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 whom 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 Bee Inventory and Monitoring Lab (BIML), Patuxent Wildlife Research Center, Beltsville, Maryland, 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 "Bee Inventory, Monitoring, and ID" discussion group. Many thanks to Dave and Janice Green, Gene Scarpulla, Liz Sellers, and Tracy Zarrillo for their many hours of editing this manual.

> "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, discussing environmentalists) – Provided by Matthew Shepherd

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Where to Find Bees

Bees are nearly ubiquitous; they occur essentially everywhere. However, in any given landscape there are usually a few good places to collect bees where they are concentrated, diverse, abundant, and easy to capture and there are many, many places where bees are difficult to find and collect. If you are interested in biodiversity, and taxonomic surveys, it will be important to discover these hotspots. In North America, in general, good collection locales will be places where floral composition is concentrated or unusual. If you are unfamiliar with an area, then exploring road/stream/river crossings, power line rights-of-way, railroad track rights-of-way, sand and gravel operations, open sandy areas, and wetlands are good places to start. In areas with a lot of development, the industrial sector often contains weedy lots and roadsides that also can have good numbers of bees. Note that just because there are few or no plants blooming (to your eye!), this doesn't mean that there are no bees present. A good collecting strategy is to put out bee bowl traps (see sections below) in the morning, and return mid-day to good potential collecting sites that you spotted earlier that morning.

Killing Bees to Study Them

In bee work, we almost all are confronted by the issue of having to kill the things we study and explaining that to the public as well as to land managers. Jessica Rykken pointed out a good essay on that topic at:

http://www.biology.ualberta.ca/uasm/alg/downloads/whywekillbugs.pdf.

Nets

Almost any type 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 greater the area of capture, however, larger hoops are more difficult to swing quickly due to air resistance, and there is more netting to snag on branches.

BioQuip Products (https://www.bioquip.com/) makes a net that is very portable for travel or backpacking. The pole disconnects into three small sections and the hoop can be folded into itself. 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 their 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 than the traditional aerial net bag can keep the smallest *Perdita* from escaping.

Zip Net – Sol Sepsenwol has created a great new net which he calls the "Zip Net" (see picture below). He has modified collecting nets so that you can attach sandwich baggies to the end of a cloth sweep or beater net. Such a modification permits you to sweep or capture an insect or group of insects and easily inspect them through the baggie walls. If warranted, you can then simply remove the baggie with the insect for further processing.

Since net handling time and the every field day inconvenience of trying to determine what you have captured is often a bottleneck in field work particularly for the new technician, this is a productivity boon. For those doing plant pollination studies, one simply has to sweep up the insect and, "Boom," pull the bag off to complete an uncontaminated collection. In his paper (Sepsenwol, S. 2014. The Zip Net: An Insect Sweep Net with Removable Capture Pouch for Serial Collecting. *American Entomologist* 60(4):207-209), Sol also demonstrates how baggies can have kill canisters inserted and how to transfer specimens to alcohol or kill jars from the baggie.

Note: this is also a great way to show people insects and bees without have to wrangle them out of nets.

The Zip Net

Netting Technique

When in the field, always hold your net in a "swing-ready" position. One hand should be below the head and the other towards the back or middle of the pole. Hold the tip of the net lightly against the pole with the hand near the head so that it does not drag in vegetation. When you start your swing drop the tip of the net.

Bees are best detected by their motion, rather than their size and shape. The mind detects motion much faster than it can process colors and shapes into bee/not bee categories. Train yourself to key in on movement; over time you will become more adept at separating bee motion from plant and other insect motion.

Bees are lost when you hesitate or check your swing. If you see something that looks like a bee, capture it in your net. Once in your net you can decide whether or not to keep it. If you spend any significant time thinking about whether you should or should not swing, the capture opportunity will be missed as the bee will have moved on.

Always keep a mental check for the presence of thorny plants in the area where you might swing - for the obvious tearing consequences to your net. Additionally, in some areas, some plants have clinging seeds that can implant themselves directly into the netting; if that is the case then you might try moving from the usual coarse weave net bag to the fine weave type that BioQuip sells.

When swinging a net, speed is important as well as follow-through. Bees are very visual and very fast. If you are timid in your swing or cut your swing short, bees will evade the net. Center your net on the bee if at all possible, even if it means having to plow through some vegetation. When a bee is flying low to the ground, it is better to slap the net over the bee than it is to try to catch it with the corner of your net.

All else being equal, it is better to swing at a bee that is just flying into or away from a flower than a bee that is actually on a flower. Particularly if you are trying not to damage the plant, a less than vigorous swing of the net will simply push a bee clinging to a flower under the net and it will fly away afterwards. After some practice you can bring your net up to a bee on a flower, wait for the bee to just begin to leave the flower, push the flower out of the way with your net and still easily capture the bee.

When looking at a clump of flowers that could contain bees, stand 4-8 feet (1.2-2.4 meters) away. Most people stand too close to the flowers, which could scare away some of the bees you might be interested in, limit both the number of flowers (and therefore bees) in your field of view, and limit your depth of field. Standing further back permits you to view large expanses of flowers, spot a bee, and either lean forward or take one step to put that bee into your net. If you have to take two steps or more, you are too far away.

On any flower patch, concentrate on the difficult to obtain bees first. In particular, look for bees that are moving very quickly, from flower to flower, and try to predict where they will move next. Usually there is some pattern to the quickly-foraging or flower-visiting bee and often they will return to the area after making their circuit. Some of these individuals never really come to rest and you have to swing ahead of where you think you are going to catch them. It also pays to look below flower clumps for low-flying bees. Some of these are nest parasites, while others simply prefer to move between clumps of flower just above the ground or grass.

Open soil of any kind and, in particular, south-facing slopes, overturned root masses, clay banks, and piles of construction dirt or sand should be scanned both for bee nests and their inhabitants, as well as for low cruising nest parasites. Nest parasites (in particular *Nomada*) usually fly just above the soil in erratic flight paths. The best way to capture them is to slap the entire head of the net over the bee and quickly lift the net bag up while leaving the rim on the ground. The bee will fly upwards rather than trying to sneak under the rim. Often this can take several seconds, so patience should be applied.

There are two ways to catch multiple individuals in a net. One way is to turn your net head sideways after capturing a bee, allowing the net bag to close over the head and hoping that the bee will not find a way out. The other is to physically hold the bag closed above the tip containing the bees (note, in between swinging at bees, you will be holding the closed net against the pole as you carry it from place to place). In both cases you will have to periodically snap the contents of the net to the bottom. Do this vigorously or some wasps (in particular) may not go to the bottom, and, if you're not paying attention, you could end up grabbing them through the net with obvious consequences to your hand.

In general, it is easier to see bees through the mesh if you go into the shade or if you shade the net with your body. Some people favor green nets over the traditional white ones to reduce this phenomenon. See the above section regarding the "zip net" for an alternative.

Two videos that demonstrate how to use a net to collect bees can be viewed at:

http://www.youtube.com/watch?v=n6ZFlz3uA7E
https://www.youtube.com/watch?v=SwYbv5bySPQ

Removing Bees from the Net

Time spent removing bees from the net is time spent not capturing bees; therefore, think about how you are removing bees from your net to see if you can speed the process up.

In the beginning, there is usually a great fear of being stung by your subjects. In reality, in North America, only *Apis*, *Bombus*, Pompilidae, Polistinae, Vespinae, and perhaps a few of the other wasps have significant stings. These are large insects and can be readily discriminated. However, even these species almost never sting while caught in a net unless they are physically grabbed or trapped against the net. Thus, over time you should concentrate on diminishing your fears, and spend more time sticking your hand and kill jar directly into the net. If you are putting your net on the ground to remove bees, you are taking too much time. Kill jars should be fully charged to quickly kill your specimens, and it helps to have multiple jars.

The most efficient means of collecting large numbers of bees is to use vials or containers of soapy water. In that way you can fill your net with bees and empty the net only periodically rather than after catching an individual bee or small numbers of bees. However, cleaning and processing bees killed in liquids requires some care to do properly (see section on washing and drying bees), but can result in better looking bees than those collected in a poorly maintained container filled with Lepidopteran scales, nectar, pollen, and moisture.

Laurence Packer has gotten to the level where he simply uses his fingers on all bees except bumblebees; he gets stung, but says it's all very minor, unless he gets stung repeatedly on the same spot. Aspirators can also be used to remove minute bees (such as *Perdita*) if you only have traditional killing jars.

Once you have captured 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 the insects to the bottom. You can then safely grab the bag just above where they 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 kill jar and closing the lid loosely, but be aware that this is a sign of a rank beginner. Keeping your jars well charged with cyanide or ethyl acetate will ensure that the specimens quiet down quickly, and you will not waste a lot of time waiting. Once your specimens are immobilized, you can open up the net and drop them directly into the kill jar without worry.

Most collectors take a more direct approach and bring the open kill jar and its lid into the net, trapping the bee against the netting. Slapping the hand on top of the kill jar through the netting is at times useful to drive the bee to the bottom of the jar. This can help prevent bees from escaping when you put the cap on. 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 collectors have taken to hanging the net hoop on the top of their head. Use one hand to hold the net out and up, and then use 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 (duh!). Use small collecting jars, aspirators, or large test tubes that can be handled easily 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 in a net, if you don't trap them in your hands or against the netting. 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, such as handball gloves, welder gloves, latex dishwashing gloves (though stinging can occur through latex), and goatskin beekeeper gloves, but again this is the sign of a beginner.

A video that demonstrates how to remove bees from a net can be seen at:

http://www.youtube.com/watch?v=n6ZFlz3uA7E

Using Ice, Dry Ice, and CO_2

If it is important to keep bees alive or very fresh, bring a cooler of ice or dry ice and two nets. You can continuously collect bees with one net, and once it is full, 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 can then continue collecting with a second net. Once one net is full, the bees in the first net have already been chilled or have perished, and you can transfer them to jars in the cooler for further storage.

Denny Johnson uses a Planet Bike® CO_2 tire inflator (< $20.00) that uses Planet Bike 16g threaded CO_2 cartridges which can be adjusted for flow manually and shut off. He notes that the cartridges seem to leak out after about a day even if you don't use the entire contents. Cartridges cost proximately $1.00 each. To use, capture the bee in a small container like a pill bottle with a small hole drilled in it. Adjust the flow from the inflator so that it releases just a small amount and then insert the needle into the container. It takes only about 2-3 seconds for the bee to pass out but Denny usually lets them lie in the CO_2 atmosphere for a minute or so after shutting off the flow. It takes about 5 minutes or more for the bees to wake up and fly off. You have to experiment with this some for an optimum "out" time. He gets about 7 - 8 injections per cartridge. It may be possible to use paint ball CO_2 cylinders or beer tap cylinders which are better regulated, but would be less portable.

Catching Bees on Flowers with Baggies and Kill Jars

These systems are particularly useful when working with individual specimens on individual flowers. Pop the open end of large baggies 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 for preservation until taken back to the lab. Similarly, putting a kill jar over a flower and tilting the flower into the jar works to preserve the flower. However, the "zip net" modification mentioned above in the section on nets is superior to the above techniques.

Bee Vacuums

Converting a Leaf Blower (contributed by Julianna K. Tuell) – "Sam Droege asked me to send out a detailed description of the modified leaf blower that was used in Michigan to collect flower visitors, because it may be of interest to members of this listserv. 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® leaf blower 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 1 m^2 plot's flowers for 30 seconds and then while the leaf blower 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 four mesh bags.

Here is the link to the actual model leaf blower 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.K., and D.A. Landis. 2007. Attractiveness of Michigan native plants to arthropod natural enemies and herbivores. *Environmental Entomology* 36 (4):751-765.

Fiedler, A.K., and D.A. Landis. 2007. Plant characteristics associated with natural enemy abundance at Michigan native plants. *Environmental Entomology* 36(4):878-886.

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

> Tuell, J.K., A.K. Fiedler, D.A. Landis, and R. Isaacs. 2008. Visitation by wild and managed bees (Hymenoptera: Apoidea) to eastern U.S. native plants for use in conservation programs. *Environmental Entomology* 37(3): 707-718."

A Cordless Hand Vacuum Adapted for Collecting Bees (contributed by H. Glenn Hall) – [*Sam's note: The recommended model is no longer manufactured or sold, but the information is being provided as a guide for how other handheld vacuums might possibly be adapted.*] The bee vacuum is useful for collecting bees that have alighted on flowers (although airborne bees are occasionally caught that come close to the end of the vacuum), when minimizing flower damage is important, for example from gardens, nurseries, and some crops, and for catching bees on flowers under foliage. Bees can be transferred to tubes or vials more readily from the vacuum than from nets.

A description of the adapted vacuum:

The recommended vacuum is a Black+Decker® 18 Volt Platinum Series Cordless Hand Vac, Model # SPV1800 [This model is no longer manufactured or sold.] (Figure 1). It is a strong vacuum with a removable battery that is charged separately. With one or more extra charged batteries on hand, the vacuum does not become unavailable when it depletes the charge.

A Black+Decker® Overmold Adaptor for round tubes (a 1¼ " opening) is pushed onto the end of the vacuum (Figure 1). The adaptor comes from the floor extension kit of a different model Dustbuster®. The adaptor is no longer being manufactured but is still available for purchase (Part number 5102314-01. Phone: 1-888-678-7278. As of April 2015, the cost was $3.22 (limited quantities in stock).

Figure 1. Bee vacuum.

Three sections of acrylic tubes of decreasing diameter are added. A 4" to 6" long, 1¼ " OD, 1" ID, piece is slid into the adaptor opening, followed by a 4" to 6" long, 1" OD, ¾" ID, piece, and finally a 3" long, ¾" OD, ½" ID, piece (with a screen on the inside end [see next paragraph]). Each tube is pushed about ½" into the adaptor or tube before it (Figure 1). The fits should be tight between the adaptor and first tube and between the first and second tube. If the inside tube is slightly too large, some sanding may be needed at the edges. If it is too narrow and loose, it may need to be glued. The fit between the last two pieces of tubing should be snug, enough to hold the last tube in place, but not so tight that it cannot be easily removed. These lengths of the tubes increase the reach of the vacuum, without making the entire length too awkward.

A disk of stainless steel screen is cut (as close to ¾"diameter as possible) and glued (epoxy is recommended) to one end of the last tube (Figure 2). Screen material is used that has a mesh small enough to catch small bees and thin wires that do not greatly restrict air flow. The screen can be purchased from McMaster-Carr (High-Volume Lightweight-Particle-Filtering Stainless Steel Wire Cloth Woven, 316 stainless steel, 22 x 22 mesh, 0.0075" wire diameter, product number 9230T51, sold as a 12" square piece at $5.21). After the screen is glued to the end of the tube, the overlapping edge is filed off (file strokes toward the tube are best, as they tend not to pull up the screen). It may be tempting to use a nylon fabric mesh rather than metal screen, because it is easier to remove the overlapping edge. However, some bees, particularly *Megachile*, easily cut through nylon. It is useful to have several of these end tubes made.

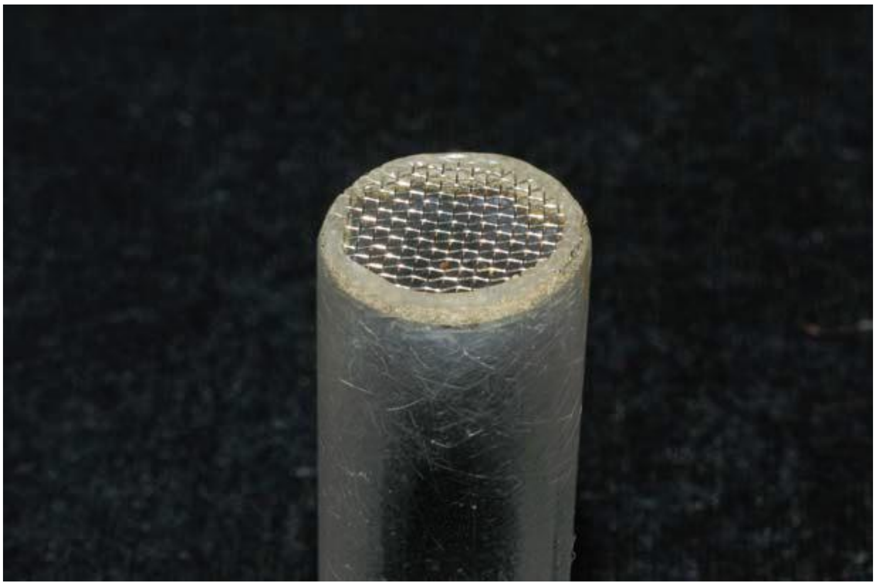


Figure 2. Close-up view of end of collecting tube.

Tips for collecting bees with the vacuum:

When a bee is caught, the open end of the last tube is covered with a finger, before turning off the vacuum. Several bees can be caught in succession, if, during the waits in between, the end is kept closed while the vacuum is kept off (to save power). The end tube is removed and the bees are transferred to a holding tube/bag or killing tube/jar. Alternately, if several end tubes have been made and are available, they can be closed with a cork to hold the bees until they can be transferred at a later time.

Bees cannot be collected off of flowers with big floppy petals, such as squash flowers, which tend to get caught in the vacuum. The big petals of sunflowers are avoided by taking good aim at the bees on the flower disk. To catch bees under foliage and avoid catching leaves, the open end of the vacuum tube may need to be quickly moved close to the flowers before the vacuum is turned on. Sometimes the bees hit the screen with such force that they appear stunned but not damaged.

A Smaller, Pocket-sized Bee Vacuum (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® (MCV 2000) for vacuuming up small bees and other insects off 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!)."

A Small Pocket-sized Bee Vacuum

David Almquist provided an additional modification to the Dirt Devil that seems to increase their ability to hold specimens following capture. He painted the clear parts of the Dirt Devil black and took out some of the internal structure so that the bees were more likely to move to the back which is now the lightest part of the device and thus became easier to remove and place bees into a vial.

<u>Plexiglas Bee Observer and Pollen Picker</u>

Brian Dykstra wanted to catch, photograph, and release bees, wasps, and other insects for identification following net captures in the field without killing them. With help from a colleague he created a Plexiglas container with a slide top and a foam plunger (see picture below). This device is similar to the queen marking cage and plunger used by some in honey bee keeping practices, except this has a square shape and a clear top for ease in quality photography without distortion. This one also has an optional screen slide for the top for collecting pollen with toothpicks. This is an excellent tool for citizen science and school groups.

Plexiglas Bee Observer and Pollen Picker

Bees through Binoculars

For those investigators who do observations of bees on flowers or around nest sites, the Pentax Papilio II 8.5x21 binocular is ideal. It has high magnification and focuses down to 0.5 m (1.6 ft), permitting sight identifications and detailed behavioral observations (once you have learned to identify specimens under the microscope).

Kill Jars

Several companies make chemical based kill jars that use either ethyl acetate or potassium cyanide as the killing agent. There are advantages and disadvantages to both types.

Ethyl acetate – Traditional jars are made of glass with a layer of plaster of Paris at the bottom. BioQuip now makes a plastic kill jar that has plaster of Paris attached to the lid instead of at the bottom of the jar. This helps prevent specimens from coming in contact with the ethyl acetate directly if one can manage to keep the jar upright. At the start of the collecting day, pour enough ethyl acetate into the jar so that it soaks into the plaster, but leaves no liquid on top. If you use the jar regularly, then the ethyl acetate will need to be recharged every couple of hours, as it will evaporate. The advantages of using ethyl acetate are: less toxic than potassium cyanide, not a controlled substance, and relaxes the specimen, which is useful if the genitalia are being pulled. The disadvantages are: needs to be replenished often (requiring either that ethyl acetate be brought into the field or that several charged kill jars remain available), can cause the jar to "sweat" inside which may mat a specimen's hairs, significantly degrades DNA, and will outgas in a hot car which is probably not good for you.

Potassium cyanide – 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. Place a layer of cyanide crystals in the bottom of the container. Next add a layer of sawdust. Finally, pour wet plaster of Paris over the sawdust. Leave the jars open for a few hours outside or in a hood, and then close them. Alternatively, a combination of cotton balls and tightly rolled paper towels can be used in place of the plaster and sawdust. The advantages of using potassium cyanide are: knocks down insects quickly, does not significantly degrade DNA, can remain effective for over a year, and does not add moisture to the jar. The disadvantages are: is relatively toxic, is a controlled substance, and can change the color of some bees (particularly yellows become orange or reddish), if bees are left too long in the jar.

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 different plant species. Tubes can also be handled easily with one hand while in the net. Vests, aprons, hip packs, and carpenter belts are useful 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 breakage 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 and regurgitated nectar from the bees collected.

After bees have been placed into a well charged kill jar, they usually quiet down in just a few seconds. If the specimens are taken out of the jar too soon, some may "wake" back up and begin to move again, albeit usually only very slowly. Usually thirty minutes or so in the kill jar will prevent this.

Soap Water Jar or Tube – An alternative to chemical based kill jars are containers filled with soapy water (a mix of water with any common dishwashing detergent) 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 and are large enough to hold a fair number of bees. They should fit in your pants pocket and be easy to hold in one hand along with the lid. Fill the vial about half full with soapy water.

The jar will form a constant head of suds while riding around in your pants pocket. Using it in the net 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 chemical based (cyanide, ethyl acetate) kill 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. At BIML, we favor using the large centrifuge tubes, as they slip into the pocket easily.

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 also easily be used to directly collect off of flowers without a net.

Specimens can be readily left in the soapy water for 24 hours and, while a bit soggy, will even last for 48 hours without too much degradation. Afterwards, specimens can be either dried and pinned, drained and put into alcohol for long-term storage, or drained, wrapped with a piece of cloth (to soak up excess moisture and to prevent breakage) and frozen in a plastic bag. Specimens look best if cleaned and dried within 24 hours of capture in bowls or soapy water, if cleaned immediately after capture some specimens can "wake-up." However, this can readily be checked by freezing any specimens that do begin moving.

The advantages of the soap jar are:

- 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
- Inexpensive

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 (see section on properly drying specimens below)

<u>Chlorocresol Humidor</u>

(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 to 1year 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:

- A pint or quart plastic container with a tight seal (I use a 4-cup or 1-quart Ziploc® Twist 'n Loc® container, but I have used on occasion up to ½-gallon containers);
- Paper towels;
- Chlorocresol (an antifungal crystalline substance with a sugar-like consistency available from BioQuip - item # 1182B - $18.95/100 grams) (chemically = p-chloro-m-cresol or 4-chloro-3-methylphenol);
- A few strips of duct tape or its equivalent; and
- 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, chlorocresol "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 Arduser). 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 Apinae and Panurginae) 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."

<u>Pinning 101</u>

Types of Insect Pins to Use – Bees are usually pinned using pin sizes 1-3, with size 2 being 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, do not 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 bumble bees. 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. Insect pins can be expensive. The cheapest way to purchase them is to order in bulk directly from Czechoslovakia, where apparently most 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.

Traditional Pinning Techniques – 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 regurgitant, wash and dry them using the protocols listed in this manual. They will result in better looking, 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 his or her 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. Use another finger from the hand holding the pin to help hold the specimen steady while 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.

Specimens are best pinned through the scutum between the tegula and the mid-line. The midline of the scutum often contains characters that are very useful in identification, which can be destroyed by a pin. Most museums prefer that specimens be pinned on the right side.

For someone new to pinning, use of a purchased insect pinning block is helpful to determine the correct height a specimen should be placed. With experience, one can use pieces of foam of the correct depth, or even adjust specimen height by eye, which will be the quickest. Remember to leave enough room at the top of the pin so that the specimen can be safely picked up by the largest of fingers. Equally important, leave enough room at the bottom for two or more labels and room for the pin to go into the foam of a collection box.

A video that demonstrates how to pin bees can be viewed at:

https://www.youtube.com/watch?v=V2F8LBQV5L0.

Gluing Small Specimens – If specimens are too small to be pinned, they can be placed on a point, glued to the side of a pin, or attached as minuten double mounts. Reversible glues, such as Elmer's Glue Gel, white glues, tacky glue, clear nail polish, shellac, hide glue, and others should be used.

Gluing to Points – The use of points is traditional. Points are very small, acute triangles cut from stiff paper using a special punch, which can be ordered from entomological supply houses. Place the pin through the base of the point. Elevate the point on the pin to the same height as a pinned specimen. Glue the small bee to the tip of the point, usually on its underside.

Gluing Directly to Pins – When gluing a specimen directly to a pin, rather than to a point, the specimen is glued on its side or the underside between the thorax and abdomen. Again, most museums prefer that specimens be glued on the right side. 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. Its axis of rotation is minimized and the paper 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 traditionally pinned.

In the past, we have used white, tacky glue in our lab. This is a thick glue which sets up within seconds. It allows the glued specimen to be set upright in a box immediately (unless extremely large), without the danger of it losing its placement on the pin. From our limited investigations, Aleene's® Original Tacky Glue® in the gold bottle or archival paper glue appears to be the best gripping, tacky glue.

We now like to use glue gels when pinning bees. Glue gels have a longer work time, dry crystal clear and are easily reversible. Because the set-up time is longer than tacky glue, leave the pin resting on the specimen on your pinning board or tray for at least 5-10 minutes prior to picking it up. Parchment paper is very helpful to have around when gluing bees. It is a silicone impregnated piece of paper that can withstand the heat of an oven, but is super slick. It provides a "non-stick, Teflon®-like" substrate on which to work, because glue does not adhere well to it. Another nice thing about parchment paper is that dried specimens can be easily re-positioned. They will slide smoothly on the paper without sticking or breaking. We now dump dried specimens onto the paper and pull up the sides of the paper, which causes the specimens to slide into the center.

Once the specimens are in a line in the center, sorting out the non-bees from bees is rapid as you can pull off a few specimens at a time to sort or pin from the bottom of the line of specimens. Run a small line of glue along your thumb or forefinger on the hand that you do not use to pick up the pins. Touch a pin to the line of glue at the height at which you want the specimen to be glued. Touch pins with a small amount of glue at the proper height gently onto the specimens that are lying on the parchment sheet. Be sure that the glue is adhering to the side or underside of the bee and not to just the hairs, legs, or wings. Pick up the specimen and the pin and move it to one side of the sheet for further drying. After the glue is set, press the pointed tip of the pin with your finger. This will cause the specimen to rise up, allowing you to grasp the top of the pin and move it into a collection box.

A video that demonstrates how to glue a bee to a pin can be viewed at:

https://www.youtube.com/watch?v=9KfLCmYOKtA.

Current BIML Techniques – While unorthodox, our current process for pinning involves: washing and drying specimens in the machines listed in this document and placing them in open, labeled Petri dishes. If time doesn't permit pinning right away, after a week or so of drying, the Petri dish cover is replaced and taped on, and the specimens are stored in their dishes with labels.

When ready to pin, all the specimens are laid out on a large foam pinning board covered with parchment paper and a pin is glued to the side or underside of each (including the largest specimens) using glue gels. 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 two of them) is handy to have on the wrist holder, as the threads will separate the pins for easier pick up. We then run a small line of glue on the side of our thumb or forefinger (Thank you Harold Ikerd for this idea) on the same hand that has the wrist holder.

A reverse set of tweezers is used to pick up a pin by the head or the tip (or you can use your fingers). 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. The pinned specimen is left on the pinning board in a line on the left or right side until the glue sets (usually about half an hour). With a little practice, it is easy to achieve pinning rates of 250+ per hour. None of these gizmos are necessary to glue bees quickly; fingers work nicely without tweezers, glue can be spread directly from the bottle, pinning boards can be stacked using small bowls as spacers, and pins are very convenient if stuck into the foam.

After the glue has dried, pins are then transferred to boxes. In some instances, that transfer can be combined efficiently with the attachment of labels, saving another step. Jane Whitaker has found that magnetizing her tweezers helps in picking up glued specimens on pins.

Minuten Double Mounts – 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 on a regular insect pin. 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 are available at:

http://nau.edu/Merriam-Powell/Biodiversity-Center/Museum-of-Arthropod-Biodiversity/Instructional-Videos/

Bee Storage Boxes – There are a variety of drawers, cabinets, and boxes available to hold specimens. We prefer to use the simple cardboard specimen box made from a pizza box with a completely detachable lid, and a crosslinked polystyrene bottom for housing everything except our synoptic collections. These boxes are stackable, the date and location can be written on the outside in pencil and then erased when reused (or organized with Post-it® notes), are relatively inexpensive, and, unlike hinged lid boxes, are convenient to use in cramped spaces on a desk or worktable. Such boxes are made from scratch. Instructions for making "pizza" insect pinning boxes can be found in this document.

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 original tag that was placed in a batch of specimens when first captured. Pin a line of specimens to the right of the tag, and continue adding insects from top to bottom, left to right, like a book, until complete. The next tag is placed immediately thereafter and so forth until the box is approximately half filled (this allows for plenty of workspace later when identifying bees). In general, it helps if each box contains specimens from only one region. On the outside 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 from a stack.

Control of Pests – Simple cardboard boxes are not pest proof. Dermestid beetles are the primary pest of insect collections. Fortunately, infestations are usually small. An infected specimen is usually easy to spot, as small black powder and shed skins are visible below the specimen. Control and prevention take place, according to the literature, by freezing the box at -20 C (~ 0 Fahrenheit) for three days, thawing for a day, and then freezing for another three. 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 small 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 in non-air-conditioned spaces, particularly those just caught, can turn into balls of mold. Either move 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 Discover Life web site. Each batch or site is given a unique site number and each specimen is 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 in the box. These data matrices are included automatically in the free Discover Life system (http://www.discoverlife.org/label/) or can be added using commercial software such as BarTender (http://www.seagullscientific.com/). Many a beginning student of bees has rued the day that they did not give their specimens unique numbers.

Dan Kjar has generalized the Discover Life label program so it will print out on a laser printer. You can use his simple web based form (http://bio2.elmira.edu/fieldbio/) following the link at the bottom of the page for insect labels. Each label is unique based on the specimen number.

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-pound paper is good label stock.

Specimen labels are quickly added to specimen pins by laying them across a piece of Ethafoam - the thickness of which is the desired height of the label on the pin. To increase the durability of the Ethafoam, glue it to a piece of plywood or corrugated piece of plastic like those found in outdoor signs, which will 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. 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 band or table 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.

Cutting out labels can be a time consuming aspect of any project. We speed up the process by cutting out rows of labels; placing them in their box and then cutting the individual labels apart with scissors. See:

http://www.slideshare.net/sdroege/preparing-insect-labels-a-faster-way and http://youtu.be/HqxrkC6xe40. Ray Geroff uses a surgical/dissection scalpel and handle. He prefers the #4 handle with a #21 or #22 blade. It works well for cutting the strips, but works really well when cutting the individual labels apart once they are in single strips.

Making labels in Microsoft® Word (Contributed by Gretchen LeBuhn) – Open up a new Word document and just type the label as 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, make the bees collected at a single species of plant an individual collection record. For example, bees collected on *Vicia sativa* at Rector Dam are collection #1 and those collected on *Lupinus bicolor* are collection # 2. Keep this system going or some similar system so that you can identify and talk about each collection separately each year. You can 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 in one long continuous roll. When I am finished, I put it into column format to fit more per page.

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

The above was for 3 bees collected in Collection #1. Leave a blank line between collection events to see where each collection event starts.

3) Click "Edit"… select "select all".
Click "Format"… select "Font"… type into the "Size" window the number 3 (for 3 point font) and click okay.

Click "Format"… select "Paragraph"… select under "Line Spacing" the word "Exactly"… under "At", select "3 pt." (this sets the leading or space between lines)
Click "Format" … select "Columns"… under "Number of Columns" start with 8… under "Width and Spacing" set the "Space" (that is space between columns) to 0.00. 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, and other times 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.

Making Labels Using Microsoft® Word's Mail Merge – Microsoft® Word's Mail Merge feature is used by a number of groups who make their own labels to increase the efficiency of generating specimen labels. In general the way that Mail Merge works in label making is that a Word document is created with collection information (Location, Latitude, Longitude, Date, Collector, etc.) and associated with an Excel or Access file that has numbering information, or, alternatively the Excel or Access file could have ALL the Location, etc. information, and you simply use the Word document to do the collating and printing of the labels. It is possible to simply use features in Excel or Access to create labels, but often people are more comfortable using Word, there are many ways to do this. The numbering system used could be a single unique number for every specimen or a separate number for the collection event along with a number for individual specimen. We recommend that both the collection event number and the specimen number be completely unique and not repeated in any of your collections. This will minimize errors where numbers are inadvertently used more than once. When experimenting with your labels we suggest you start by looking at a font size around 4 and to not use fonts that have serifs since you will be printing very tiny letters. Additionally you will want to make sure that your printer is printing at is maximum resolution (given as dpi) so that your tiny labels are as visible as possible.

Determination Labels – These labels are used to write the species name along with the person who did the identification (the determiner). You can email Sam Droege (sdroege@usgs.gov) for Excel spreadsheets that will print out blank determination labels 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 modern technical pens, as Rapidographs tend to clog when left unused for any length of time. Technical pens in sizes 01 and 005 are the best and are available from art and 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 be freely 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 without using a guide. As new species are detected, a determination label is created (available as a modifiable Microsoft Excel file from us). The determination label is pinned to the board separately from the specimens, so that it can be easily viewed when entering the data. All subsequent specimens of that species are then placed to the right of the determination label. Bees that cannot be immediately identified are kept separate and identified at the end using computer and paper guides. If you have a large number of unidentified species, then morpho-sorting them to species or species groups is a big timesaver. In general, it is best not to struggle with the identification of any individual specimen, but set it aside and return to it after you have looked at the other specimens and much of the time you will find that the identification of that specimen was partially resolved by looking at the other specimens.

Within a box, bees are placed in the box in loose 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 to check the label and saves time for the person who has to do the original labeling.

<u>Entering Specimen Data</u>

In the system that we use, each specimen has a scannable data matrix on its label. Data entry consists of scanning each specimen directly from the box into an Microsoft 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. 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 that 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 with the database entries. After that final check, pins with tiny squares of colored paper are interspersed into the box designate which bees should be dispersed to final resting spots in museums, sent to other colleagues, or their pins are recycled for reuse.

<u>Shipping Pinned Specimens</u>

The box you ship bees in should have the specimens firmly pinned into the foam so that they do not come loose during shipping and destroy other specimens. Cut a piece of cardboard that will fit snuggly inside of the box and rest that cardboard on top of the specimens. (Do not use foam for this layer as it can engulf the tops of the pins and cause problems when removed.) Place either pinned specimens or empty pins in all four corners of the box to support the cardboard. 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. Two pieces of tape can be affixed to the top of the cardboard in such a way as to 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 is space between the top of the cardboard and the lid of the box, put in some bubble wrap or packing peanuts there, 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 or rubber band 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 United States, 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.

<u>Microscopes</u>

When using bowls or nets, it is easy to quickly amass a large collection of bee specimens. Unfortunately, unlike most butterflies, bees (even the bumble bees) 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, are difficult to set to fixed heights, and their stands are usually lightweight and often designed in such a way that makes 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 onetime 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 eBay or Craigslist is more risky as the seller has less of a reputation to risk). There are many used microscope sites; 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. Of special consideration are the Bausch & 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 the zoom magnification is on the top, rather than on the side. 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.

What follows is a list of microscopes recommended by other bee researchers and amateurs. They are listed alphabetically and include high, middle, and low end scopes.

Bausch & Lomb StereoZoom 5 – $150 used (these are the standard college student scopes of the past)
Leica 2000 – $850
Leica EZ4 – $820 to $1150 (several people responded that they use this line)
Leica MZ12.5 – $6,000 - 8,000
Leica S6E – $1100
Leica S8 APO – $3400
Meiji EMZ-5TR body – $2000 (10 years ago)
Olympus SZ60 zoom
Olympus SZ61 with an aftermarket ring-light – $2,000 - $2,400 range
Olympus SZX12
Olympus SZX16 – $6,000 - 8,000
Omano Stereoscope OM9949 – <$1,000
Wild M3Z – $1500 used
Wild M8 – $1500 used
Zeiss Stemi DV4 – ~$2000

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, 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 are readily removed (as some of you who have turned a microscope upside down have found out). However, 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 that 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. As a practice of work, the magnification is often 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. Note that for simple measurements of total body length, it is easier just to have a ruler handy that you can lay your specimen next to.

Adjusting, Cleaning, and Storing Microscopes – Most good scopes are fairly sturdy and don't go out of adjustment without suffering 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 the images the oculars are processing are out of alignment. If the images do not completely align no matter how much you play with the width of adjustment of the eyepieces, the scope probably 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, the 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 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 happens, after adjusting in both directions you still cannot get the eyepiece in focus, try sliding the eyepiece up slightly. If that doesn't work, it is 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, you will have to work out some other mechanical means. Usually, however, such an extreme situation indicates that something is generally wrong with the scope or the oculars. You might check the oculars to see if a lens is loose or if you have mismatched oculars from some other scope.

The objective lens of a microscope almost never needs to be cleaned. However, the top lenses of the oculars often do, particularly if the person using the scope likes to press his or her eyes close and wears make-up (mascara is the worst). We use lens paper and window cleaner as needed. When the scope is not in use, put a microscope cover or a large baggie over both ocular lenses to keep the dust out.

Charlie Guevara reports a clever way to modify an iPhone into a field microscope at:

http://hacknmod.com/hack/turn-an-iphone-into-a-microscope-for-10/

Holding Specimens and General Microscope Setup - Most people when viewing specimens under the microscope, place them on a piece of clay, foam, cork, or some sort of stand. We 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 held between the thumb and forefinger allowing you full 360 degree rotation.

It is important to place the bottom sides of 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, 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 provided valuable additions to this section.

Ping Pong Ball/Plaster of Paris Specimen Holder (Contributed by Gary Alpert) – You place the plaster-filled ping pong ball in a large heavy washer of some sort and like a track ball you can swivel it around to get the best look at your bee. While we are not fans of using platforms to indentify bees, this ping-pong ball stand is useful for some circumstances and beats all other stands hands down.

General steps:

1. Buy a ping pong ball.
2. Drill small hole in said ball.
3. Mix a fairly liquid batch of plaster of Paris.
4. Quickly transfer plaster of Paris to ping pong ball using a syringe or eye dropper.
5. Wash equipment immediately.
6. Wait for ping pong ball to dry.
7. Drill a small hole in plaster.
8. Plug hole with clay.
9. Optional: paint photographer gray.
10. Set ball in washer.
11. Put specimen in clay.
12. Pivot as desired.

The Bee Bowl Trap

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 traps) and 12 oz. salad bowls were used. Field experience and experiments have demonstrated that bowl size is 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 unpublished experiments on such).

Several manufacturers make such cups, but Solo® is the line that most people have experience with. These cups are usually translucent, which is not at all attractive to bees. However, the 3.25-oz. Solo Polystyrene Plastic Soufflé Portion Cup is steep-sided, stable on the ground and does come in white (model number: P325W-0007). This particular model works well because plain white is highly attractive to bees and it also provides a nice base color when painting fluorescent blue or fluorescent yellow.

For travel, the Solo 0.75-oz. and 2-oz. cups are nice sizes to carry in your luggage as they minimize water use. However, they will lose water very quickly in hot, low humidity environments. 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 2500 cups). Translucent models 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 http://www.solocup.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. Denny Johnson located a source at http://www.cometsupply.com/ that, as of the writing of this version of the manual, is still available. See the bottom of the next section for a source of pre-painted bowls.

Painting Bowls – Prior to using soufflé cups, 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 bowls (red bowls attracted absolutely zero bees in the East). He didn't compare these with fluorescent colors, but both of these colors collected more *M. texana* than did either white or yellow. Note that fluorescent colors are not reflecting ultraviolet colors (which we cannot see) but are translating ultraviolet reflectance into visible reflectance and thus creating "brighter" colored bowls. 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.

Guerra Paint and Pigment – Commercial fluorescent spray and brush paints vary in their color characteristics and availability by brand and location. In 2004, we experimented with some different formulations and found a fluorescent combination from Guerra Paint and Pigment that works better than the system we had tried earlier. The liquid pigments mix much more readily than the dry pigments and their base paint 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”. Run the scroll bar down to the bottom and click on “Fluorescent.” Choose “Fluorescent Blue” or Fluorescent Yellow” in 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 ounces of pigment to 1 gallon of Silica Flat binder. 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%
• Fluorescent 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.

Pre-painted Fluorescent Blue or Yellow 3.25 ounce Soufflé Cups – You can purchase pre-painted fluorescent blue or fluorescent yellow 3.25 ounce soufflé cups from New Horizons Support Services (http://www.nhssi.org/) which uses developmentally disabled workers to paint the bowls. They also sell unpainted white bowls in any quantity desired, which can be useful for smaller projects. Email your queries to Cynthia Swift-King (cking@nhssi.org) or call the number at the web site listed above.

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 become 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. 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 ColorMaster™ Plastic Primer. 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 fluorescent blue. Sanding the bowls also allows paint to adhere better, but this 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 one another, the completely-painted bowl caught significantly more bees (about 50% more). 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). Hannah Gaines has informed us that ACE Hardware fluorescent spray paints are manufactured by Krylon – opening up additional possibilities. One issue people have had with spray paints appears to be loss of longevity of the plastic. Several have noted that the bottom of the bowls drop out. Best guess at this point is that this may be due to the paint being too heavily applied and the solvents degrading the plastics; no such problem has been noted with latex paints and bowls have been left outside for many trapping intervals with only gradual fading and brittleness. In 2010, Tracy Zarrillo noted that the "Can -Gun® Spray Handle" from Ace Hardware greatly increases the ease and consistency of using spray paint and only costs about $5.00.

In general, yellow colors fade quickly compared to blue colors. Over time, plastic bowls become brittle and need to be replaced, however, they can last for several rounds of painting and several months of exposure to the sun.

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 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, it is best to place specimens in alcohol for several hours, or put them in the freezer prior to pinning. Unpinned insects that do begin to move after being in a bowl never regain full functionality and should be put into the freezer overnight.

We have found that the amount of water in a bowl does not affect the capture probability. However, in hot and arid climates, bowls can dry out, sometimes within a day, if not completely filled, or if the bowl is too shallow. We suggest that people use Dawn Ultra® blue dishwashing liquid for a surfactant. It is 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 and 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 carrying jugs in a vehicle or a backpack, Gene Scarpulla recommends using 96 fl oz (2.8 L) Lactaid® milk jugs; they are thicker-walled and stronger than regular gallon milk jugs that tend to split easily.

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. Glycol seems to favor larger rather than smaller bees, so be sure to add detergent to the soap to decrease surface tension.

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 found at veterinarian supply houses (mostly online), RV centers, swimming pool, auto and livestock supply stores, and heating and cooling supply houses. Heating and cooling suppliers have glycol with a few additives, usually come only in blue, but are mostly not diluted with water (which evaporates). RV and swimming pool glycol is usually red (the red can be eliminated by adding a tablespoon or two of household bleach), and are diluted to some unknown extent with water and thus will need to be recharged. Veterinarians use food grade propylene glycol that is not diluted and is readily available online. It is more expensive, but would be the best to use. You can also order large drums of propylene glycol directly with no added colorants. One common supply company for the basic material is ComStar International (http://www.comstarproducts.com/) or Bulk Apothecary (http://www.bulkapothecary.com/).

If you have problems with animals getting into your propylene glycol (Note: To date, most people have found propylene glycol not to be of interest to mammals.) you might want to add denatonium benzoate, a bittering agent used in antifreeze to keep animals away. It is EXTREMLY bitter and so you should wear gloves when using it or you will end up tasting it in your food and what you drink (handling this chemical can make you realize how easily chemicals can migrate from your hands to your mouth). One of the suppliers mentioned that a good starting point is 30-50 ppm; which seems to correspond to a healthy pinch per gallon of liquid.

Tracy Zarrillo and Kim Stoner use quinine sulfate (a common fish medication sold in pet supply stores) in their glycol traps to prevent animal disturbance. Most of the time disturbance to glycol traps comes not from animals that are trying to drink the liquid or eat the contents, but from animals attracted to the traps themselves, perhaps simply to chew on.

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 by 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 greatly reduced in the spring if temperatures are in the 50s F, or below, during the day. 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 where bees are likely to see them (e.g., fields, roadsides, grassy areas, barrens, sand, etc.). 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 much taller than the traps (e.g., creosote bush, mesquite, a field of blooming mustard) often appears to lead to low bowl capture rates. All that said, it has been the experience of many that small openings, rabbit paths, trails, open tree canopies etc. can be places where you will find bees, so experiment even if the habitat is not completely open.

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, crabronids, pompilids, vespids, skippers, thrips, flies, and other things that often come to flowers.

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, perhaps simply due to height of forging differences.

Most researchers put bowls out in strings or transects 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. A study by Leo Shapiro demonstrated that the variances for characterizing the species richness of a single site levels out around 15-30 bowls.

Bowls placed immediately adjacent to one another have been shown to have reduced individual per bowl capture rates. Studies in Maryland using three separate trapping webs in open fields showed a distance of 3-4 meters to be the threshold below which bowls competed with one another for capture. They did not compete above that level. In Brazil, additional species were captured when bowls were elevated off the ground. In the Eastern United States, no additional species were captured in elevated bowls and actually, 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; however, adding small *Andrena*-sized markings to a bowl did not change capture rates.

How to Collect the Bees Once Trapped – At each bowl, it is best to remove all moths, butterflies, skippers, 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. It is very important to choose a strainer with extremely fine mesh in order to retain 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 Whirl-Pak® plastic bags. We usually use a plastic spoon to gather the specimens from the brine shrimp net and then transfer them to the Whirl-Pak. 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 Whirl-Pak. However, Frank Parker uses a larger sized

Whirl-Pak 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.

Recently we have started shifting to the use of disposable cone shaped paint strainers (thanks to a suggestion by Jim LaBonte) used by commercial painters. The easiest way to find these strainers is to Google the search string "disposable paint strainer" and look at the images. These filters are nice in that they can be taken out in the field, labeled directly in pencil, the strainer placed in a funnel (for support), and when finished straining they can be folded, stapled and frozen in a Ziploc bag or they can be folded and placed in Whirl-Pak with alcohol. When purchasing paint strainers use the largest mesh size available as the very fine mesh sizes tend to clog quickly. An alternative to paint strainers used by several researchers are coffee filters (Thanks Tracy Zarrillo, Nick Stewart, et al.).

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 pint bottles of ethyl alcohol, ethanol, or denatured alcohol (be aware that alcohol names are not consistent). If not readily available in the store, it is possible to have the pharmacy order what you want. Hardware stores carry gallon and pint cans of denatured alcohol. We find that drug store alcohol is easier to work with, as it is made with a smaller amount of methanol.

Often 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. 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. However, for highest-quality DNA extraction from specimens, they should be stored in 95-100% ethyl alcohol.

The process for washing bees after they have been stored 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 or with the spoon/screen combination mentioned below and placed into 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 are readily identifiable to species.

Another alternative to Whirl-Paks is to dump the catch into small numbered squares of cloth which are rolled up and rubber banded together. Once back from the field, put them into Ziploc baggies and freeze until you are ready to pin.

Each bag, fishing box, or cloth should have a tag inside 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 Whirl-Pak bag, as they inevitably get wet with alcohol or water and the writing will run.

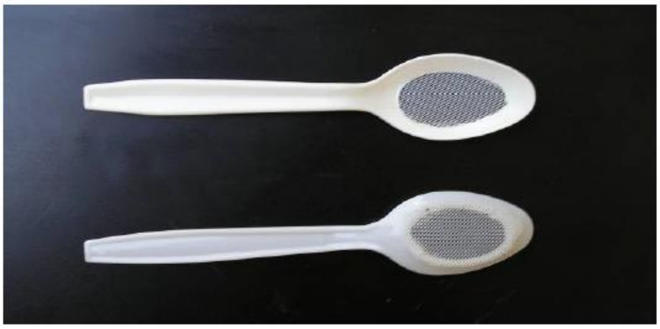
The following figure was clipped from *The Volunteer Monitor* 20(2):11, Fall 2009, and should be handy for removing individual bees from individual bowls.

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 - We have found that it is helpful to create your sets of bowls the day before setting them out. In particular, it is very handy to have an empty, divided flat, like those found holding plant starts at your local nursery, as this holds the separate sets of bowls quite nicely. Wire flags (very useful for re-finding 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. While getting out, drivers 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 using GPS as you put out the transect 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. 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.

<u>Glycol Pan/Cup/Bowl Traps</u>

(Information Provided by Dave Smith – FWS) – As part of a native pollinator inventory, I have been looking for a reliable pan trap 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, fluorescent yellow, or left white, is attached to ½-half inch PVC pipe with a 5-inch hoop cut from a plastic culvert pipe (see photo below). 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 3 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 an automobile oil funnel to dump the sample into a labeled Whirl-Pak. Funnels with a 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.

The BIML lab has taken the above technique and modified it slightly. You can view a YouTube video for how to deploy these at:

http://youtu.be/z0DAY7bNOR4

and you can view how to make the stands at:

http://youtu.be/x87CXM7mq54

and you can read a pilot report on using these in long-term monitoring at:

ftp://ftpext.usgs.gov/pub/er/md/laurel/Droege/Draft%20USFS%20Glycol%20Report%2022711.docx.

Glycol traps have the following advantages:

- They catch bees continuously, thus circumventing problems of shifts in phenology from year to year.
- Once deployed, they are easy to tend and the times for tending the traps can be scheduled rain or shine.
- They can be associated with weather stations where other devices are also tended regularly.
- They provide a continuous record of bees in the area.

Propylene Glycol Cup Trap

Flower Traps

Alex Surcică, has developed a modified bowl trap for squash bees that holds promise for capturing other crop and flower specific species. He writes: "I'm interested in monitoring bees in the *Cucurbita* fields with the bee bowl trap method. This summer I've used the 3.5-oonce blue, yellow, and white cups and had little or no success in trapping squash bees. It looks like the bee bowls cannot successfully compete with *Cucurbita* flowers in attracting bees. Therefore, I thought submerging flowers in cups with soapy water would yield better results. Because of the size of the flowers, I used the bottom end of a one-gallon milk jug. The results were great. In a field with a high squash bee population I got more than two dozen males and a couple of female squash bees in less than two hours (photo below). I would like to know if this method works for any of you that have *Cucurbita*-related projects. I'm also wondering if submerging the flowers in soapy water would work in monitoring bees for other crops. When trying this method, you should use the least amount of detergent, since high concentrations would cause flowers to lose their colors very fast."

***Cucurbita* Flower Trap**

Trap Holders

Alex Surcică has developed a nice adjustable trap design – "A screw (it can be seen in the upper left photo, although it is blurry) allows me to easily make the proper height adjustment for each cross along the rebar. Although it might not be pertinent for mass bee trapping, this system presents the following advantages: 1) allows one to put traps in places where the vegetation is dense and high, while still making the bowls visible to bees; 2) there is less chance for the bowls to be overturned by wind or wildlife; 3) saves some time in measuring and searching for the flat and visible spots where bowls can be placed; 4) requires a little less bending over; 5) the bowls can be placed as high as 3 feet above the ground – this is based on a 4-foot long rebar, with one foot being in the ground; 6) the traps are relatively cheap (less than $1 per 4-bowl trap holder); 7) are easy to install and take apart; and 8) can be used over several years (the bowls might need to be replaced).

Bowl Trap Holder

Similarly Zak Gezon has developed a trap for wetlands. – He writes: "I am catching bees in a seasonal marsh in Costa Rica. I had to come up with something similar to Alex's to solve a slightly different problem. When I started sampling the marsh was bone dry, but before long the water level started to rise, so I made PVC platforms for the bee bowls, as you can see in the attached photos. The platform itself is made of fine mesh so that if a sudden rain overflows the bowls, anything caught up until that point will be caught in the mesh. When the marsh water level is really low (and therefore the platform is high above the water surface), the wind can whip the platforms a bit, which is a problem, so I have been thinking about drilling the T-joint all the way through so the platform would slide down the PVC pole, and adding a wing nut to the platform so that the height would be easily adjustable. I added Velcro® to the platform mesh and to the bottom of the bee bowls to ensure I don't lose any bowls due to wind. One problem I have had is that sometimes birds land on the platforms and slosh all the soapy water out. I don't have a solution to that problem yet, but I have only seen it happen twice and I don't think it has been a major issue.

In any case, the platforms were pretty simple to make, are very easy to transport, are (hopefully) durable, and didn't cost an arm and a leg."

Bowl Trap Holder for Wetlands

Field Trip Checklist

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

"Bee Inventory, Monitoring, and ID" Discussion Group 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 Bee Inventory and Monitoring Lab's standard protocol for an individual site:

Setting Out Bowls – Follow these steps:

- Put one heavy squirt of dishwashing liquid in a 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.
- Place bowls level on the ground.
- Fill each bowl with soapy water about 3/4 or more full.
- Bowls can be left out for the middle part of the day or for 24 hours.
- Set bowls out in transects with 30 bowls spaced 5 meters apart (pacing is fine) alternating blue, yellow, and white.

- Avoid putting bowls in any heavy shade, as few to no bees will come to those bowls. There do not have to be flowers nearby to have bees come to bowls, as often there are bees scouting over flowerless areas.

Straining Bowls – Strain insects from bowls by dumping water from bowls through the brine shrimp net or use a disposable paint strainer.

After all bowls are strained, scoop out specimens with a spoon or your fingers, put insects in a Whirl-Pak; fill with just enough alcohol to cover the specimens. 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. The best kind is ethanol, but isopropyl will also work. Hardware store alcohol should be considered 95% alcohol … dilute it to 70%.

Add in with Whirl-Pak, a contents label written IN DARK PENCIL on a scrap of HEAVY paper saying collector, DATE (with month spelled out) and location. It would be useful to show where you collected on a map, but not absolutely critical.

Remove the air from the Whirl-Pak 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.

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 Whirl-Paks 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 Whirl-Paks as outlined in the section above. Put all the Whirl-Paks into a Ziploc bag and then, into another larger Ziploc bag to make sure nothing leaks. Some paper towels should be placed in the outer bag for added insurance.

Processing Bees that Have Been Stored in Alcohol of Glycol

Pinning bees directly from water or alcohol usually results in matted hairs and altered colors, along with a good coating of pollen, scales, and other detritus picked up from the sample. However, if specimens are processed within 24 hours of their capture, their hair and wings return easily to their natural state. Thus, if you are collecting bees from bowls into disposable paint strainers you can fold the top of the paint strainers over, staple them, leave them in an open container, freeze them, and then you can simply dump them onto a surface for pinning without washing. The specimens, while dirty, are very identifiable. For specimens that have been left in liquid for longer periods of time, 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 Whirl-Pak with water and then dump the 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).

Dump the specimens into a plastic container with a lid (put a knife hole in the lid to let out the foam). Add warm water and dishwashing liquid (more if the specimens are stored in glycol), and **very** vigorously shake the specimens around for **60 SECONDS**. IF YOU DO NOT WASH YOUR SPECIMENS WELL, YOU ARE DOOMED TO UGLY SPECIMENS.

Place specimens back into the strainer and rinse under warm to hot 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 or brine shrimp net as possible. A cloth towel is more environmentally friendly than using a lot of paper towels.

Either squirt 95%+ alcohol onto the specimens, dip the strainer into a bowl of 95%+ alcohol, or drop them into a jar of 95%+ alcohol and blot again.

Dump the specimens onto a set of 3-6 paper towels and fold the paper towels over the specimens and roll them around with your finger, pencil, or tweezers and refold a few times to remove the bulk of the alcohol.

At this point, you can fold the 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. See next section about using power dryers.

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. Étienne Normandin processes his specimens after a drying period of half an hour after which he puts them in a small sandwich bag filled with tiny pieces of cross-linked polyethylene, inflating the bag slightly, and shakes the bag vigorously to fluff the specimens.

Pin as normal.

Note that the paper towels can be reused many times. Note that the best looking bees are those that are cleaned within 24 hours of capture.

Bee Washer and Dryer – We have found that you can obtain beautifully coiffed hair on even the longest-haired bumblebees, if you spend the time shaking them around in a paper towel. Unfortunately, that can take a while. Most people shake them only until their wings unfold and then pin them, leaving the specimen less than presentable. We then have to identify 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, or modification thereof, below to speed things up.

You will need the following:

A small clear glass pint or half pint jar (a quart will do, but Morgan Lowry reports that smaller ones dry things faster) that has a canning jar lid of the kind with a removable central metal disk.

Replace the center of the canning jar lid with a similar sized section of fiberglass screening. We use the fiberglass type, but metal might be okay, 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. You can leave the screen loose and let the lid clamp it to the top or you can glue it with waterproof glue.

Using a Hair Dryer – Follow the same procedure as listed under the strainer section above but just do a quick blot of the specimens on the paper towels to get the bulk of the alcohol off.

Dump the specimens from the paper towel into the canning jar (we use a homemade funnel from the end of a large plastic soda bottle to help with this.

Put the lid back on the killing jar with the screen in the middle; make sure the screen is snug around the entire lid. Note that Tracy Zarrillo has had good success in extra fluffy bees by adding small rolled up bits of paper towel in with the specimens.

Turn on the hair dryer. We use high heat, although heat is not always necessary, particularly if the specimens are rinsed in quick-evaporating alcohol.

Place the jar on its side on the folded hand towel and place the hair dryer pointing into the jar as close as possible, without causing the hair dryer to cut out (usually about 1 inch). This can be hand held or set up in a wide variety of ways so that you don't need to hold the blower.

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 to 3 minutes.

Zak Gezon notes that he places the drying jar on its side in the top drawer of his desk drawer and tapes the hairdryer to the desk pointed into the jar. This frees his hands for more bee processing and makes sure he dries the bees long enough. Dave Smith does something similar by laying both the jar and the dryer on a towel on a counter.

Nick Stewart washes and dries bees in tea balls. The specimens are put into the balls, placed in the dishwashing liquid, swished around, rinsed and then placed in front of a blower used to blow up air mattresses.

Denny Johnson believes that Doctor Bronner's Magic Soaps are particularly effective at washing bees.

Using Compressed Air – We have found that using compressed air (or high-volume air from a blowers used for cleaning out the insides of computers) results in the quickest drying of wet bees. When using compressed air, be aware that there can be moisture in the air lines. 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. 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.

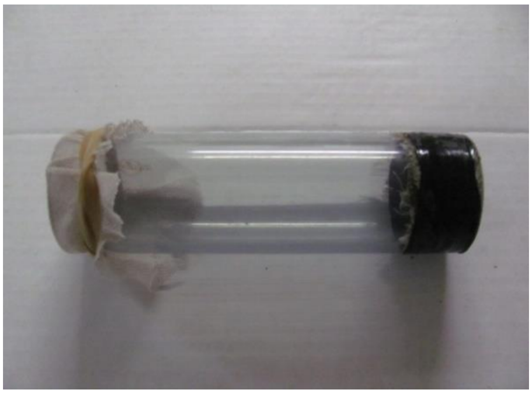
Making and Using an Autobeedryer – If you are involved in collecting and processing many specimens, you may want to invest in the creation of an autobeedryer. A slideshow and video that demonstrate how to make such a device can be viewed at:

> http://www.slideshare.net/sdroege/how-to-create-an-autobeedryer
>
> http://www.youtube.com/watch?v=935jlJep6go

Tracy Zarrillo notes that if you pin specimens that have been stored in alcohol immediately after drying them, alcohol inside the specimen will leak out and ruin their coiffed hair. She has found that putting them into a chlorocresol humidor for a week before pinning eliminates that problem. She also notes that pricking the ventral side of the thorax once and placing the specimen, ventral side down, on a piece of paper toweling can help with oozing issues as you place your pin into the scutum.

Upright Blow Dryer Bee Dryer – Dave Smith developed this system and writes: "The advantage to this system is it is fairly compact and easy to transport to BioBlitzs and pollinator-oriented activities.

I built this dryer out of a piece of 1X4 lumber and a few small pieces of PVC from my nearest hardware store. The blower sets upright and blows air through the tube placed on top of the dryer and dries the bees. The specific design of the wooden frame depends upon the size and shape of the particular blow drier that is used. I literally built the frame around the dryer, making certain I could slide it in and out of the frame for when I am travelling. Make sure you get a blow dryer that has a "cool" temperature setting. "Warm" or "hot" will bake the bees and make them brittle (even though it speeds things up to hit *Bombus* and other large hairy bees with a few minutes of "warm" air"). I strongly recommend taping the heat setting button in the "cool" position to prevent accidently "baking" your bees.

Upright Blow Dryer Bee Dryer

I use a clear plastic tube, but any PVC that fits into the larger piece glued on top of the dryer would work. The clear tube lets you watch your bees bounce around like air-popped popcorn (it is also entertaining when you are doing this at a public event). Glue or use electrical tape to attach fine netting at the bottom of the tube; close the top with another piece of netting and a rubber band.

After washing and partially drying your bees (following the explicit directions in Sam's slide shows); drop the wet bees in the plastic tube, set it in the large PVC tube holder on top of the dryer and turn it on. By the time you have washed the next batch of bees and prepped them, the bees should be dry (if you follow the one minute or more washing protocol)."

Modifying a Hot Air Popcorn Popper – Denny Johnson (DERMJOHN@aol.com) has created a lovely bee drier by modifying a hot air popcorn popper. You can email him for complete and detailed instructions.

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 scopae. 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.) Place bees into a rehydration container, a humidor or a covered Petri dish with a moist paper towel inside. It can take anywhere from a few hours to several days for larger specimens to relax. John Plant and Andreas Dubitzky found that you can speed the process of rehydration by adding boiling water to a small container, floating the specimens in the container on a small piece of Styrofoam, and closing the container with a tight fitting lid.

To prevent mold, add a few drops of ethyl acetate, a few mothballs, or a large dose of alcohol in the water. A useful technique (learned from the Packer Lab) is to affix foam into the bottom of a small plastic food container, put specimens you would like to rehydrate into that container, invert the container and place a slightly wet paper towel or two on top of the lid of the now inverted container and leave overnight. This rehydrates the specimens quickly but you don't have to worry about water dripping down from above onto the specimens or labels. However, if the paper towels are too wet then you can still get some beading of water on the specimens. Laurence Packer notes that the longer the bee has been pinned, the longer it takes to relax and the more fragile it becomes. Jim LaBonte uses household ammonia for his rehydrating fluid, as another potentially more powerful alternative, though some caution should be used as Jim primarily uses it for beetle specimens and it may not work as well on bees. Thanks to Jack Neff and Jason Gibbs for their contributions on this topic.

Preparing Dirty, Dry Bees for Photography

There are now many ways to take incredibly detailed pictures of bees using stacking software and either dedicated commercial systems or high quality camera equipment (see: http://www.youtube.com/watch?v=4c15neFttoU). Such techniques reveal minute structural details of bees, but also reveal hair matting, dust, and clumps of pollen that can detract from the specimen. Below is a process that is used at BIML for reconditioning old dry and dirty bees. The age of specimen does not seem to matter and material that is decades old has been successfully reconditioned. Note that hair that has been matted down by nectar or internal "juices" from the insect itself may or may not be completely recoverable. In general, short sparse hair recovers more readily than long hair. Testing is best with old *Bombus* specimens as their long hair is often a challenge.

Rehydrate the specimens at least overnight (we use the inverted food container mentioned in the previous section). Take a Falcon® tube/centrifuge tube/small container and add a small amount of VERY HOT water and a drop of dishwashing detergent. Drop the specimen STILL ON THE PIN but without the labels into the tube and shake VIGOROUSLY for about a minute or two – don't be shy about shaking, these specimens are tough. Take the specimen out and rinse under gentle running water. Quickly blot on some paper towels and drop into a tube of ACETONE (this replaces the remaining water with something that evaporates quickly and acts as a further solvent of goo, alcohol IS NOT as effective). Shake for only a few seconds. Remove and drop onto a paper towel to blot off excess acetone and immediately pick up and blow compressed air over the specimen. Compressed air is important because you need a high speed, precise air source or the hair will remain matted. Be aware that while you can use quite a strong air flow the wings are quite fragile and the tips will readily shred if the air passes directly over them, thus you will want to work on your technique with a few expendable specimens initially. We set the air flow to a moderate rate and hold the specimen directly in front of the compressed air nozzle, holding the specimen (still on the pin) with our fingers. In this way air can be precisely directed onto to the specimen without impacting the wings (which we will often hold together with our fingers). Depending on the specimen, a pin or tiny brush can help serve to unclump unruly hair during the process. Photographically, this also has the advantage of darkening the eye and making for a better looking picture, if taken right away.

Inexpensive, but Powerful LED and Florescent Light Sources

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 subpar light compared to that from fiber optic lights. We discovered that the Gerber LX3.0 LED mini-flashlight works extremely well as a microscope light (lovely white color). Laurence Packer has discovered that high wattage compact fluorescent bulbs work well and provide great surface details, and currently many people now use Ikea's JANSJÖ LED work lamp which is incredibly inexpensive, but does have a slight yellowish cast.

Note that if any light is creating too much glare or reflectance on your specimens you can soften that by adding a small piece of translucent plastic bag or velum paper over the top of the light. We found that the Gerber flashlight (and likely others) fits very nicely into the standard Bausch & Lomb microscope stand's illuminator hole. As flashlight batteries drain down, the light will dim. When in the field, it is useful to have spare sets of batteries charging to replace drained batteries as needed.

In an office or lab setting, you can convert these flashlights to use household current.

To convert, you will need a wall cube transformer of some kind that converts 120V AC 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 120V AC (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.

German Perilla has created a wonderful PowerPoint how-to presentation about converting flashlights into microscope lights and that is available at:

http://www.slideshare.net/sdroege/how-to-make-a-microcope-light-ppt

We recommend that you use the PowerPoint 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. Attach a tiny screw to one end of a dowel. Then attach a wire to that screw and tighten 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). Tape the wire to the dowel and run the whole thing to the bottom of the flashlight. For the return, grind off some of the outer nonconductive anodized finish on the flashlight body, and simply tape the end of another wire to the body. Cut the end of the wall cube off and attach its wires to the wires coming off of the flashlight. 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 and recommendations about other similar flashlights, check out the LED or similar discussion forums at the Candle Power Forums searchable flashlight discussion site at:

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

Compact fluorescent bulbs in the 100-150 watt equivalent range work extremely well as microscope lights.

They are superior to all other lights for illuminating subtle microsculpture on specimens (and they are very important in groups like *Lasioglossum* and *Hylaeus*). Bulbs can be added to student lamps or articulating lamps available from many online stores, as well as well stocked office supply and household goods stores. Note that these bulbs do produce a lot of heat from the ballast at their base and if the lamp is too restricted (i.e., no holes at the base of the bell of the lamp), this heat will burn out the electronics of the bulb. As with all light sources, the closer you can get the bulb to the specimen, the better.

How to Make a Pizza Insect Pinning Box

Written by Rob Walker and Sam Droege (Refer to figure at end of this section)

Because of the volume of insects collected at BIML, 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 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 foam in bulk you will save a great deal by going directly to a local manufacturer (look under "foam" in the Yellow Pages). We have them cut the foam to 3/8-inch thickness and ship as 2-ft x 4-ft sheets. Often these manufacturers have blocks of foam that are scrap or overruns in their building, so you might ask them if any are available and have them cut that scrap into 3/8-inch pieces for you for a discount.

Assembly directions for a standard pizza box:

Use a knife, scissors, or paper cutter to cut and separate Section I from Section II, along red arrows as shown.

Take Section II and assemble by taking side flaps A and turning in end tips.

Fold flap B over end tips so that the tabs are securely in the slots provided.

At other end, fold end tips in and fold up flap C.

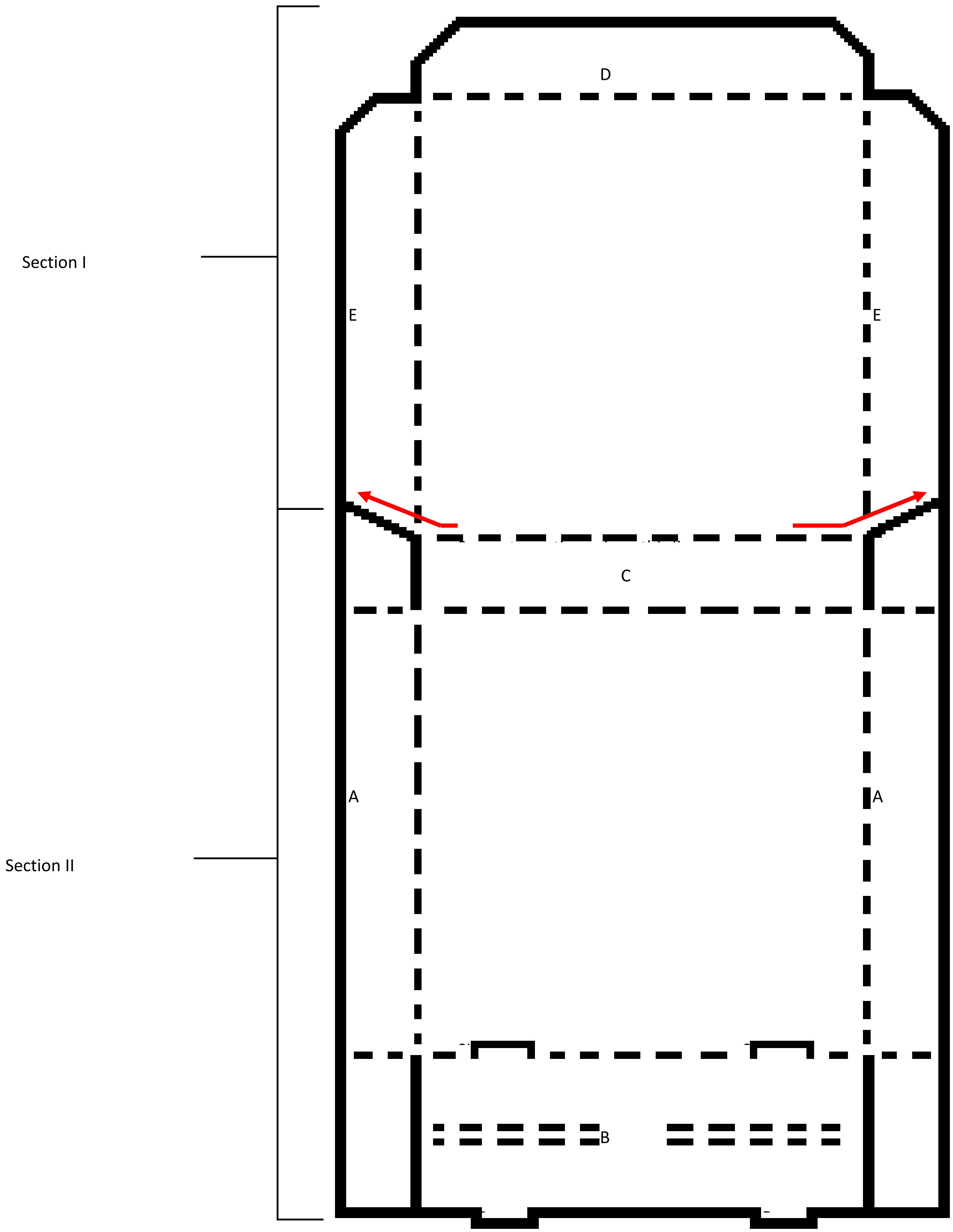
Staple flap C and the end tips together so that flap C stays upright. (Staple 4 times per end tip to secure them.)

With blade or paper cutter, remove flap D completely from Section I.

Fold up flaps E.

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.

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. Use good quality glue sticks and avoid the generic types whose gluing abilities can be quite low.



D
Section I
E
E
C
A
A
Section II
B

Theodore Mitchell's Guide: *Bees of the Eastern United States*

While published in 1960 and 1962, Theodore Mitchell's 2-volume set on *Bees of the Eastern United States* is still a very valuable reference book and source for identification keys, illustrations, and species accounts. These two volumes are now quite expensive to purchase via rare book dealers, however they are available for free as a series of PDF files from the Insect Museum, Department of Entomology, North Carolina State University. They can be accessed at:

http://www.cals.ncsu.edu/entomology/museum/easternBees.php

Additionally, Glenn Hall scanned the indices. They can be accessed at:

http://xa.yimg.com/kq/groups/17598545/896478426/name/Mitchell index.pdf

Note that Mitchell's taxonomy is out of date. 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. You can cross-reference names for synonymy by either going to one of the genera guides directly or, better, going to the world bee checklist home page of Discover Life (http://www.discoverlife.org/mp/20q?guide=Apoidea_species&flags=HAS:). The world checklist of bees can be filtered by country, genus, subgenus, family, and subfamily and has been put together by John Ascher and John Pickering. To locate synonymies go to the "**Checklist**" link in the blue banner at the top of the page and then use your browser's "find" function to locate synonymies, note that the checklist always shows all of the species around the world and is unaffected by any of the filters that you may have applied at an earlier stage.

Mike Arduser's Midwest Keys

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

http://www.pwrc.usgs.gov/nativebees/Keys.html

Canadian Identification Guides

Laurence Packer's Lab has produced a guide to "The Bee Genera of Eastern Canada":

http://www.biology.ualberta.ca/bsc/ejournal/pgs_03/pgs_03.html

They also have a "Key to the Bee Families of the World":

http://www.yorku.ca/bugsrus/BFoW/Images/Introduction/Introduction.html

And an image database of the "Bees of Canada":

http://www.yorku.ca/bugsrus/bee_canada/Bee_Genera_Canada.html

And a pictorial catalog of the "Bee Tribes of the World":

http://www.yorku.ca/bugsrus/bee_genera_of_the_world/Bee_Tribes.html

A Guide to Identifying Bees Using the Discover Life Bee Keys

Discover Life keys cover all the bee species in North America east of the Mississippi; coverage extends to all of North America for some genera, with the objective of covering the entire region eventually. All of the bees in the Caribbean, Mexico, United States, Canada, and the British Isles are covered in the genus identification guide.

he section below provides guidance for the use of online Discover Life guides or keys. These instructions are designed for use with the guides to 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 at the end regarding the use of already identified specimens. A set of identified specimens that you can practice with can be obtained at no charge from Sam Droege (sdroege@usgs.gov). Note, that using already identified specimens is the best way to learn how to identify bees.

All of the Nature Guides are located at:

http://www.discoverlife.org/mp/20q

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

http://www.discoverlife.org/mp/20q?search=Apoidea

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

Discover Life 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.

On the bee page at Discover Life, there are a series of guides listed for **Eastern North American** bees (states and provinces east of the Mississippi River). Many of these guides have been expanded to include Western species and over the coming years we will expand all guides to include the western states and provinces. Guides are constantly being updated with pictures, corrections, and better wording.

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.

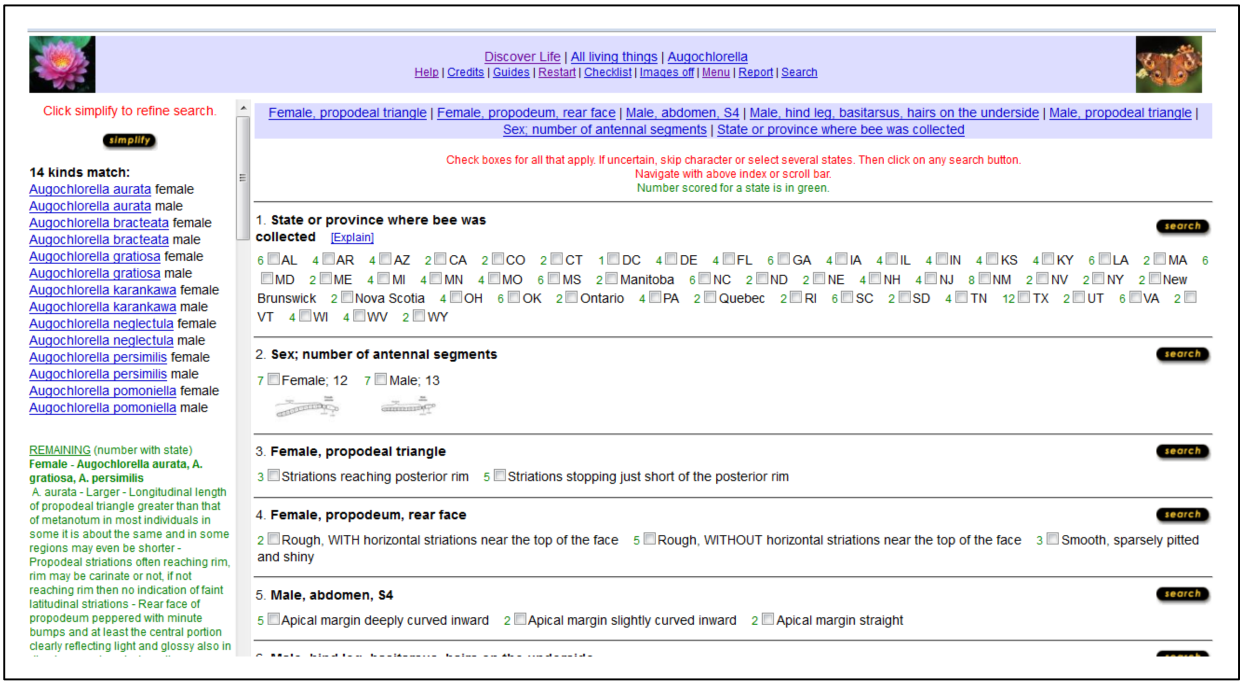
Hint: If you are unfamiliar with the bee genera we suggest that you start your identification process by using the guide to bee genera to divide your collection into genera.

The instructions that follow apply equally to the bee genera guide or to each individual bee genus guide.

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, when any "**search**" button is clicked. Similarly, one can flip the process, by clicking the "**simplify**" button, 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 certain 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 two types of species that remain on the list: 1) Those species that have the characters you have indicated, and 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 characters 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 identification 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. At the top of the left hand column, click on the "**characters**" option. Next, click on the species you wish to review. Finally, hit the "**submit**" button to get a list of scored characteristics.

One nice feature of the Discover Life 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 that 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 – B y 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 two 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 "**only**", "**has**", and "**not**" 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 Discover Life 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. Sam's 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 identification 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, you know which state of which character you entered that led 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 free identified specimens that you can keep and use.

Worldwide Checklist of Bees and Bee Synonymies – A list of the bee species of the world that lets you sort them by country, various taxonomic units, and some life history traits is available from John Asher and John Pickering at:

http://www.discoverlife.org/mp/20q?guide=Apoidea_species&flags=HAS:

A list of all the known synonymies for each of the species is similarly available at:

http://www.discoverlife.org/mp/20q?act=x_checklist&guide=Apoidea_species

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 (i.e., stylopized) by a strepsipteran. The Order Strepsiptera is a mysterious taxon 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. Female 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 peeking out from beneath the rim. Sometimes the apical rim of the tergite covers most of the parasite's 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.

Strepsipterans 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.

Stylopized *Andrena vicina* – The female strepsipteran cephalothorax is the pale rounded extrusion poking out between the tergites. (Photographed by Ellen Bulger)

<u>Affixing bee wings to microscope slides</u> – (Contributed by Tulay Yilmaz and Gökce 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

Procedure:

- Place the slide on a white piece of paper for easy visibility.
- Put some warm water into the Petri dish.
- Turn on the lamp and leave until it’s hot.
- Take the bee’s wing with the tweezers.
- Place the wing into the warm water; wait for a while to get it as smooth as possible.
- Remove the wing from the water and put it onto the drying surface (be sure the wing stays flat).
- Leave it under the light to dry.
- Remove when dried.
- With that glass rod, drip some Entellan onto the slide and spread it.
- 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).
- Don’t use a coverslip!
- Then look at the preparation under the microscope.
- If you see any air bubbles 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.
- 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 preparation in the open air.
- Clean your glass rod immediately (because when Entellan hardens, it gets difficult to clean).
- Preparations can be used when Entellan is dry (usually within an hour).

Bee Wings in Entellan on a Microscope Slide

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 Entellan or disappear.

<u>Specimen Donations and Income Taxes (United States)</u>

Doug Yanega nicely researched the following advice to the United State collector who wishes to donate specimens to museums and write-off those donations on their income taxes. If your specimen donation is above $5000, you evidently must have a certified appraisal performed. Below that amount, you must demonstrate "fair market value" from an independent pricing guide – and, to my knowledge, there is only one such guide that lists miscellaneous insects, and the price there is a flat $3.00 per specimen. If you go to http://www.bioquipbugs.com/Search/WebCatalog.asp?category=1110, you will see the catalog listings for Hymenoptera, and if you click on any of the bee families, you will see that the minimum price for any bee (identified or unidentified) is $3.00 per specimen.

<u>Introduced and Alien Bee Species of North America (North of Mexico)</u>

Information on distributions and status of the approximately 40 alien species 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, and Robbin Thorp.

Updated: January 2015

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

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* (sweetclover).
A *Hylaeus hyalinatus* 1990. Europe. Currently found in urban areas from New York City, southern Ontario, New Jersey, Pennsylvania. Has potential to spread throughout North America.
A *Hylaeus punctatus* 1980. Europe. Currently found in central California, Mid-Atlantic states, Ontario, Chicago region, Denver area. Has potential to spread throughout North America.
A near *Hylaeus* (*Prosopis*) *variegates* 1990. North Africa. Currently detected only in the Greater New York City region, the exact species name is unclear but being pursued.

Andrenidae

A *Andrena wilkella* 1900s. Europe and northern Asia. Common throughout the north-central and northeastern United States and southern Canada.

Halictidae

A *Lasioglossum eleutherense* 1990. Bahamas and Cuba. Four individuals found in the University of Miami Arboretum and a recent specimen from Biscayne National Park. Not expected to spread outside 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 United States and southern Canada. Molecular work indicates that actual introduction could have been significantly earlier than 1900 when first detected.
A *Halictus tectus* 2000. Southern Europe to Mongolia. Currently known only from Philadelphia, PA, and the Baltimore, MD/Washington, DC region. Appears to prefer highly disturbed sites with European weeds.
A *Lasioglossum zonulum*?. Europe and SE China. A species similar to *L. leucozonium*. Recently thought to possibly be an introduced rather than a native species. Records in North America go back many years.

Megachilidae

A *Anthidium manicatum* 1960. Europe, North Africa, Near East, south-central and southeastern South America. Currently found predominantly in northeastern United States, upper Midwest, and southern Canada, however, now established in the central Rockies and 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* (hedgenettle).
A *Anthidium oblongatum* 1990. Europe and the Near East. Currently common in northeastern United States and southern Canada and moving into the central states and provinces, scattered records now exist for Colorado and Washington state. 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 northern North America.
A *Chelostoma rapunculi* 1960. Europe and the Near East. Found in Upstate New York and southern Ontario. Has potential to spread throughout northern North America.
A *Coelioxys coturnix* 2000. Southwestern Europe, North Africa, India. Currently found in the Baltimore, MD/Washington, DC corridor west to southern Pennsylvania and Allegany County, MD and also recorded in southern New England. Has potential to spread throughout the range of *Megachile rotundata* (its presumed host).
A *Heriades truncorum* 2010. Europe and the Near East. Two females and a male found in Washington County, MD in 2013. A common and spreading hole-nester in at least parts of Europe, should be watched for in trap nests throughout North America.
A *Hoplitis anthocopoides* 1960. Europe. Uncommonly found from West Virginia and Maryland to southern Ontario. Potential spread perhaps limited to the range of its reported preferred pollen source, Common Viper's Bugloss (*Echium vulgare*).
A *Lithurgus chrysurus* 1970. Europe, Near East, North Africa. Found in the Phillipsburg, NJ area and a 50-mile radius in Pennsylvania and New Jersey, but in 2011 noted well to the west near State College, PA. Until 2007 there were no recent records, but perhaps due to no one making an effort to look. Apparently oligolectic on Spotted Knapweed (*Centaurea stoebe* ssp. *micranthos*) and burrows into wood to make a nest. This species has the potential to be much more destructive than *Xylocopa virginica* to wooden buildings. Noted nesting in old firewood piles, timber frame covered bridges, and in wooden shingles.
A *Megachile apicalis* 1930. Europe, North Africa, Near and Middle East. Western and eastern United States. Relatively few records in the East but widespread in California and parts of the Pacific Northwest where it specializes on Yellow Star-thistle (*Centaurea solstitialis*), and is often moved around with *Megachile rotundata* pollinator tubes.
A *Megachile concinna* 1940. Africa. West Indies, Mexico, uncommon throughout the southern and western United States.
A *Megachile ericetorum* 2000. Europe, Near East, China. Now established in southern Ontario and recent records from Rochester, NY. Should be expected to spread.
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. Common 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 United States, Colorado, 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 United States and southern Canada. Appears to be less common than it once was, at least towards the south. Few 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 United States, with some establishment noted for the Pacific Northwest. 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, spreading north and south. Males in particular are very similar to *O. cornifrons* and may be confused. Appears to be rapidly

spreading and often abundant.
A *Pseudoanthidium nana* 2000. Europe and the Near East. Currently detected in New York, NY, Baltimore, MD, and western Maryland. So far, only found in the most industrial, disturbed, and urban sites.

Apidae

I *Apis mellifera* 1620. Originally from northern Europe, later more from 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, DC metropolitan area where it nests in the ground under porches or in the dirt of uprooted trees and frequents planted azaleas (*Rhododendron* spp.) and other garden flowers. Records now exist for Frederick County, MD and nearby Pennsylvania and spread from there is expected. This species 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, TX.
A *Ceratina dallatorreana* 1940. Mediterranean region. Central California.
I *Ceratina smaragdula* 1960. Pakistan, India, SE Asia. Introduced into California but not found since its introduction, however abundant in the Hawaiian Islands.
A *Centris nitida* 2000. Southwestern United States, Texas, Mexico, Central America and northwestern South America. Recently discovered in southern Florida. Not expected to spread outside of Florida.
A *Euglossa dilemma* 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.
I? near *Plebeia frontalis* 2010. Mexico, Central America, South America. One colony detected in Palo Alto in 2013 that has remained active until the writing of this account (November, 2015). Could possibly spread down the coast of California. Population status is unclear and the exact species is not known either.
A *Xylocopa appendiculata* 2010. Japann and China. One specimen collected in 2012 and another photographed in 2013 in San Jose, California. Populations status is unclear.
A *Xylocopa tabaniformis parkinsoniae* 1990. 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.

<u>Mini-summary of the Genera of Eastern North American Bees</u>

(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 Largest of the bright metallic green 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 a few females with yellow on clypeus. Several species are willow (*Salix* spp.) 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; 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 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, Pseudoanthidium*

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 very uncommonly encountered, usually only in high-quality habitat. 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, Pseudoanthidium*

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, DC area and should be expected elsewhere soon. An early spring bee and occurs in woodlands as well as urban and field habitats. The other species are usually uncommon late spring to summer species that occur in mixed habitats. Some species look superficially like bumble bees by body shape, while others look like the eucerines. The hairless internal cells of the forewing narrow the possibilities down to *Anthophora* and the rarer *Habropoda* and *Melecta* genera. *Habropoda*, *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*. Very small bees, about the size of *Lasioglossum*, both males and females extremely hairy, particularly the hind legs. *Exomalopsis*

Ap *Apis mellifera*(1) N SPSUFL |NE|MAa|DS|GL|MW|OQ|AC| 9-20mm Note that this species is relatively uncommon in pan traps. Long hair on eyes and the unique hind leg architecture is a giveaway. *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 wooded. Most often confused with *Augochlorella* spp. Told by minutely truncate tip of marginal cell, the female's large dark forked tip of the mandible, and the suture pattern 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. *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, Anthophora, Habropoda*

An *Calliopsis*(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; other species extremely rare. The small size, two submarginal cells, the bright yellow legs of the male and the three vertical ivory-colored facial markings of the females are a distinctive combination. *Perdita, Andrena*

C *Caupolicana*(2) N SUFL |-|-|DS|-|-|-|-| 18-22mm 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 native morning glories (*Ipomoea* spp.), very rarely detected. The rim of the clypeus has two 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 bumble bee/*Anthophora*-looking group. Currently restricted to Florida and southern Georgia, but the introduced *C. nitida* could spread beyond the states. 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*(5) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 2-9mm Found in most habitats. Small metallic steel blue to dark green bees with white markings on their clypeus (one tiny species nearly jet black), 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,* who they parasitize, but usually narrower. Most females with a clearly pointed and extended abdomen tip. The tip of most male abdomens 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 honey bee. Face heart-shaped due to the angling inward of the compound eyes. Distinctive that lower portion of the second recurrent vein 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; found primarily in deep sandy areas (this is not the case in the West). 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, Pseudoanthidium*

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. Two 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*.* *Andrena, Halictus*

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 Nova Scotia and Connecticut. 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 <u>almost</u> 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 and not seen for many years. 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 to be 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 dilemma*(1) N |-|-|DS|-|-|-|-| 11-44mm A recently discovered introduction, currently only occurring in South Florida. Bright green in color, 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 honey bees, 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 near wetlands with Pickerelweed (*Pontederia cordata*). 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 An early spring bumble bee-like species, often associated with blueberries (*Vaccinium* spp.). Technically closer to some of the more uncommon *Anthophora* species than bumble bees. The shape and configurations of the marginal/submarginal cells 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 this species' metallic body. This confusion will extend to *H. tectus* a new metallic invasive that has been detected in Philadelphia, PA and the Baltimore, MD/Washington, DC areas. 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 and are uniform and complete. 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*(4) 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 of Mexico 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 An uncommon and minute group of parasitic species. Males unique (and therefore confusing) in that they have only 12 antennal segments unlike all other genera with 13. Abdomens red in the most common Eastern species, 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*(25) 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-shaped 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. 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 so in males where it can be difficult at times to detect and consequently males may key out to the genus *Sphecodes* or *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, Sphecodes*

Mg *Lithurgus*(3) N SPSU |-|mar|DS|gl|-|-|-| 8-19mm Uncommon but 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. Labrum is longer than broad. A pygidial plate is present in both sexes, though spine-like in the female. *Megachile*

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 yellow loosestrife (*Lysimachia* spp.) plants.

Mg *Megachile*(44) N SPSUFL |NE|MAc|DS|GL|MW|OQ|AC| 5-21mm Bees in this genus are generally larger than other species where the female has scopa under 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. Somewhat similar to eucerines, but separation technical in females. Males have two small cones or obvious spikes projecting backwards out of the scutellum. 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, bumble bee-like bodies. Males have extremely long antennae. Females told from other eucerines by the shape of the front of the tegula, however, this is often hidden by dense hair and must be scraped 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 Extremely rare, not seen in years. 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 usually 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 dilated, sometimes greatly so. *Dieunomia*

Mg *Osmia*(29) N SPSU |NE|MAc|DS|GL|MW|OQ|AC| 5-17mm Stubby, most are dark metallic blue or green, a few of the larger species are brown. Has a nearly absent or limited parapsidial line on thorax that is either just an enlarged pit or travels in a few cases only a very short distance. *Hoplitis, Ashmeadiella, Heriades, Chelostoma*

An *Panurginus*(3) N Spsu |-|MAu|DS|-|MW|-|-| 5-10mm Small, uncommon, black species with relatively unpitted scutums, the males often having yellow on their faces. Two submarginal cells with the recurrent vein intersecting directly with the cross vein between the two 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's 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| 5-8mm 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 on Asteraceae. *Pseudopanurgus, Panurginus, Protandrena*

An *Protandrena*(3) N SPSUfl |-|MAr|DS|GL|MW|-|-| 7-10mm A very uncommon group, best told by keying them out through the guide. Females with extensive yellow on clypeus.

Mg *Pseudoanthidium nanum*(1) N sp?SUFL |ne|mar|-|-|-|-|-| 5-8mm Industrial and urban habitats. One introduced species currently (2010) found in the Mid-Atlantic and the Northeast, but expected to spread. Ports and industrial areas should be searched for new records. Small, stocky bees, fast fliers with strong yellow markings, particularly noticeable on the abdomen, this species is smaller than bees in the genus *Anthidiellum*, the smallest native species. Females have multiple teeth on their mandibles. *Trachusa, Stelis, Anthidium, Dianthidium, Anthidiellum*

An *Pseudopanurgus*(15) N SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 3-10mm Similar to *Panurginus*. Small, dark bees, with two submarginal 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 Bumble bee-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 (note that males are often all black). Similar to *Lasioglossum* but females lack scopa, wings have no weak veins, most species have strongly sculptured propodeums. *Nomada, Lasioglossum*

Mg *Stelis*(12) P SPSUFL |NE|MAu|DS|GL|MW|OQ|AC| 3-12mm Uncommon, small to medium-sized. Variable in look, varying from small and black to larger specimens with extensive yellow and sometimes red markings. Females lack scopa. *Dianthidium, Anthidium, Anthidiellum, Paranthidium, Trachusa, Pseudoanthidium*

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 between the scutum and scutellum and along the base of T2. *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 identification 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, Pseudoanthidium*

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, bumble bee-like, with flattened faces. Males have prominent white facial markings, both with a very long marginal cell, hind wing with a jugal lobe, black abdomen with few hairs and slightly iridescent surface readily visible. *Bombus, Ptilothrix*

Example Account 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 Megachilidae, 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. The third letter following the mid-Atlantic code indicates the commonness status of that group in the mid-Atlantic area.

Many thanks to Mike Arduser, John Ascher, Rob Jean, John Pascarella, and Cory Sheffield 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 March 2015

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 /ag-uh-PAN-thin-us/
Agapostemon /ag-uh-PAHST-eh-mon/
Agapostemonoides /ag-uh-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/
Anthidium /ann-THID-ee-yum/
Anthodioctes /ann-thoh-dee-OCK-tees/
Anthophora /ann-THAH-for-uh/
Apis /A-piss/
Ashmeadiella /ash-MEAD-ee-el-uh/
Atoposmia /ate-op-OZ-me-yuh/
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 /brack-ee-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/
Cephalotrigona /seph-al-oh-trig-OH-nuh/

Ceratina /ser-uh-TIE-nuh/
Chelostoma /chel-AHST-oh-mah/
Chilicola /chill-LICK-oh-luh/
Chlerogella /clair-oh-GELL-uh/
Coelioxoides /seal-ee-ox-OID-ees/
Coelioxys /seal-ee-OX-ees/
Colletes /koh-LEE-teez/
Conanthalictus /koh-nanth-hal-ICK-tuss/
Crawfordapis /kraw-ford-A-piss/
Ctenioschelus /ten-ee-oh-SHELL-us/
Deltoptila /delt-op-TIL-uh /
Diadasia /die-uh-DAY-zee-uh
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 /ep-EE-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-eh-oh-POHD-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/
Melipona /mel-lih-POE-nuh/
Melissodes /mel-ih-SOH-deez/
Melissoptila /mel-lis-SOP-till-uh/
Melitoma /mel-lih-TOE-mah/
Melitta /mel-IT-tuh/
Meliwillea /mel-lih-WILL-ee-uh/
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/
Nannotrigona /nan-oh-trig-GOH-nuh/
Nanorhathymus /nan-oh-rath-THIGH-mus/
Neocorynura /knee-oh-CORE-ey-nur-uh/
Neolarra /knee-oh-LAIR-uh/
Neopasites /knee-oh-pass-EYE-teez/
Nesosphecodes /knee-zoh-sfeck-O-deez/
Nogueirapis /no-GAYR-A-pis/
Nomada /no-MOD-uh/
Nomia /NO-mea/
Odyneropsis /oh-dee-ner-OP-sis/
Oreopasites /oh-ree-oh-pass-EYE-teez/
Osiris /oh-SIGH-ris/
Osmia /OZ-me-yuh/
Oxaea /ox-AYE-ee-uh/
Oxytrigona /ox-ee-trig-OH-nuh/
Panurginus /pan-ur-JINE-us/
Paragapostemon /pear-ag-uh-PAHS teh-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 /par-tuh-MO-nuh/
Peponapis /PEE-po-nay-piss/
Perdita /per-DIH-tuh/
Pereirapis /pear-ee-eye-RAPE-is/
Plebeia /pleb-ee-EE-uh
Protostelis /proe-toe-STEEL-iss/
Protandrena /prot-an-DREE-nuh/
Protodufourea /pro-toe-dew-four-EE-uh/
Protosiris /pro-toe-SIRE-is/
Protosmia /pro-TOZ-mee-uh/
Protoxaea /pro-tox-EE-uh/

Pseudaugochlora /sood-aug-oh-KLOR-uh/
Pseudopanurgus /sue-doe-pan-UR-gus/
Psithyrus /SITH-ih-russ/
Ptilocleptis /till-oh-KLEP-tiss/
Ptiloglossa /till-oh-GLOSS-uh/
Ptilothrix /til-o-THRIX /
Ptilotrigona /till-oh-trig-OH-nuh/
Rhathymus /rath-THEE-mus/
Rhinetula /rhine-ET-tule-uh/
Rhopalolemma /rope-al-oh-LEM-uh/
Scaptotrigona /scap-toe-trig-OH-nuh/
Scaura /SCOUR-uh/
Simanthedon /sigh-MAN-theh-don/
Sphecodes /sfeck-OH-deez/
Sphecodogastra /sfeck-kode-oh-GAST-ruh/
Sphecodosoma /sfeck-kode-oh-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 /tha-LES-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 /zeer-o-mel-LECK-tuh/
Xylocopa /zile-low-COPE-uh/
Zacosmia /zack-OZ-mee-uh/
Zikanapis /zick-ann-A-piss/

Glossary of Bee Taxonomic Terms

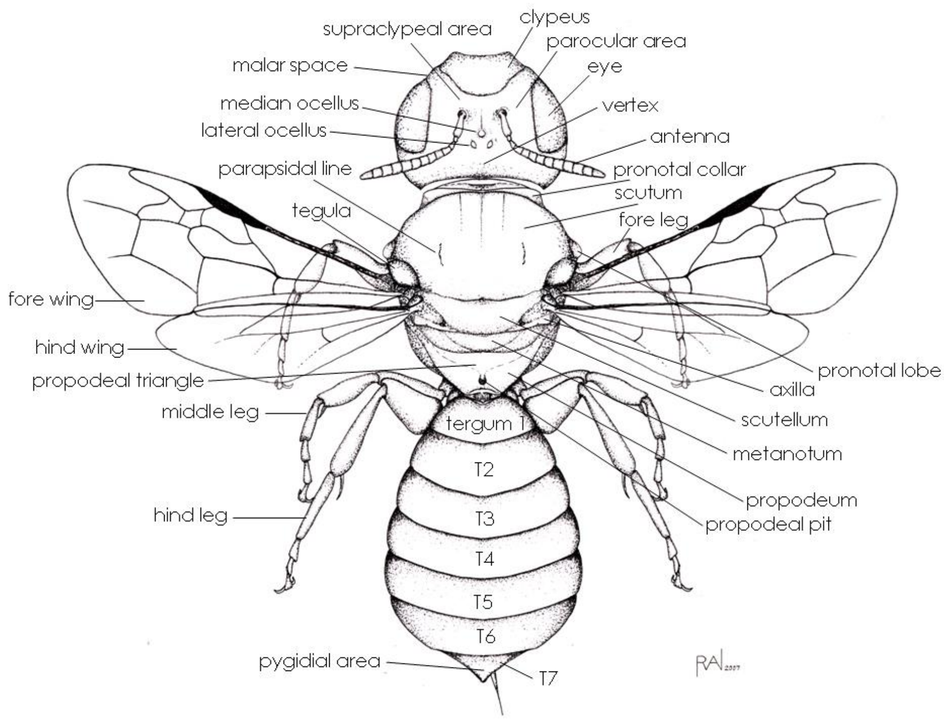
Angulate – forming an angle rather than a curve
Anterior – toward the head or on the head side of a segment being described
Apex – end of any structure
Apical – near or at the apex or end of any structure
Appressed – tight and flat against the body of the bee, usually used to describe hair
Arcuate – curved like a bow
Areolate – an area dissected by reticulating raised lines forming clear and strongly defined cells
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 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 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. Bumble bees and honey bees have this on their tibia, while *Andrena* have a patch on the sides of their propodeum
Costa – a wing vein
Coxae – the basal segment of the leg
Cubital – a 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 opposed 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 farthest 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
Fovea – 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
Gena – The cheek or the region below the eye seen when viewing the head from the side and holding the head so that the flat of the face is at right angles to your line of site – like a carpenter would sight down a piece of wood
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 – Located near the top of the mesepisternum, it is the raised, mound-like area just below the attachment of the front wing and often contains slightly different pitting and reticulating patterns than the rest of the mesepisternum
Hypostoma – the notched region underneath the head and behind the mandible that holds the folded tongue
Imbricate – lined with microscopic inscribed lines that form a fish scale-like pattern
Impressed area – almost always refers to the apical part of the upper abdominal segments, these areas often being very slightly (often very difficult to detect) lower than the basal part of the segment
Impunctate – not punctate or marked with punctures or pits
Infuscate – 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 – a wing vein
Interstitial – when describing veins, it refers to the end of one approximating the beginning of another, as in a grid intersection

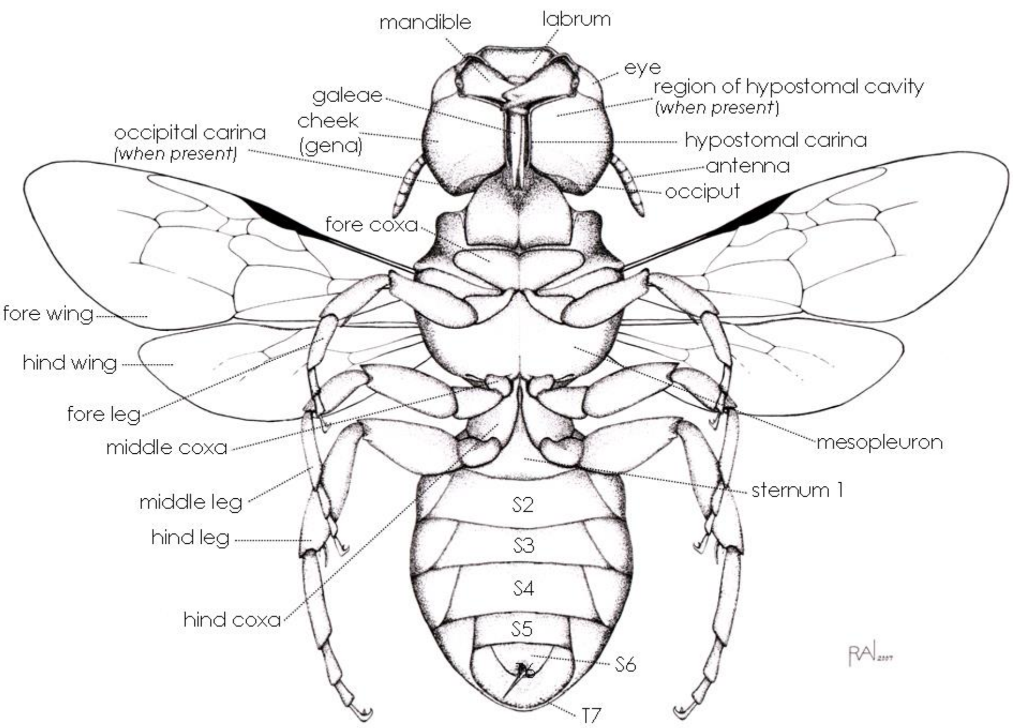
Labrum – abutting the clypeus in front of the mouth
Macula (Maculation) – a spot or mark
Maculate – spotted or made up of several marks
Malar space – the shortest distance between the base of the mandible and the margin of the compound eye often completely absent in bees
Mandibles – bee "jaws," so to speak, usually crossed and folded in front of the mouth
Marginal cell – a wing cell located on the front edge (margin) of the wing
Mesally (Medially) – pertaining to, situated on, in, or along the middle of the body or segment
Mesepisternum, Mesopleura, or Mesothorax – 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 three simple eyes or lenses that sit at the top of the head of bees
Ochraceous – pale yellow
Outer – usually refers to legs and specifically to the surfaces facing away from the body
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
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 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 it 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 a 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
Reflexed – bent up or away
Repose – in a retracted physical state
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 well above the surface
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 – a 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 bee's leg, attached to the tibia
Tegula – 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
Tomentose – cove red with tomentum
Tomentum – a form of pubescence composed of short matted, woolly hair
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
Trochanter – the segment of the insect leg between the coxa and the femur
Truncate – cut off squarely at the 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 to the undersurface of the abdomen
Vertex – the top of the head
Violaceous – violet-colored

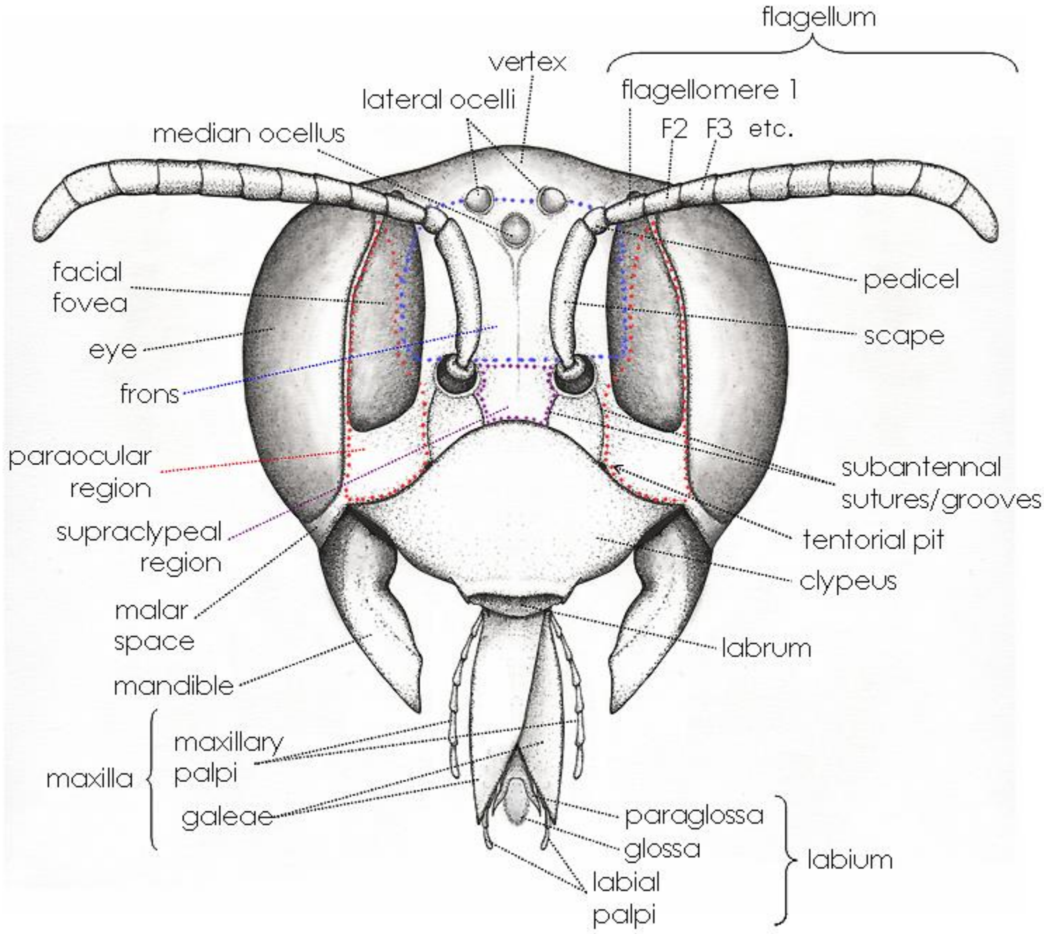
<u>Bee Body Part Figures</u> – Drawn by Rebekah Nelson



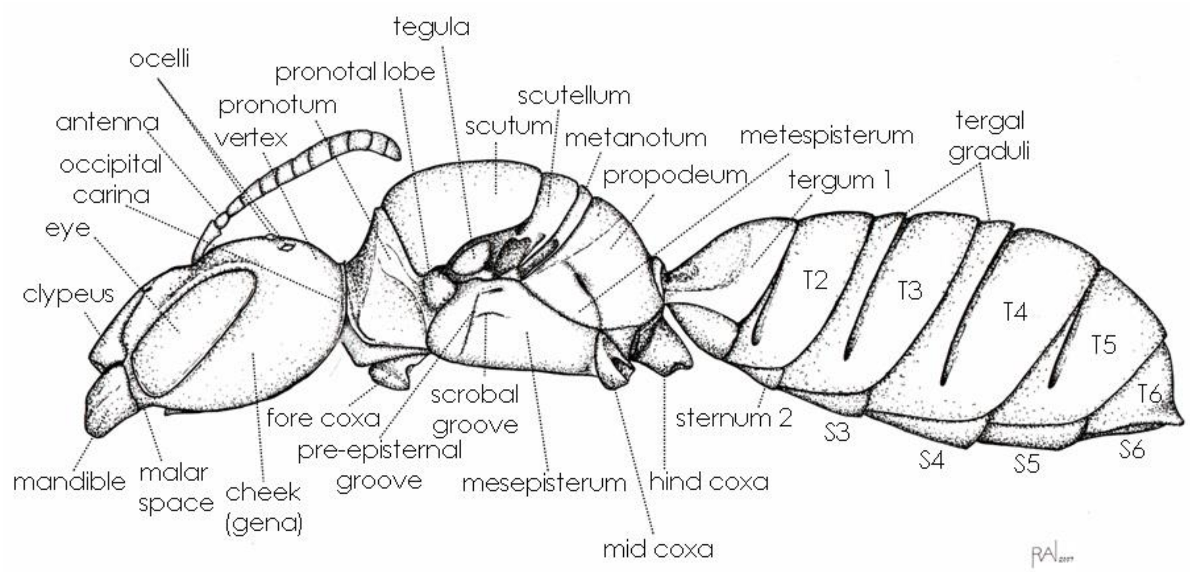
Dorsal View



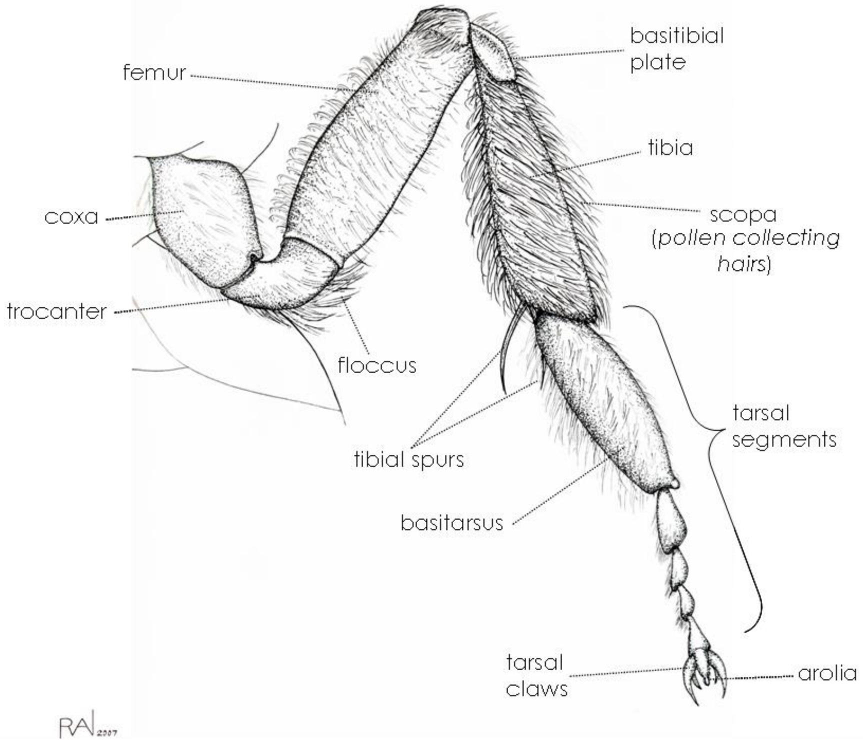
Ventral View



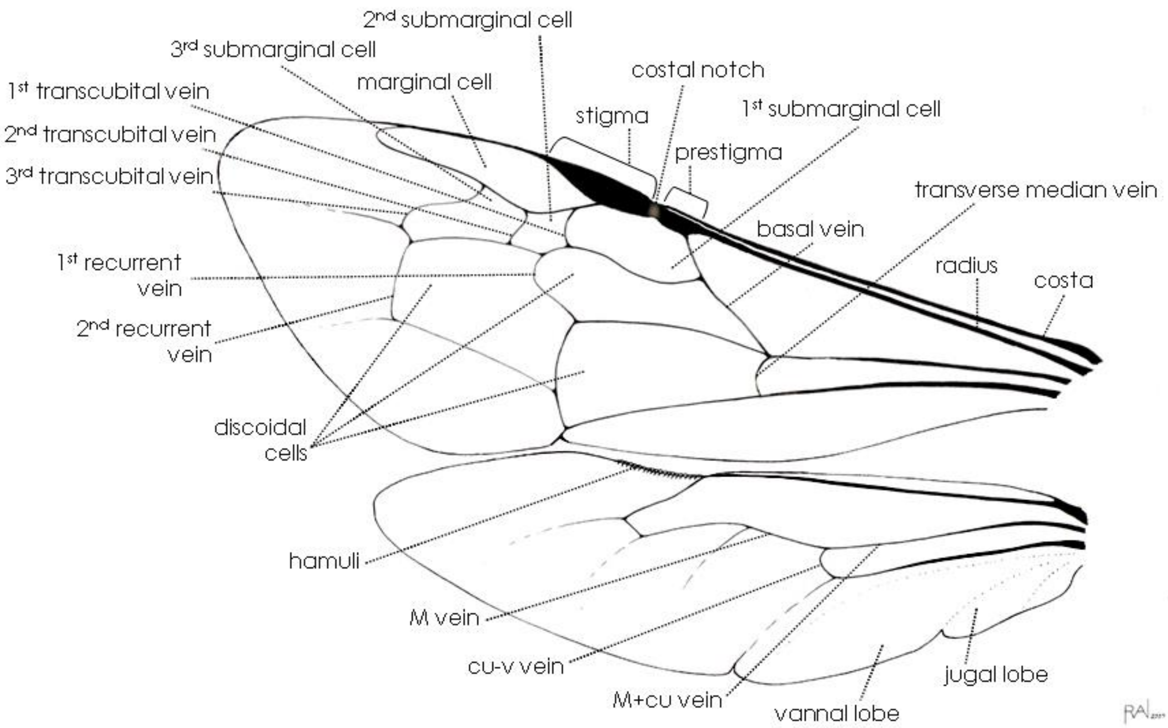
Frontal View



Lateral View



Hind Leg



Fore Wing and Hind Wing