microscope Camera Settings - Turn the camera on to manual focus. Under the viewing screen, press the “M-FOCUS” button until an image of a flower appears in the top right corner of the monitor (macro setting).



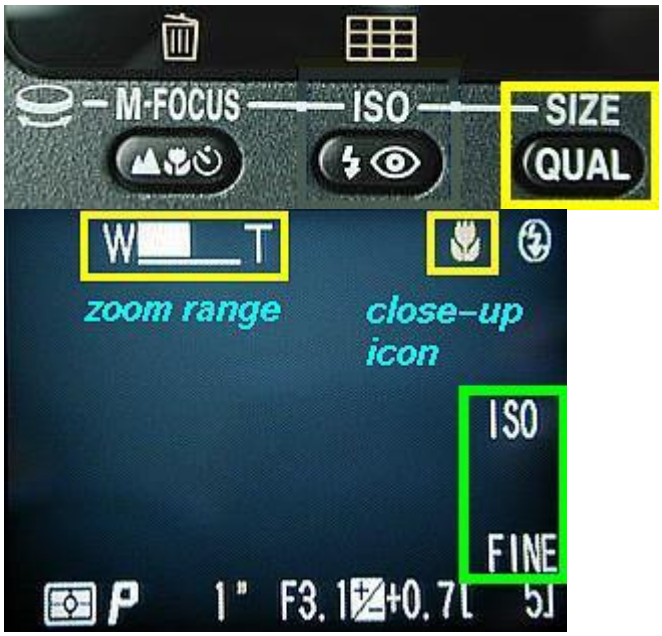
Next, press the button to the right of the previously used one (ISO).



Turn the flash off. There will be an icon of the flash symbol with a line through it in the top right corner of the monitor indicating the flash is off.

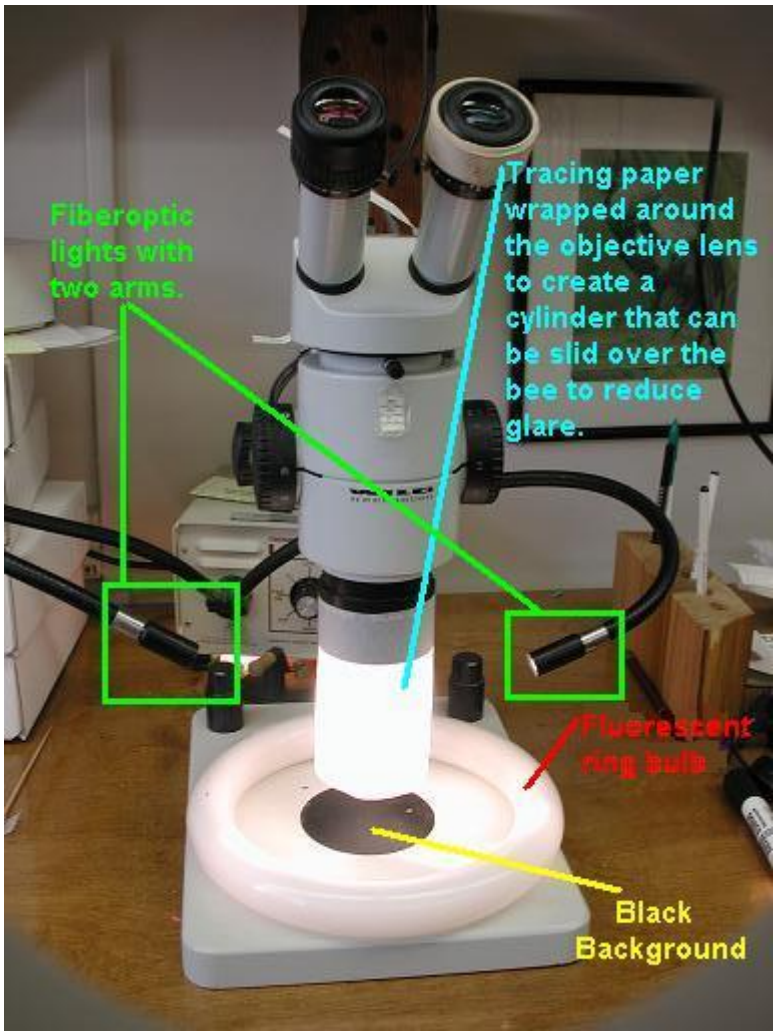
Press the ISO button again, and, while the button is being held down, turn the command dial until the number 100 appears on the right side of the monitor.

Press the next button to the right to change the setting to “FINE” (see green box).



Next push the menu button above the monitor. Open the option “metering” and change the setting to “spot.” Once again in the menu section, go down to “focus options.” Continue to “AF Area Mode” and change the setting to “manual.” Press the menu button to get back to the monitor. On the top of the camera, hold down the “mode” button and turn the command dial until an “A” appears in the bottom left corner of the screen.

Taking the pictures - Lighting should include a two-armed Fiberoptic light source and a fluorescent ring bulb with an 8-inch diameter that has been taken from a household lamp. The fluorescent ring bulb should be placed directly on the base of the microscope so that the bee can rest inside of it (see red writing in the below picture). The Fiberoptic lights should be turned on high, and the arms swung so that each light is shining on the bee from the opposite direction at a slight angle (see green writing in the below picture). The arms may be adjusted as necessary to provide the correct amount of lighting and to reduce shadow and glare in certain areas. Tracing paper should be loosely taped around the objective lens to form a cylinder that can be slipped up and down around the bee to further reduce glare (see blue writing in the below picture). The bee should be placed in modeling black clay (hint: schools are a good free source for small amounts of modeling clay), which should in turn be placed on a black background.



Once the bee is put into place, focus in on the bee at the desired magnification. Slide the tracing paper down until it hits the base of the microscope and completely encompasses the bee, taking care not to disturb the bee. Fit the camera snuggly onto the eyepiece and zoom in and out with the camera's zoom feature until the bee is in focus. If the bee is not in the exact center of the camera, use the arrows to the left of the monitor to select the set of focusing brackets that are closest to the area where you want to focus. The selected box will be red while the rest will be white (see below picture).

Take your shot. Often it is better to use the zoom feature of the camera to reach your final magnification. In that way you maximize your depth of field and the image will come out square instead of round.

Additional useful websites on taking digital photographs through a microscope objective:

http://www.inspect-ny.com/digipix.htm

http://www.funsci.com/fun3_en/upic/upic.htm

Affixing bee wings to microscope slides

Contributed by Tulay Yilmaz and B. Gokce Ayan

Materials:

Entellan fixative or mounting media
Slide
Tweezers
Brush, or glass rod (the glass rod is easier to clean afterwards)
Petri dishes with warm water
Microscope
Pin
Desk lamp with an incandescent bulb (not a fluorescent one)
Something to put the wings on while the wings dry under the light's heat

1. Place the slide on a white piece of paper for easy visibility
2. Put some warm water into the Petri dish
3. Turn on the lamp and leave until it's hot
4. Take the bee's wing with the tweezers
5. Place the wing into the warm water, wait for a while to get it as smooth as possible
6. Remove the wing from the water and put it onto the drying surface (be sure wing stays flat)
7. Leave it under the light to dry
8. Remove when dried
9. Drip some Entellan onto the slide with that glass rod and spread it
10. Hold the wing with the tweezers and gently put it onto the surface of the Entellan (be careful about putting it on the right way)
11. Don't use a coverslip!
12. Then look at the preparation under the microscope
13. If you see any air bubble under the wing, press them out with the help of the head of a pin (not with the pointed part of it) and pop them (with the pointed part) once you manage to bring them out from under the wing
14. Now, leave it in a closed box so it's not affected by dirt and dust floating in the air. Entellan is really sticky and readily picks up dust when you leave the preaparation in the open air
15. Clean your glass rod immediately (because when Entellan hardens, it gets difficult to clean)
16. Preparations can be used when Entellan is dry (usually within an hour).

Such preparations are faster and more practical than other slide preparations we have used and the slides keep for a long time. We have found the slides to be usable one or two years later and they may last much longer.

Do your preparation in a well-ventilated area as the solvents in Entellan can give you a headache.

While preparing the wing don't breathe on the slide and be careful when you talk or laugh, because it can causes the wing to sink into the Entallan or disappear.

North American (North of Mexico) Introduced and Alien Bee Species

Information on distributions and status come from the literature, active North American collectors, online collection data available via the global mapper on www.discoverlife.org, and John Ascher's compilation of distributional data. Thanks for the contributions from Mike Arduser, John Ascher, Rob Jean, Jack Neff, Cory Sheffield, Robbin Thorp.

Updated: July 2009

Account Layout: I = purposely introduced, A = accidental introduction or possibly natural colonization (although this would be unlikely for most), Genus, Species, Decade of Establishment, Probable Source Population, Current Status in North America north of Mexico

Apidae

I Apis mellifera 1620. Europe, Mediterranean region. Feral colonies present throughout North America. Colony numbers and persistence recently have declined following the introduction of parasitic mites in the 1980s and 1990s.
I Anthophora plumipes 1980. Europe and southern China. Introduced at the USDA Beltsville, MD Honey Bee Laboratory. Numbers were initially low, but this species is now found commonly in early spring throughout the Washington D.C. metropolitan area where it nests in the ground under porches or in the dirt of uprooted trees and frequents planted azaleas and other garden flowers. Has the potential to spread throughout North America.
A Ceratina cobaltina 1970. Mexico. While it is possible this is simply a disjunct Texas population, specimens for this distinctive Mexican species were only recently discovered in Travis and Hidalgo counties.
A Ceratina dallatoreana 1940. Mediterranean region. Central California.
I Ceratina smaragdula 1960. Pakistan, India, SE Asia. Introduced into California but not found since its introduction.
A Centris nitida 2000. Southwestern U.S., Texas, Mexico, Central America and Northwestern South America. Recently discovered in southern Florida. Not expected to spread outside of Florida.

A Euglossa viridissima 2000. Mexico and Central America. Recently discovered in southern Florida. Currently found only on the eastern side of the state. Expected to spread to the western side but not invade much further north.
A Xylocopa tabaniformis parkinsoniae Recent. South Texas. Recently appears to have left its historical haunts along the Rio Grande and now found commonly in urban areas of Central Texas, perhaps translocated there via firewood, but possibly colonized naturally.

Andrenidae

A Andrena wilkella 1900s. Europe and northern Asia. Common throughout the north central and northeastern U.S. and southern Canada.

Colletidae

A Hylaeus leptocephalus 1900. Europe. Found throughout the U.S. and southern Canada. Particularly associated with gardens, urban and disturbed sites. Often found on *Melilotus*.
A Hylaeus hyalinatus 1990. Europe. Currently found in urban areas from New York City, southern Ontario, New Jersey, Pennslyvania. Has potential to spread throughout North America.
A Hylaeus punctatus 1980. Europe. Currently found in central California, southern South America, New York City, Washington D.C. Has potential to spread throughout North America

Halictidae

A Lasioglossum eleutherense 1990. Bahamas and Cuba. Four individuals found in the University of Miami Arboretum. Current status unknown. Not expected to spread out of Florida.
A Lasioglossum leucozonium 1900s. Europe and northern China. Despite its extensive range in Europe and Asia it is limited to the northern areas of central and eastern U.S. and southern Canada.
A Halictus tectus 2000. Southern Europe to Mongolia. Known from 3 sites: downtown Philadelphia, PA, Baltimore, MD and Beltsville, MD. Appears to prefer highly disturbed sites with European weeds.

Megachilidae

A Anthidium manicatum 1960. Europe, North Africa, Near East, South Central and South Eastern South America. Currently found predominantly in northeastern U.S. and southern Canada, however, individuals have shown up in the central states, Idaho, and on the West Coast where it is well established in California. Likely to spread throughout North America. Associated with large urban and suburban gardens, particularly planted with *Stachys*.
A Anthidium oblongatum 1990. Europe and the Near East. Currently common in northeastern U.S. and southern Canada and moving into the central states and provinces. Found in most open habitats. Has potential to spread throughout North America.
A Chelostoma campanularum 1960. Europe and the Near East. Found in Upstate New York, Connecticut, and southern Ontario. Has potential to spread throughout North America.
A Chelostoma rapunculi 1960. Europe and the Near East. Found in Upstate New York and southern Ontario. Has potential to spread throughout North America.
A Coelioxys coturnix 2000. Southwestern Europe, North Africa, India. Currently found in the Baltimore-Washington D.C. corridor. Has potential to spread throughout the range of *Megachile rotundata* (its presumed host).
A Hoplitis anthocopoides 1960. Europe. Found from West Virginia to southern Ontario. Potential spread perhaps limited to the range of its reported preferred pollen source, Viper's Bugloss (*Echium vulgare*).
A Lithurgus chrysurus 1970. Europe, Near East, North Africa. Found only in the Phillipsburg, New Jersey are and a 50 mile radius in Pennsylvania and New Jersey. Until 2007 there were no recent records, but perhaps due to nobody making an effort to look. Apparently oligolectic on Spotted Knapweed (*Centaurea maculosa*) and burrows into wood to make a nest. This species has the potential to be much more destructive than *Xylocopa virginica*.
A Megachile apicalis 1930. Europe, North Africa, Near and Middle East. Western and eastern U.S. Relatively few records in the East but widespread in California and parts of the Pacific Northwest where it specializes on star-thistle *Centaurea solstitialis,* and is often moved around with Megachile rotundata pollinator tubes.
A Megachile concinna 1940. Africa. West Indies, Mexico, throughout the southern U.S.

A *Megachile ericetorum* 2000. Europe, Near East, China. One recent record from southern Ontario.
A Megachile lanata 1700-1800. India and China. Introduced into the West Indies and northern South America where it possibly made its way secondarily to Florida. Found throughout much of Florida but not likely to spread farther unless it is brought to the southwestern deserts.
A Megachile rotundata 1920-1940. Europe to China. Throughout North America to northern Mexico. Available commercially, used in alfalfa seed production.
A Megachile sculpturalis 1990. Far eastern China, Korea, Japan. Eastern and central U.S. and southern Canada. May move throughout the continent as they use widely planted, introduced summer blooming leguminous trees and shrubs.
A Osmia caerulescens 1800s. Europe, North Africa. Near East, India. Northeastern and Northcentral U.S. and southern Canada. Appears to be less common than it once was, at least towards the south. No recent records for the mid-Atlantic area despite a great deal of collecting, but still common in upstate New York.
I Osmia cornifrons 1960. Eastern China, Korea, and Japan. Introduced to pollinate tree fruit crops. Feral populations established in the Mid-Atlantic and Northeastern U.S. Available commercially.
I Osmia cornuta 1980. Europe, North Africa, Near East. Introduced as a pollinator of tree fruit crops in California, but its establishment has not been documented.
A Osmia taurus 2000. Eastern China, Japan. Mid-Atlantic area and Appalachian Mountains. Males in particular are very similar to *O. cornifrons* and may be confused. Appears to be rapidly spreading and often abundant.

Mini-summary of the Genera of Eastern North American Bees

(See information at the end of the document for an explanation of the codes and formatting)

H Agapostemon(4) N SpSUFL |NE|MAc|DS|MW|GL|OQ|AC 7-13mm Biggest of bright metallic green halictid bees. Bright green; strongly arched basal vein; raised line (carina) completely encircling the rear face of the propodeum. Some species surprisingly difficult to separate without experience, particularly males. Augochlorella, Augochlora, Augochloropsis
An Andrena(120) N SPsufl |NE|MAa|DS|MW|GL|OQ|AC 5-18mm Prominent facial fovea on females; most black, some males and females with yellow on clypeus. Several species are willow specialists and a few species have a reddish abdomen. Many subtle characters available to separate species, but when using guides score these very conservatively as there are more opportunities for error when the species number is high and the number of questions long and then double check against species accounts and the complete scoring for the species. Melitta, Colletes
Mg Anthidiellum(2) N spSUfl |ne|MAu|DS|GL|mw| - | - | 5-10mm Overhanging shelf on Dry habitats, often associated with legumes. Small, round, fast, chubby, black with strong yellow markings and dark wings. Scutellum extends backwards over metanotum and propodeum as a thin flat shelf. Trachusa, Stelis, Anthidium, Dianthidium
Mg Anthidium(4) N spSUfl |ne|MAu|DS|GL|MW|OQ|ac| 8-17mm Gardens and fields. Two introduced species are spreading throughout the region, both are common in gardens, the two native species are uncommonly encountered. Moderate-sized, stocky bees, fast fliers with strong yellow markings, particularly noticeable on the abdomen. Females have multiple teeth on their mandibles. Trachusa, Stelis, Anthidium, Dianthidium
Ap Anthophora(6) N SPSUfl |ne|MAu|DS|GL|MW|oq|AC| 8-19mm The introduced *A. plumipes* is spreading rapidly out of the Washington D.C. area and should be expected elsewhere soon. It is an early spring bee and occurs in woodlands as well urban and field habitats. The other species are late spring to summer species and occur in mixed habitats. Some species look superficially like bumblebees by body shape, while others look like the other Eucerines. The hairless internal cells of the forewing narrow the possibilities down to Anthophora and the rarer Habropoda and Melecta genera. Melecta, Xeromelecta, Florilegus, Tetraloniella, Melissodes, Svastra, Peponapis, Melitoma, Eucera
Ap Anthophorula(2) N suFL |-|-|ds|gl|mw|-|-| 4-9mm Open habitats. Very rare bees that have only been recorded from Indiana (last collected in Indiana in 1962), Virginia, and Mississippi. Similar to Exomalopsis in appearance and formerly included in that group, males have yellow or white on clypeus and labrum, which are dark in Exomalopsis. A bit smaller than a honeybee but both males and females extremely hairy, particularly the hind leg. Exomalopsis
Ap Apis mellifera(1) N SPSUFL |NE|MAa|DS|GL|MW|OQ|AC| 9-20mm Note that this species is only uncommonly caught in pan traps. Long hair on eyes and the unique hind leg architecture is a give away. Colletes

Mg Ashmeadiella(2) N spSU |-|-|DS|GL|mw|-|-| 4-11mm Uncommon to rare bees told from Hoplitis by the carina or raised line that defines the edge of narrow front section of the mesepisternum from the main side section. Chelostoma, Heriades, Osmia, Hoplitis
H Augochlora pura(1) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 5-9mm Open habitats and partially wooded. Most often confused with Augochlorella sp. Told by minutely truncate tip of marginal cell, the female's large dark forked tip of the mandible, and the rear suture of the clypeus. Also, female Augochlora have a keel or projection on the 1st sternum, which is not present in Augochlorella. Augochlorella, Augochloropsis, Agapostemon
H Augochlorella(3) N SPSUFL |NE|MAa|DS|GL|MW|OQ|AC| 3-10mm Fields and other open habitats. Most often confused with *Augochlora pura*. Told by the lack of a minutely truncate tip to the marginal cell. The female's mandible tip with a subapical tooth similar to most other halictids and the shape of the rear suture of the clypeus. Augochlora, Agapostemon, Augochloropsis
H Augochloropsis(3) N SPSUFL |ne|MAu|DS|GL|MW|oq|-| 6-12mm This bright green group regularly occurs in low numbers in most collections. The D-shaped non-oval tegula is distinctive in both sexes. Agapostemon, Augochlorella, Augochlora
Ap Bombus(28) p SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 7-29mm Common throughout all environments. In non-parasitic females the flattened tibia with a shiny, hairless area on the outer tibia face, surrounded by long hairs is distinctive. Under the microscope the lack of a jugal lobe is definitive, but often difficult to determine. Ptilothrix, Xylocopa, Centris
An <u>Calliopsis</u>(3) N spSUfl |NE|MAc|DS|GL|MW|OQ|AC| 4-10mm Open fields. The very common *C. andreniformis* often inhabits heavily used playing fields and other human-impacted sites. The small size, 2-submarginal cells, the bright yellow legs of the male and the 3 vertical ivory-colored facial markings of the females are a distinctive combination. Hesperapis, Andrena
C Caupolicana(2) N SUFL |-|-|DS|-|-|-|-| 8-12mm A rarely observed genus restricted to coastal dune areas in the deep south and the sandy central Florida ridge. These fast flying large species are usually only active at dawn and dusk. The first recurrent vein usually joins or nearly joins the first submarginal crossvein.
Ap Cemolobus ipomoeae(1) N SU |-|mar|DS|GL|MW|-|-| 10-17mm A large specialist on morning glories, very rarely detected. The rim of the clypeus has 2 lateral projecting knobs and a central latitudinally extended, projecting lobe. The other Eucerines have uninterrupted clypeal rims. Melitoma, Anthophora, Eucera, Melissodes, Tetraloniella, Melecta, Xeromelecta, Peponapis, Svastra, Florilegus
Ap Centris(3) N SPsufl |-|-|DS|-|-|-|-| 9-15mm An uncommon large fast-flying bumblebee/anthophora looking group. Currently restricted to Florida, but the introduced C. nitida could spread beyond the state. The males have a great deal of yellow on their clypeus and both the male and female have very robust rear legs, covered in thick hair. Bombus, Ptilothrix, Xylocopa
Ap Ceratina(4) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 2-9mm Found in most habitats. Small metallic steel blue bees, that tend to keep their abdomens more upright than other species. Abdomen parallel-sided, shaped like a plastic spring water bottle. Abdomen of the females comes to a distinct point and in the same region the males have a small projecting plate or flange.
Mg Chelostoma(3) N SPSU |ne|MAu|DS|GL|MW|oq|-| 4-9mm Small, exceedingly slender black bee. T1 does not have a carina and propodeum lacks pits beneath the metanotum. Ashmeadiella, Heriades, Osmia, Hoplitis
Mg Coelioxys(22) P SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 5-17mm Similar to appearance to Megachile whom they parasitize but usually narrower. Most females with a clearly pointed and extended abdomen tip. The tip of most male's abdomen with a unique set of spines or projections. The tips of the axillae extend out and back from the edge of the scutellum. Megachile, Lithurgus
C Colletes(35) N SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 6-15mm General body shape often similar to a honeybee. Face heart-shaped due to the angling inward of the compound eyes. Distinctive that lower portion of the second recurrent arches out toward wing tip. Apis
Mg Dianthidium(3) N SPSUFL |ne|mar|DS|GL|MW|oq|-| 5-12mm Uncommonly detected group in the East. Close in aspect to some Stelis but much less heavily pitted on mesepisternum. Has a rounded scutellum, aroliae, and a carina that runs part way down from the pronotal lobe partially down the mesepisternum. Paranthidium, Anthidium, Anthidiellum, Trachusa, Stelis
H Dieunomia(3) N SUFL |-|mar|DS|GL|-|-|-| 8-19mm An uncommon genus. The usual bent vein of the basal vein is only weakly present. 2 submarginal cells. Larger than almost all the other Halictid species other than Nomia. An overall dark bee without many distinctive features in the female. The male has greatly dilated mid tarsi. Lasioglossum, Halictus, Dufourea
H Dufourea(3) N SU |NE|MAr|ds|GL|mw|OQ|AC| 5-11mm Very uncommon bees. Antennal bases well below middle of face and separated from clypeus by not much more than diameter of an antennal

socket; clypeus short and wide, its upper margin not much arched up into face; labrum nearly as long as clypeus; pre-episternal groove present. Dieunomia, Halictus, Lasioglossum
Ap Epeoloides pilosula(1) P SU |ne|mar|-|gl|mw|oq|ac| 5-12mm A parasite of Macropis, not seen for years but recently spotted in NS and CT. Lacks the dense patches of appressed scutum hairs of Triepeolus and Epeolus. The marginal cell is separated from the wing margin and its apex is gradually bent away from the wing margin (the marginal cell touches the wing margin and has an apex that is on the wing margin and is more abruptly truncate than in most other similar bees). Triepeolus, Epeolus, Ericrocis
Ap Epeolus(19) N SPSUFL |NE|MAr|DS|GL|MW|OQ|AC| 5-12mm - Uncommon to rare robust bee with strong patterns of black and white on the thorax and abdomen, often with amber patches of integument present. Upon close inspection these patterns are made up of tiny fat hairs that lie prostrate across the surface of the integument. Can look remarkably like Triepeolus, but almost always smaller, otherwise the differences are technical and are addressed in the guides. Triepeolus, Epeoloides, Ericrocis
Ap Ericrocis lata(1) P SPSUFL |-|-|DS|-|-|-|-| 9-14mm - Known only from Florida where very rare. Most similar to Xeromelecta, has prominent patches of light hair on the abdomen and thorax and a distinctly pointed rear of the abdomen. A dramatic bee. Epeolus, Triepolus, Epeoloides
Ap Eucera(7) N SPSU |NE|MAu|DS|GL|MW|OQ|AC| 8-19mm Moderately common to uncommon bees, not as common as the similar, and often mistaken for, Melissodes, but expected in any large collection. Unlike Melissodes, these are most common in the spring. Identification of males depends on a careful examination of the triangular projections on the sides of T7. Care must be taken to look closely among the hairs for the complete lack of these angles. Females have completely oval tegula, unlike Melissodes. Other Eucerine groups need to be evaluated in the guides. Melissodes, Tetraloniella, Melecta, Xeromelecta, Cemolobus, Anthophora, Florilegus, Xenoglossa, Peponapis, Svastra
Ap Euglossa viridissima(1) N A recently discovered introduction, currently only occurring in South Florida. Bright green in color, we don't have sizes yet, but likely to be about the size of a honey bee, does not have the arched basal vein of the green halictids and has no arolia between its tarsal claws.
Ap Exomalopsis(2) N SPSUFL |-|-|DS|-|-|-|-| 4-9mm Extremely rare. Only a few specimens known and only from Florida. Smaller than honeybees, similar to Anthophorula, males have dark clypeus and labrum, extremely hairy, particularly for something so small. Anthophorula
Ap Florilegus condignus(1) N spSU |-|MAu|DS|GL|mw|-|-| 7-14mm – Uncommon in general, but may be locally common. Often mistaken for Melissodes but see the guides for details on how to separate. Melissodes, Melecta, Eucera, Tetraloniella, Melitoma, Svastra, Anthophora, Peponapis
Ap Habropoda laboriosa(1) N SP |ne|MAu|DS|GL|MW|-|-| 11-18mm A spring bumblebee like species, often associated with blueberries. Technically closer to some of the more uncommon Anthophora species than bumblebees. The shape and configuration of the marginal cell are key to telling this species. Anthophora
H Halictus(6) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 5-14mm Common field and urban species. Most often confused with Lasioglossum, particularly H. confusus specimens because of its metallic body. This will also be true of *H. tectus* a new metallic invasive that has been detected in Philadelphia and Washington D.C. The cross veins of the submarginal cells are all the same width, though this can take some time to be able to become familiar with, the hair bands on terga originate from the rim of the segment rather than from the base. Additionally the bottom of basal vein is usually more strongly arched than Lasioglossum and this group has a larger, more robust feel in direct comparison. Dieunomia, Lasioglossum, Dufourea
Mg Heriades(3) N SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 4-9mm Dark black, small size, narrow aspect along with a row of deep rectangular cells below the metanotum and T1 with a raised line (carina) surrounding the concave surface area is a distinctive combination. Ashmeadiella, Chelostoma, Osmia, Hoplitis
Mt Hesperapis(2) N SUFL |-|-|DS|GL|-|-|-| mm Very uncommon bees, restricted to coastal barrier islands in the Gulf and dunes of the Great Lakes. Abdomen noticeably flattened and integument soft compared to other groups. Calliopsis
Ap Holcopasites(3) P SPSUFL |NE|MAr|DS|GL|MW|OQ|AC| 2-9mm A rare and minute groups of parasitic species. Males unique (and therefore confusing) in that they have only 12 antennal segments. Abdomens are red with bright white patches of hair, often in small regular patches.
Mg Hoplitis(10) N SPSU |NE|MAc|DS|GL|MW|OQ|AC| 4-14mm Black somewhat elongate bees with parallel-sided abdomen. Similar to some of the black-colored Osmia but have in this case long parapsidial lines, in Osmia these lines don't run for more than 5 pit diameters. Ashmeadiella, Osmia, Chelostoma, Heriades

C Hylaeus(24) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 2-11mm Black, small, narrow, with relatively few hairs and no scopa as this genus carries pollen internally. Most females have elongate, thin, diamond yellow or ivory markings between the eye and clypeus/antennae while the males usually have more extensive yellow markings, with yellow throughout the area below the antennae.
H Lasioglossum(126) p SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 2-12mm A diverse group of largely small bees. Most species have one or two of the outer submarginal crossveins weakened. The weak veins are SLIGHTLY thinner and therefore appear a bit fainter; a subtle character that takes time to detect consistently. This character is most noticeable in females but less in males where it can be difficult at times to detect and consequently males may key out to the genus Halictus. Body type varies from all black to the common slightly metallic dark green and blue forms. The genus Halictus almost always has a hair fringe on the rims of the abdominal tergites that extends over the base of the next tergite. Lasioglossum, when a fringe or band of hair is present, has hair that is absent from the rim but is located at the very base of the segment and runs underneath the preceding segment. The effect is that in both groups the band of hairs appear in about the same location so an inspection under the microscope is necessary to determine where the band's true location lies. Lasioglossum specimens are, on average, a bit smaller and slighter in build than Halictus. Halictus, Dieunomia, Dufourea
Mg Lithurgus(3) N SPSU |-|mar|DS|gl|-|-|-| 8-19mm Similar to Megachile in appearance. Females have prominent projections or lobes arising just below their antennae and the males and females have the middle tooth of the mandible longest and most prominent. Males' labrum is longer than broad. Megachile, Coelioxys
Mt Macropis(4) N SPSUfl |NE|MAr|DS|GL|MW|OQ|AC| 5-12mm Rare bees, apparently much less common than in the past. Associated with loosestrife plants.
Mg Megachile(42) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 5-21mm Bees in this genus are larger than other species where the female has scopa on its abdomen. These are common wide-bodied bees, most with narrow white bands of hair on their abdomens. Has no arolia between the tarsal claws. Usually fly quickly between flowers often producing an audible hum. Lithurgus, Coelioxys
Ap Melecta pacifica(1) P SPSU |-|MAr|DS|GL|-|-|-| 10-15mm Very rare. Similar to other Eucerines, but separation technical. See genera guide. Xeromelecta, Anthophora, Tetraloniella, Svastra, Eucera, Melissodes, Melitoma, Florilegus, Peponapis, Xenoglossa, Cemolobus
Ap Melissodes(27) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 6-18mm Most common in summer and early fall. All very hairy, females with thick long scopa, fast fliers, robust, bumblebee-like bodies. Males have extremely long antennae. Females told from other Eucerines by the shape of the tegula, however, this is often hidden by dense hair and must be scrapped off with a pin tip in order to see. Melecta, Xeromelecta, Anthophora, Xenoglossa, Peponapis, Florilegus, Melitoma, Eucera, Svastra, Tetraloniella, Cemolobus
Ap Melitoma taurea(1) N SPSUfl |-|MAc|DS|GL|MW|-|-| 7-15mm Strong black and white bands on abdomen, not as hairy as Melissodes and Eucera. Unique in having a tongue that even when folded reaches to the abdomen. Melecta, Xeromelecta, Anthophora, Xenoglossa, Peponapis, Florilegus, Melissodes, Eucera, Svastra, Tetraloniella, Cemolobus
Mt Melitta(3) N SPSU |NE|MAr|DS|GL|MW|-|-| 7-14mm Andrena like, very rarely encountered. Scopal hairs on female only on tibia not on femur and trochanter-like Andrena; females also lack facial foveae. Males lack a basitibial plate. Andrena
Ap Neolarra cockerelli(1) P SPSUFL |-|-|DS|-|MW|-|-| 1-6mm Very rare. Probably the smallest bee in the East. Has but one submarginal cell.
Ap Nomada(70) P SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 2-17mm Wasp like, in their reduced body hair and thin legs. Both sexes with extensive yellow and red/orange markings, females more so. Abdomen usually held slightly above horizontal. Setae on the apical end of the hind tibia often very useful in identification, more so in females than males. Sphecodes
H Nomia(2) N SPSUFL |-|MAr|DS|gl|MW|-|-| 7-20mm Unique in that the terga have short bands along the rim that are enamel-like and mother-of-pearl colored with a strong green reflectance. Males have hind tibia that are dialated, sometimes greatly so. Dieunomia
Mg Osmia(29) N SPSU |NE|MAc|DS|GL|MW|OQ|AC| 5-17mm Stubby, most dark metallic blue or green, a few of the larger species are brown. Has no parapsidial line on thorax just a dot that is at most a few pit diameters wide. Hoplitis, Ashmeadiella, Heriades, Chelostoma
An Panurginus(3) N SPsu |-|MAu|DS|-|MW|-|-| 3-10mm Small, uncommon, black species with the males often having yellow on their face. 2 submarginal cells. Close to Pseudopanurgus, but told apart by vein patterns. Pseudopanurgus, Perdita, Protandrena
Mg Paranthidium jugatorium(1) N SUFL |-|MAr|DS|gl|MW|-|-| 6-11mm Uncommonly encountered. Similar to Dianthidium and Trachusa, see guide for details. Dianthidium, Stelis, Anthidium,

Anthidiellum, Trachusa
Ap Peponapis pruinosa(1) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 9-16mm Often confused with Melissodes, but has rounded tegulae. The female basitarsus is sparse compared to Eucera and Melissodes. Melecta, Xeromelecta, Anthophora, Xenoglossa, Florilegus, Melitoma, Eucera, Svastra, Tetraloniella, Cemolobus, Melissodes
An Perdita(26) N SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 2-10mm Among the smallest of bees. Most males and females have patterns of white or pale yellow on their face, thorax and abdomen. Short, truncate, marginal cell. Uncommonly collected but can be common in sandy localities. Pseudopanurgus, Panurginus, Protandrena
An Protandrena(3) N SPSUfl |-|MAr|DS|GL|MW|-|-| mm A very uncommon group, best told by keying them out through the guide.
An Pseudopanurgus(15) N SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 3-10mm Similar to Panurginus. Small, dark bees, with 2 marginal cells. Males have often extensive amounts of yellow on their faces. Can be difficult to differentiate species. Panurginus, Protandrena, Perdita
Ap Ptilothrix bombiformis(1) N SUFL |-|MAc|DS|GL|MW|-|-| 10-20mm Bumblebee-like, longer than normal legs that have long hooked claws, hair short and tightly packed, rounded crown to the head and lack of arolia pad between tarsal claws. Bombus, Xylocopa
H Sphecodes(41) P SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 2-13mm Many species have a bright red abdomen contrasting with dark black bodies, has a strongly bent base of the basal vein. Similar to Lasioglossum but females lack scopa, wings have no weak veins, most species have strongly sculptured propodeums. Nomada
Mg Stelis(12) P SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 3-12mm Uncommon, small to medium-sized. Variable in look but almost all have some yellow or white markings on their face and abdomen. Females lack scopa. Dianthidium, Anthidium, Anthidiellum, Paranthidium, Trachusa
Ap Svastra(5) N SPSUFL |-|MAu|DS|GL|MW|oq|-| 10-21mm Uncommon large Eucerine group. Both males and females have distinct, but often difficult to find, flattened hairs with spoon-shaped tips interspersed among the other hairs located on the abdomen. Melecta, Xeromelecta, Anthophora, Xenoglossa, Peponapis, Florilegus, Melitoma, Eucera, Melissodes, Tetraloniella, Cemolobus
Ap Tetraloniella(2) N SPSUfl |-|-|DS|GL|mw|-|-| 6-12mm A very uncommon Eucerine species, see guide for technical details. Melecta, Xeromelecta, Anthophora, Xenoglossa, Peponapis, Florilegus, Melitoma, Eucera, Svastra, Melissodes, Cemolobus
Mg Trachusa(5) N spSUFL |-|MAr|DS|GL|MW|-|-| 7-16mm Uncommon species. Females lack scopa. Dianthidium, Anthidium, Anthidiellum, Stelis, Paranthidium
Ap Triepeolus(24) P SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 6-18mm Like black and white oriental rug, swirling patterns on abdomen and thorax that under close inspection are made up of minute fat little hairs that are lying down across the surface. Told from the very similar Epeolus by features on the rear of the abdomen. Epeolus, Epeoloides, Ericrocis
Ap Xenoglossa(2) N SPSUFL |-|MAr|DS|GL|MW|-|-| 12-19mm Similar to Peponapis told apart by antennae and mandible characters. Melecta, Xeromelecta, Anthophora, Melissodes, Peponapis, Florilegus, Melitoma, Eucera, Svastra, Tetraloniella, Cemolobus
Ap Xeromelecta(2) P SPSUFL |-|-|-|GL|-|-|-| 6-17mm Rare. Similar to Melecta, see guide for technical details. Melecta, Melissodes, Anthophora, Xenoglossa, Peponapis, Florilegus, Melitoma, Eucera, Svastra, Tetraloniella, Cemolobus
Ap Xylocopa(2) N SPSUFL |NE|MAc|DS|GL|MW|OQ|-| 13-24mm Large, bumblebee-like. Males have prominent white facial markings, both with a very long marginal cell, hind wing with jugal lobe, black abdomen with few hairs and slightly iridescent surface readily visible. Bombus, Ptilothrix

Example Account and Followed by an Explanation of Formatting:

Ap Triepeolus(24) P SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 6-18mm Like black and white oriental rug, swirling patterns on abdomen and thorax that under close inspection are made up of minute fat little hairs that are lying down across the surface. Told from the very similar Epeolus by features on the rear of the abdomen. Epeolus, Epeoloides, Ericrocis

Ap = Family of Bees
Triepeolus = Genus
(24) = Number of species east of the Mississippi
P = Nest Parasitism
SPSUFL = Seasonal Occurrence

|NE|MAu|DS|GL|MW|OQ|AC| = Regional Occurrence
6-18mm = Size range
Like….= Genus notes
Epeolus, Epeoloides, Ericrocis = Similar Genera

Explanation of Codes

Families of Bees: An Andrenidae, Ap Apidae, C Colletidae, H Halictidae, Mg Megachile, Mt Mellitidae

Nest Parasitism: N No species parasitic, P All species parasitic, p Some species parasitic, most not

Seasonal Occurrence: SP Spring, SU Summer, FL Fall. Lowercase indicates that group only uncommonly occurs during that season.

Regional Occurrence: NE New England, MA Middle Atlantic, DS Deep South, GL Great Lakes, MW Mid-West, OQ Ontario and Quebec, AC Atlantic Canada. Lower case indicates that this genus only occurs rarely in the region. A hyphen indicates the genus is absent in that region.

Many thanks to Mike Arduser, John Pascarella, John Ascher, Cory Sheffield, and Rob Jean for their corrections and additions to this section.

Pronunciation Guide to the Bee Genera of the United States and Canada (and Selected Subgenera)

Created: Fall 2003 – Modified Fall 2008

This pronunciation guide is designed to help the beginning bee biologist. What are presented appears to be the most commonly understood pronunciation of the bee genera (and a few important subgenera) occurring in North America north of Mexico. You can expect to hear a number of differing pronunciations as you talk with researchers and taxonomists, as pronunciation is governed by cultural rules rather than strict definitions. Suggestions for changes or additions are encouraged and can be sent to Sam Droege (sdroege@usgs.gov).

Acanthopus */a-CAN-tho-puss/*
Agapanthinus */aguh-PAN-thin-us/*
Agapostemon */ag-uh-PAHST-eh-mon/*
Agapostemonoides */aguh-pahst-em-OH-noy-dees/*
Aglae */AG-lee/*
Aglaomelissa */ag-lay-oh-mel-ISS-uh/*
Ancylandrena */ann-sill-ann-DREE-nuh/*
Ancyloscelis */ann-sill-oh-SELL-iss/*
Andinaugochlora */ann-din-aug-oh-KLOR-uh/*
Andrena */ann-DREE-nuh/*
Anthedonia */ann-theh-DOE-knee-yuh/*
Anthemurgus */ann-theh-MURG-us/*
Anthidiellum */ann-thid-e-ELL-um/*
Anthodioctes */ann-thoh-dee-OCK-tees/*
Anthidium */ann-THID-ee-yum/*
Anthophora */ann-THAH-for-uh/*
Apis */A-piss/*
Ashmeadiella */ASH-mead-ee-el-uh/*
Atoposmia */ate-op-oz-meeuh/*
Augochlora */awe-go-KLOR-uh/*
Augochlorella */awe-go-klor-EL-uh/*
Augochloropsis */awe-go-klor-OP-sis/*
Aztecanthidium */Az-tech-ann-THID-ee-yum/*
Bombus */BOM-bus/*
Brachynomada */bracky-no-MOD-duh/*

Caenaugochlora */seen-aug-oh-KLOR-uh/*
Caenohalictus */seen-oh-hal-ICK-tus/*
Calliopsis */cal-LEE-op-sis/*
Caupolicana */kaup-po-lih-CAN-uh/*
Cemolobus */sea-moh-LOW-bus/*
Centris */SEN-tris/*
Ceratina */sera-TIE-nuh/*
Chelostoma */chel-AHST-oh-mah/*
Cephalotrigona */seph-al-oh-trig-OH-nuh/*
Chlerogella */clair-oh-GELL-uh/*
Chilicola */chill-LICK-oh-luh/*
Coelioxoides */sealy-ox-OID-ees/*
Coelioxys */sealy-OX-ees/*
Colletes */koh-LEE-teez/*
Conanthalictus */koh-nanth-hal-ICK-tuss/*
Crawfordapis */kraw-ford-A-piss/*
Ctenioschelus */ten-eeoh-SHELL-us/*
Deltoptila */delt-OH-till-uh/*
Diadasia */die-uh-DAY-zeeuh*
Dialictus */die-uh-LICK-tuss/*
Dianthidium */die-ann-THID-ee-um/*
Dieunomia */die-u-NOH-mee-uh/*
Dinagapostemon */dine-ag-uh-PAHST-eh-mon/*
Dioxys */die-OX-eez/*
Doeringiella */dew-er-rinj-ee-EL-uh/*
Dolichostelis */dole-ih-koe-STEEL-iss/*
Duckeanthidium */duck-ee-ann-THID-ee-um/*
Dufourea */dew-four-EE-uh/*
Epanthidium */epp-ann-THID-ee-um/*
Epeoloides */e-pee-oh-LLOYD-eez/*
Epeolus */e-pee-OH-lus/*
Epicharis */EPI-care-us/*
Ericrocis */air-ih-KROE-sis/*
Eucera */u-SIR-uh/*
Eufriesea */u-FREE-jee-uh/*
Eulaema */u-LEE-ma/*
Eulonchopria */u-lon-chaw-PREE-uh/*
Evylaeus */ev-uh-LEE-us/*
Exaerete */ex-ee-RAY-tee/*
Exomalopsis */ex-oh-mal-LOP-sis/*
Florilegus */flor-ih-LEG-us/*
Frieseomelitta */freeze-ee-oh-mel-IT-tuh/*
Gaesischia */jee-sish-SHEE-uh/*
Geotrigona */jee-oh-trig-OH-nuh/*
Habralictus */hab-rah-LICK-tuss/*
Habropoda */hab-roh-PO-duh/*
Halictus */ha-LICK-tuss/*
Hemihalictus */hem-ee-hah-LICK-tuss/*
Heriades */her-EYE-ah-deez/*
Hesperapis */hes-per-A-piss/*
Heterosarus */het-er-o-SAUR-us/*
Hexepeolus */hex-ee-PEE-oh-lus/*
Holcopasites */hole-koe-pah-SITE-eez/*
Hoplitis */hop-LIE-tuss/*
Hoplostelis */hop-low-STEE-liss/*
Hylaeus */hi-LEE-us/*
Hypanthidioides */hi-pan-thid-EE-oid-eez/*
Hypanthidium */hi-pan-thid-EE-um/*
Lasioglossum */laz-ee-oh-GLOSS-um/*
Leiopodus */lee-ee-oh-pod-us/*

Lestrimelitta */less-trih-mel-IT-tuh/*
Lithurge */LIH-thurj/*
Macropis */ma-CROW-piss/*
Macrotera */ma-CROW-terr-uh/*
Martinapis */mar-TIN-a-piss/*
Megachile */meg-uh-KILE-ee/*
Megalopta */meg-uh-LOP-tah/*
Megaloptilla */meg-uh-lop-TILL-uh/*
Megandrena */meg-ann-DREE-nuh/*
Megommation */meg-ohm-MAY-shun/*
Melecta */mel-LECK-tuh/*
Melissodes */mel-ih-SOH-deez/*
Melissoptila */mel-lis-SOP-till-uh/*
Melipona */mel-LIH-poe-nuh/*
Melitoma */mel-lih-TOE-mah/*
Melitta */mel-IT-tuh/*
Meliwillea */mel-lih-WILL-eeuh/*
Mesocheira */meez-oh-KEER-uh/*
Mesoplia */meez-oh-PLEE-uh/*
Mesoxaea */meez-ox-EE-uh/*
Metapsaenythia */met-uh-see-NEE-thee-uh/*
Mexalictus */mex-al-LICK-tus/*
Micralictoides */mike-crugh-lick-TOY-deez/*
Microsphecodes */mike-crow-SFECK-code-eez/*
Monoeca */mon-EE-kuh/*
Mydrosoma */my-droh-SOH-muh/*
Nanorhathymus */nan-oh-rath-THIGH-mus/*
Nannotrigona */nan-oh-trig-GOH-nuh/*
Neocorynura */knee-oh-CORY-nur-uh/*
Neolarra */knee-oh-LAIR-uh/*
Neopasites */knee-oh-pass-EYE-teez /*
Nesosphecodes */knee-zoh-sfeck-O-deez /*
Nogueirapis */nog-yew-ee-eye-RAPE-is/*
Nomada */no-MOD-uh/*
Nomia */NO-mea/*
Odyneropsis */oh-dee-ner-OP-sis/*
Oreopasites */oh-reeoh-pass-EYE-teez/*
Osiris */oh-SIGH-ris/*
Osmia */OZ-me-yuh/*
Oxaea */ox-ee-aye/*
Oxytrigona */ox-ee-trig-OH-nuh/*
Panurginus */pan-ur-JINE-us/*
Paragapostemon */pear-uh-ag-uh-PAHST-eh-mon/*
Paralictus */pear-uh-LICK-tuss/*
Paranomada */pear-uh-no-MOD-uh/*
Paranthidium */pear-uh-an-thid-EE-um/*
Paratetrapedia */pear-uh-tet-rah-PEE-dee-uh/*
Paratrigona */pear-uh-trig-OWN-uh/*
Partamona */part-uh-MOW-nuh/*
Peponapis */PEE-po-nay-piss/*
Perdita */per-DIH-tuh/*
Pereirapis */pear-ee-eye-RAPE-is/*
Plebeia */pleb-ee-EE-uh*
Prostelis */proe-toe-STEEL-iss/*
Protandrena */prot-an-DREE-nuh/*
Protodufourea */pro-toe-dew-four-EE-uh/*
Protosmia */pro-TOZ-mee-uh/*
Protosiris */pro-toe-SIRE-is/*
Protoxaea */pro-tox-EE-uh/*
Pseudopanurgus */sue-doe-pan-UR-gus/*

Pseudaugochlora */sood-aug-oh-KLOR-uh/*
Psithyrus */SITH-ih-russ/*
Ptilocleptis */till-oh-KLEP-tiss/*
Ptiloglossa */till-oh-GLOSS-uh/*
Ptilothrix */til-O-thricks/*
Ptilotrigona */till-oh-trig-OH-nuh/*
Rhathymus */rath-THEE-mus/*
Rhinetula */rhine-ET-tule-uh/*
Rhopalolemma */rope-al-oh-LEM-uh/*
Scaptotrigona */scrap-toe-trig-OH-nuh/*
Scaura */SCOUR-uh/*
Simanthedon */sigh-MAN-theh-don/*
Sphecodes */sfeck-OH-deez/*
Sphecodogastra */sfeck-kode-oh-GAST-ruh/*
Sphecodosoma /sfeck-oh-doe-SOH-ma/
Stelis */STEEL-iss/*
Svastra */SVAS-tra/*
Syntrichalonia */sin-trick-uh-loan-EE-uh/*
Temnosoma */tem-no-SOH-mah/*
Tetragona */tet-rah-GOHN-uh/*
Tetragonisca */tet-rah-go-NISK-uh/*
Tetraloniella */tet-rah-LOAN-ee-el-uh/*
Tetrapedia */tet-rah-pee-DEE-uh/*
Thalestria */thal-lest-TREE-uh/*
Thygater */thigh-GATE-er/*
Townsendiella /town-send-ee-EL-uh/
Trachusa */trah-KOOS-uh/*
Trigona */trig-OH-nuh/*
Trigonisca */trig-oh-NIS-cuh/*
Triopasites */tree-oh-pass-EYE-teez/*
Xenoglossa */zee-no-GLOSS-uh/*
Xeralictus */zeer-ah-LICK-tus/*
Xeroheriades */zeer-oh-her-EYE-uh-deez/*
Xeromelecta */zero-mel-LECK-tuh/*
Xylocopa */zile-low-COPE-uh/*
Zacosmia */zack-OZ-mee-uh/*
Zikanapis */zick-ann-A-piss/*

Glossary of Bee Taxonomic Terms

Angulate – forming an angle rather than a curve
Anterior – Toward the head or on the head side of a segment being described
Apex – end of any structure
Apical – near or at the apex or end of any structure
Appressed – tight and flat against the body of the bee, usually used to describe hair
Arcuate – curved like a bow
Areolate – integumental (skin) sculpture pattern: divided into a number of small irregular spaces, very similar if not used interchangeably with reticulate
Arolia –the pad between the claws found at the ends of some bees legs
Bands – Usually referring to bands of hair or bands of color that traverse across an abdominal segment from side to side
Basad (Basally) – toward the base
Base (Basal Area) – on whatever part being described, this would be the section or the area at or near to the point of attachment, or nearest the main body of the bee, the opposite end of which would be the apical area
Basitarsus – the segment of the tarsus that is the nearest to the bee's body….usually the largest of all the tarsal segments
Basitibial plate – a small plate or saclike projection at the base of the hind tibia (like a bee knee pad)
Bifid – cleft or divided into 2 parts; forked
Carina – a clearly defined ridge or keel, not necessarily high or acute, usually appears on bees as

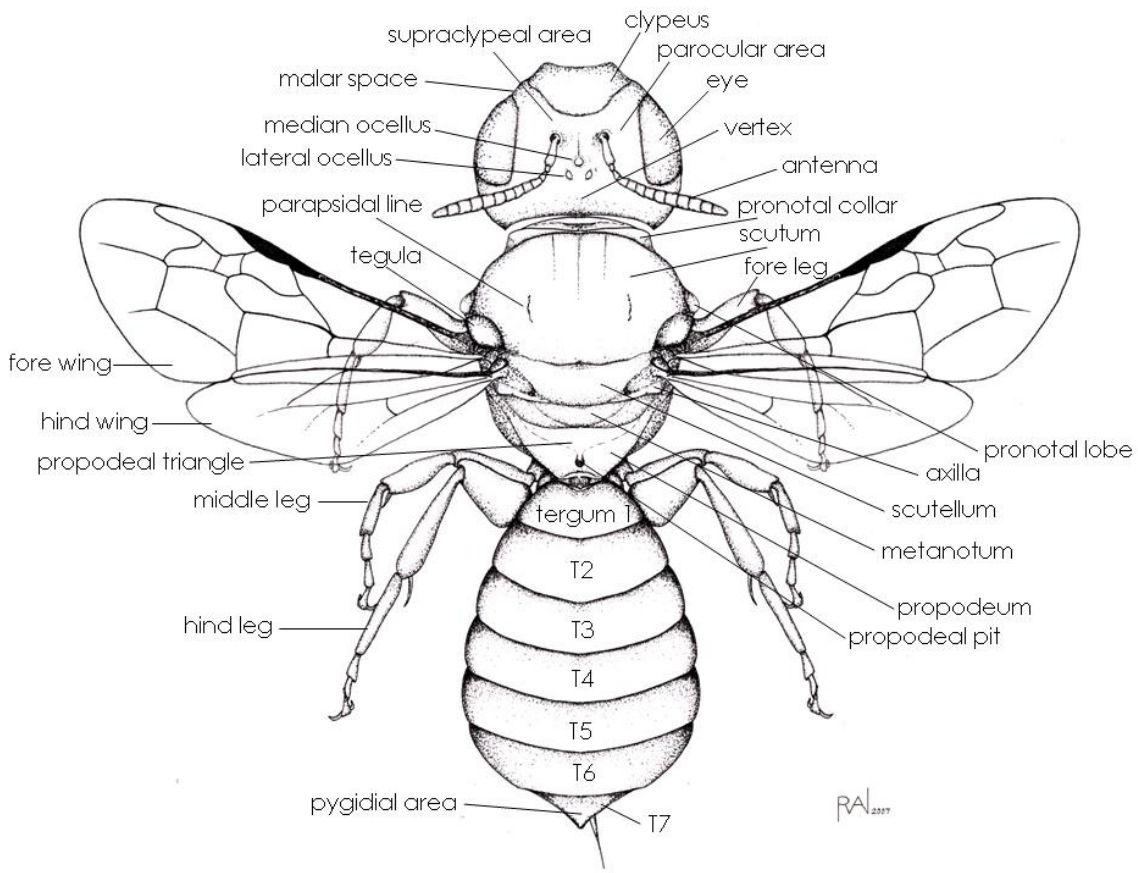
simply a raised line
Carinate – keeled; having keels or carinae
Caudad – towards the tail, or on the tail side of a segment being described
Cheeks – the lateral part of the head beyond the compound eyes, includes the gena and the subgena
Clypeus – a section of the face below the antennae, demarcated by the epistomal sutures
Conically – cone shaped, with a flat base, tapering to a what is usually a blunt or rounded top
Convex – the outer curved surface of a segment of a sphere, as opposed to concave
Corbicula – a hairless area or patch surrounded by longer hairs used to hold and transport pollen. Bumblebees and honeybees have this on their tibia, while Andrena have a patch on the sides of their propodeum
Costa – wing vein
Coxae – the basal segment of the leg
Cubital – wing vein
Denticle – a small tooth-like projection
Disc – a generic term for the middle surface of a plate (usually in reference to an abdominal segment) as apposed to what might be going on along the sides
Distal – away from the body or a description of a place on a segment that is furthest from the place of attachment with the body of the bee
Dorsum – in general, the upper surface
Echinate - thickly set with short, stout spines or prickles
Emarginate – a notched or cut out place in an edge or margin, can be dramatic or simply a subtle inward departure from the general curve or line of the margin or structure being described
Fasciae – a transverse band or broad line, in bees often created by a band of light colored hairs on the abdomen
Ferruginous – rusty, red – brown, orange-brown
Flagellum – the third and remaining part of the antenna beyond the pedicel and scape, containing most of the antennal segments
Fore – usually refers to the first pair of legs, the ones closest to the head
Foveae – a depressed region of cuticle, in bees this depressed area is usually only very slightly hollow and usually on the face
Fulvous – a brownish yellow-tawny color to orange brown
Fuscous – dark brown, approaching black; a plain mixture of brown and red
Glabrous – a surface without any hairs
Glossa – part of the tongue
Gradulus – a line that runs from side to side on abdominal segments of some bees that is formed by the step between two regions that differ in height, often that difference is only apparent upon very close inspection
Hyaline – transparent, glassy
Hypoepimeral – area on the thorax
Hypostoma – the notched region underneath the head and behind the mandible that holds the folded tongue
Impressed area – almost always refers to the rear part of the upper abdominal segments, these areas often being very slightly (often very difficult to detect) lower than the front part of the segment
Impunctate – not punctate or marked with punctures or pits
Infuscated – smoky gray-brown, with a blackish tinge
Inner – usually refers to legs and refers to the part that faces the body
Integum – the outer layer of the bee; the skin or cuticle
Intercubital – wing vein
Interstitial – when describing veins it refers to the end of one approximating the beginning of another, as in a grid intersection
Labrum – abutting the clypeus in front of the month
Macula – a spot or mark
Maculations – spotted or made up of several marks
Malar space – the shortest distance between the base of the mandible and the margin of the compound eye often completely absent in bees
Mandibles – bee teeth, so to speak, usually crossed and folded in front of the mouth
Marginal cell - a wing cell located on the edge (margin) of the wing
Mesally (Medially) - pertaining to, situated on, in or along the middle of the body or segment
Mesepisturnum, Mesopleura, or Mesothorax or the second or middle segment of the thorax bearing the middle legs and the forewings, the pronotum is the first segment

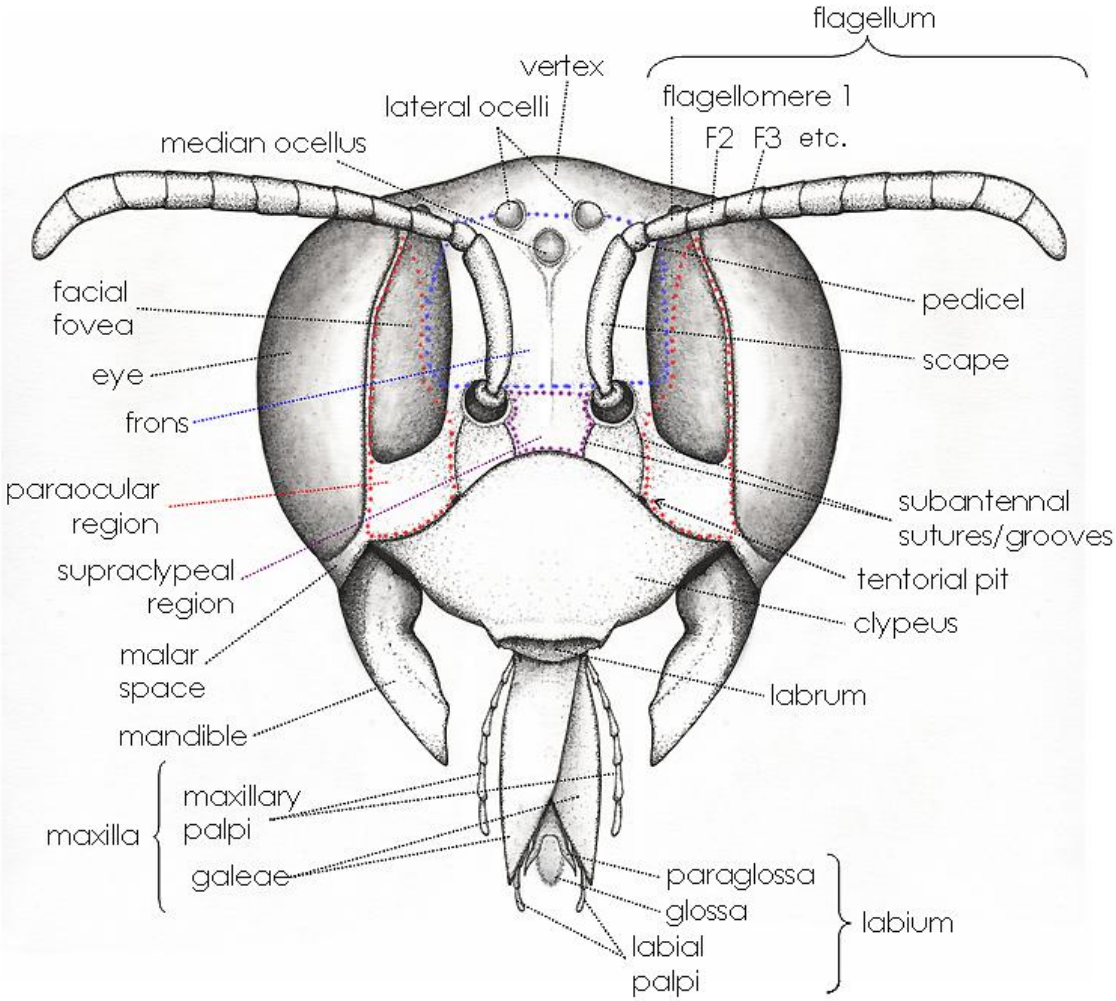
Metapleura – thorax segment bearing the hind legs and hind wings
Notaulices - a pair of lines on some bees that appear on either side of the scutum near the base of the wings
Ocelli – the 3 simple eyes or lenses that sit at the top of the head of bees
Ochraceous – pale yellow
Papillae (Papilate) – very tiny short hard cone-like projections usually in bees only found on the wing or legs and often having small hairs arising from the top
Outer – usually refers to legs and specifically to the surfaces facing away from the body
Pectinate – comb-like, having large comb-like teeth that are clearly separate from one another
Petiolate – having a stalk
Piceous – glossy brownish black in color, pitch like
Pleura - the lateral or side areas of the thorax, excluding the lateral surfaces of the propodeum
Plumose – feather-like
Pollex – a thumb; the stout fixed spur at the inside of the tip of the tibia
Posterior – toward the tail end or on the tail end of a segment being described
Preapical – referring to a section of a bee that is just physically found just before the outermost (or apical) end of the section or segment
Pronotum – a collar-like segment on the thorax and directly behind the head; extends down the sides of the thorax toward the first pair of legs
Propodeum – the last segment of a bees thorax (although you wouldn't know to look at it, it is considered anatomically part of the abdomen)
Prothoracic – of or pertaining to the prothorax
Protuberant – rising or produced above the surface or the general level, often used as a term to define a single or pair of small bumps
Proximal - that part nearest the body
Pubescent – downy; clothed with soft, short, fine, loosely set hair
Pygidial plate – unusually flat area (a plate) surrounded by a ridge or line and sometimes sticking well off of the end of the bee. If present, found on the sixth upper abdominal segment in females, seventh in males
Repose – in a retracted physical state
Reflexed – bent up or away
Reticulate - made up of a network of lines that creates a set of netlike cells, similar to areolate except perhaps more of a regular network of cells….undoubtedly both have been used to describe the same patterns at times
Rugose – a wrinkled set of bumps that are rough and raised like a short nappy rug
Scape - the first or basal segment of the antenna
Scopa - a brush; a fringe of long dense and sometimes modified hairs designed to hold pollen
Scutellum - shield shaped plate behind scutum
Scutum - the large segment on top of the thorax located between the wings and behind the head
Serrate – notched on the edge, like a serrated knife
Setose - covered with setae or stiff short hairs
Sinuate – the margin with wavy and strong indentations
Spatulate – shaped like a spatula
Spicule - small needlelike spine
Spinose - armed with thorny spines, more elongate than echinate
Sterna – the plates on the underside of the abdomen
Stigma - a thickened colored spot or cell in the forewing just behind the costal cell
Striae – a set of parallel lines (usually raised) and can be thick or thin
Subapical - located just behind the apex of the segment or body part
Subcontiguous – not quite contiguous or touching
Subequal - similar but not necessarily exactly equal in size, form, or length
Submarginal cells- one or more cells of the wing lying immediately behind the marginal cells
Subrugose- a bit bumpy but not forming an extensive set of wrinkled bumps
Sulcus - groove; more of an elongate hole or puncture in the skin of the bee
Supra - above, beyond or over
Supraclypeal area - the region of the head between the antennal sockets and clypeus, demarcated on the sides by the subantennal sutures
Suture - a groove marking the line of fusion of two distinct plates on the body or face of a bee
Tarsus - the leg segments at the end of the bees leg, attached to the tibia
Tegulae - the usually oval, small shield like structure carried at the extreme base of the wing where it

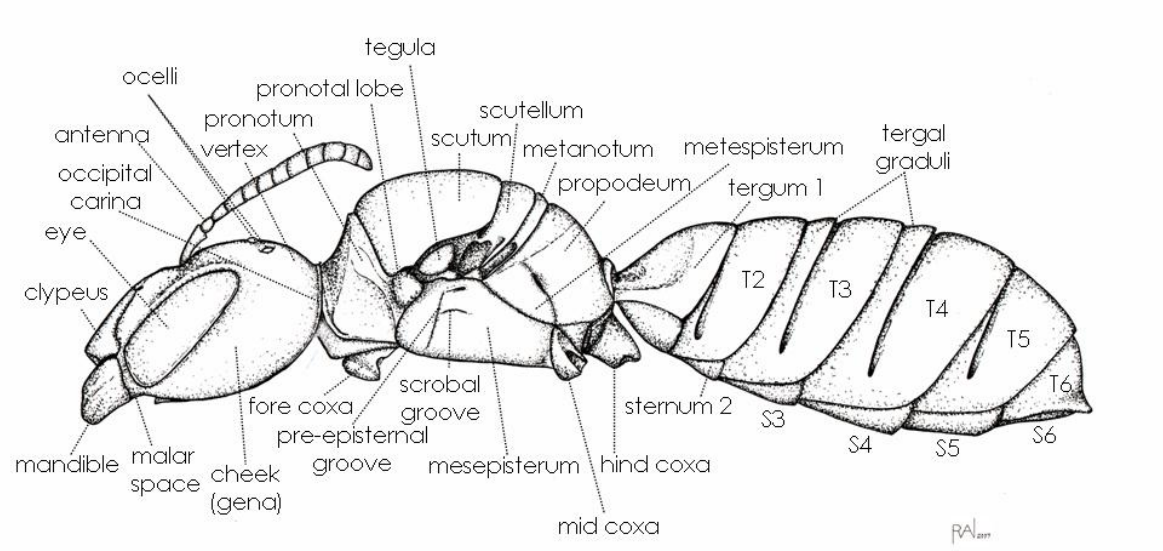
attaches to the body
Tergum – the segments on the top side of the abdomen
Tessellate – small very fine lines that make up a network of squares like a chessboard on the surface of the skin. Can often be very faint markings that appear like fingerprints on the shiny surface of the skin.
Testaceous - brownish-yellow
Tibia - segment of the leg, between the femur and the tarsus
Tomentum – a form of pubescence composed of short matted, woolly hair
Tomentose - covered with tomentum
Transverse – across the width of the body or segment rather than the length, in other words at right angles to the head to abdomen axis of the body
Trochanters – segment of the insect leg between the coxa and the femur
Truncate – cut off squarely at tip
Tubercle – a small knoblike or rounded protuberance
Undulate – wavy
Venter – the undersurface of a section of a bee or bee part, usually the abdomen
Ventral – pertaining the undersurface of the abdomen
Vertex – the top of the head
Violaceous – violet colored

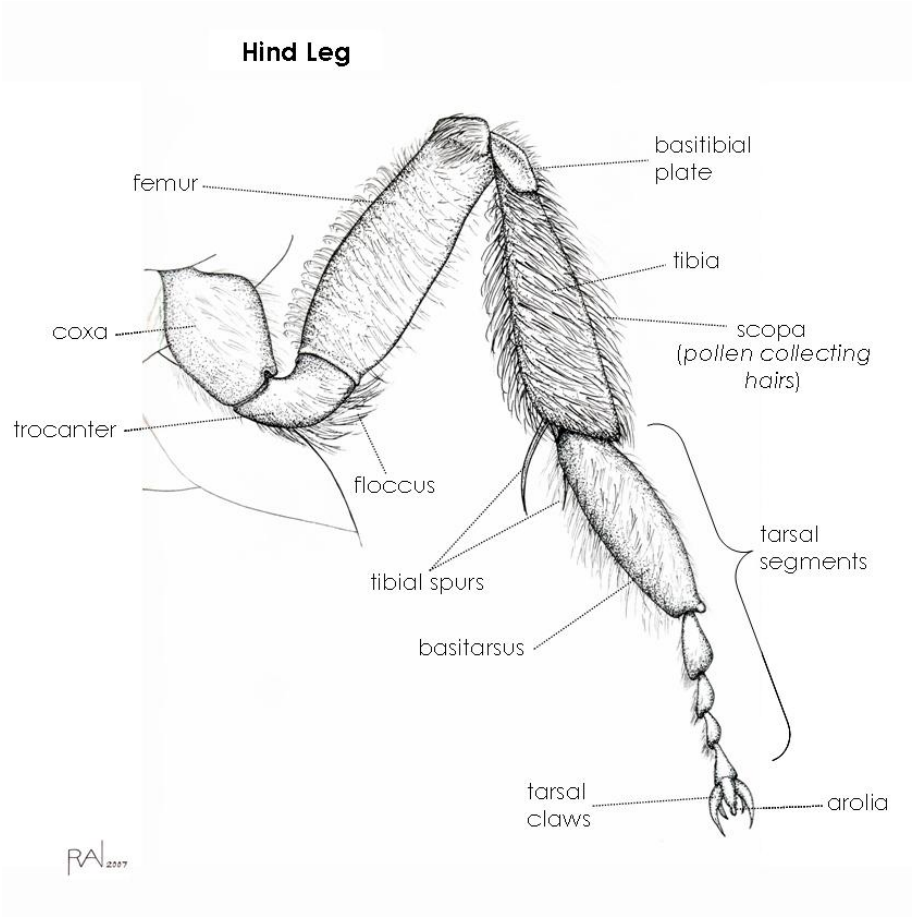
Bee Body Part Figures

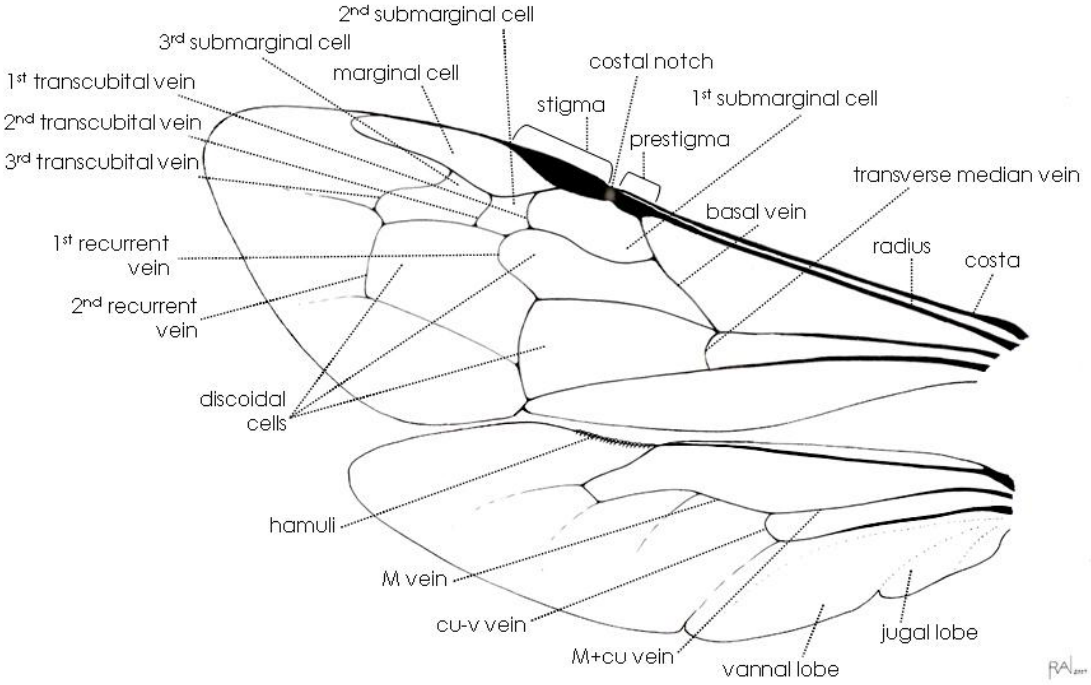
Drawn by Rebekah Nelson

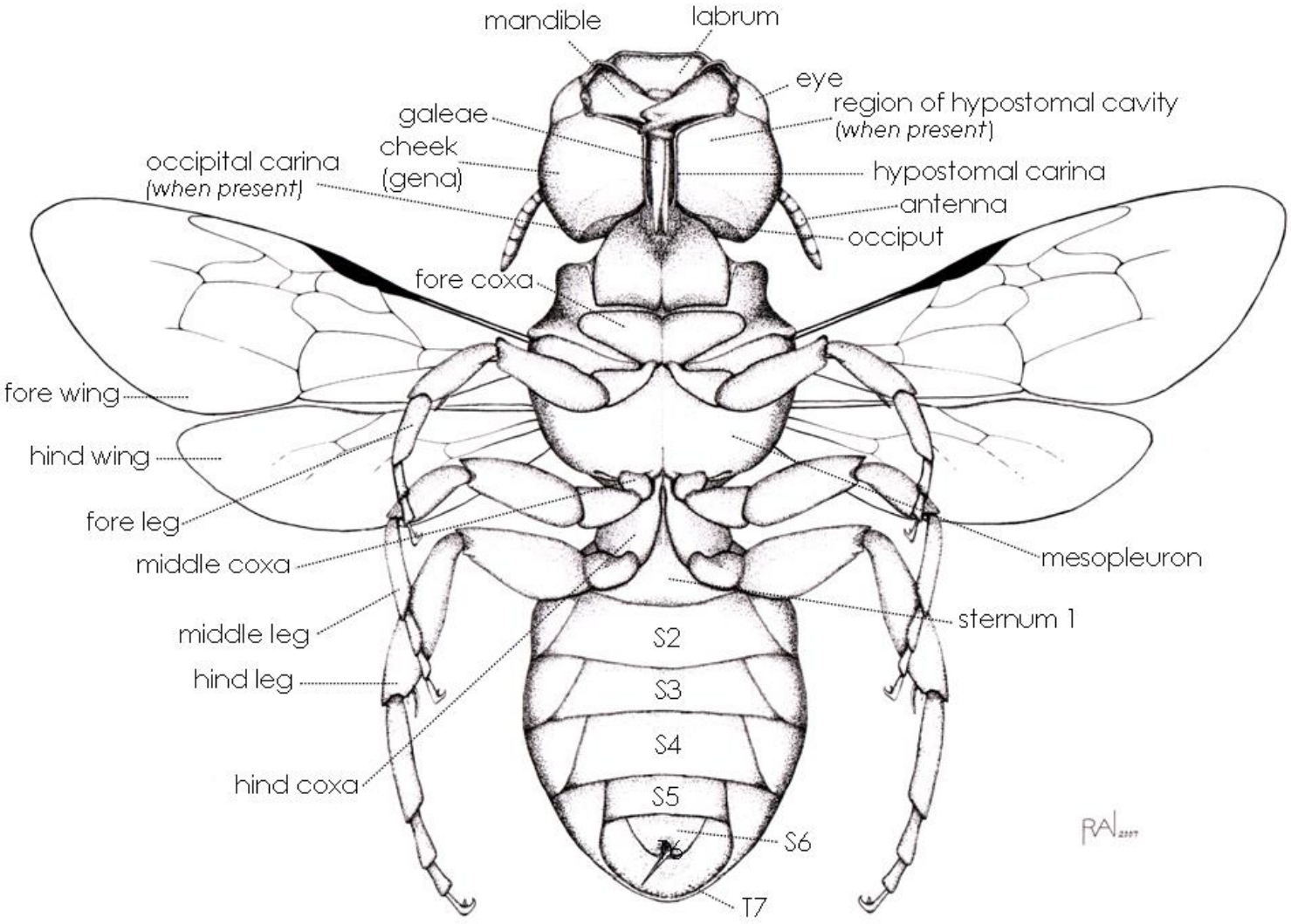



flagellum
vertex
lateral ocelli
flagellomere 1
F2 F3 etc.
median ocellus
facial fovea
pedicel
eye
scape
frons
paraocular region
subantennal sutures/grooves
supraclypeal region
tentorial pit
clypeus
malar space
labrum
mandible
maxillary palpi
maxilla
galeae
paraglossa
glossa
labium
labial palpi


tegula
ocelli
pronotal lobe
pronotum
scutellum
antenna
vertex
scutum
metanotum
metespisterum
tergal graduli
occipital carina
propodeum
tergum 1
eye
clypeus
T2
T3
T4
T5
T6
scrobal groove
fore coxa
sternum 2
S3
S4
S5
S6
pre-episternal groove
mandible
malar space
cheek (gena)
mesepisterum
hind coxa
mid coxa


Hind Leg
femur
basitibial plate
tibia
coxa
scopa (pollen collecting hairs)
trocanter
floccus
tarsal segments
tibial spurs
basitarsus
tarsal claws
arolia


2nd submarginal cell
3rd submarginal cell
marginal cell
costal notch
1st transcubital vein
stigma
1st submarginal cell
2nd transcubital vein
prestigma
3rd transcubital vein
transverse median vein
basal vein
1st recurrent vein
radius
costa
2nd recurrent vein
discoidal cells
hamuli
M vein
cu-v vein
jugal lobe
M+cu vein
vannal lobe


mandible
labrum
eye
region of hypostomal cavity
(when present)
galeae
cheek
(gena)
occipital carina
(when present)
hypostomal carina
antenna
occiput
fore coxa
fore wing
hind wing
fore leg
middle coxa
mesopleuron
sternum 1
middle leg
hind leg
hind coxa
S2
S3
S4
S5
S6
T7