

About the Michigan Odonata Survey

The Michigan Odonata Survey was formed in the summer of 1996. Mike Kielb, Mark O'Brien and Ethan Bright have formed the nucleus of the MOS, and with cooperation from other individuals and organizations, we feel that the MOS will grow to be a successful survey of the state of Michigan's Odonata fauna. We thank Bob Glotzhofer of the Ohio Dragonfly Survey for inspiring our effort. The Ohio people have really done a remarkable job with their survey, which is supported by the Ohio Biological Survey, Ohio EPA and Ohio DNR as well as the Ohio Historical Society. The Ohio survey has taken at least 7 years, and has amassed quite a set of data based on historical records and recently collected specimens. The cooperation amongst various agencies is quite remarkable, and I hope the MOS will attain a similar level of cooperation among various agencies and people in Michigan.

There are many areas of the state that are poorly documented in regards to their Odonata fauna (and other groups as well). Most of the Upper Peninsula, the lower tier of counties along the Indiana/Ohio/Michigan border, the central part of the state, and the western shore are poorly represented in collections. Even fairly-well studied areas such as the southeastern corner of Michigan have recently given up some surprises, such as *Anax longipes* and *Nasiaeschna pentacantha*. Persistence in examining a site on a daily basis may reward you with some very worthwhile records. This is why we are depending on others to be in the field. Obviously, the more eyes and hands we have, the more complete the survey will be. Hopefully, this handbook will be a useful reference and will aid your collecting efforts for the MOS.

If you would like to volunteer in the museum's Odonata collection, and learn by doing, let me know and I will arrange a time for us to meet. Working with the collection is an excellent way to learn curatorial methods and the taxa that we deal with.

There have been several suggestions about field trips, and hopefully the MOS will be able to set up a schedule for trips later in 1997. Send an e-mail or letter to the MOS to let us know if you'll be traveling somewhere to collect. Perhaps we'll be able to coordinate it so that some of us can meet in the field for an enjoyable outing.

The survey could not be more timely. Public interest in dragonflies is at an all-time high, and a new crop of enthusiasts is providing the impetus for new field guides, books, and surveys all over the U.S. I am hopeful that you, as a volunteer participant for the MOS will be able to not only help the MOS in whatever capacity that you can offer, but also to learn from us and with us about this fascinating group of gossamer-winged aerial acrobats.

Mark O'Brien

Contacts for the Michigan Odonata Survey

If you need to contact the MOS for anything, please call, write or e-mail any one of us, and the message will be passed on to the appropriate contact. If a park manager or other official needs more information about the MOS, have a copy of the one-sheet summary of our survey available. Additional sheets will be mailed on request. If you or someone you know is interested in doing any volunteer work for the survey, contact Mark or Mike.

Mark O'Brien

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Other Important Addresses

Dragonfly Society of the Americas
c/o Nick Donnelly
2091 Partridge Lane
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David Cuthrell
Michigan Natural Features Inventory
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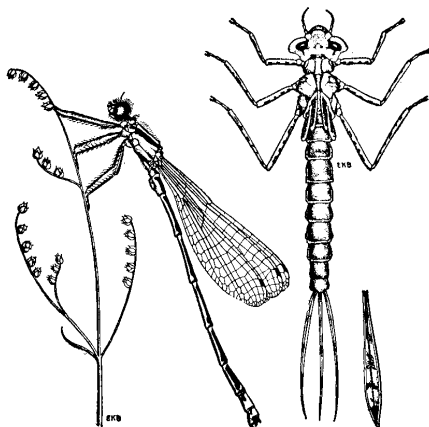
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ODONATA COLLECTION GUIDELINES FOR THE MICHIGAN ODONATA SURVEY

Mark F. O'Brien
UMMZ Insect Division

The Michigan Odonata Survey (MOS) is dedicated to providing a better understanding of the distribution, biology, and taxonomy of the Michigan Odonata fauna. To this end, MOS participants will be collecting in a wide variety of habitats all across the state of Michigan. Collected specimens are required to voucher the presence of a species at a particular locale, and the collection may comprise adults, larvae, exuviae, or all three stages. Collections of specimens form the basis of a permanent record of the fauna at a particular place and time. The specimens and their associated data can then be checked later for authentication or verification. They also may be useful to any future taxonomic studies. Therefore, collection of Odonata specimens is a very important aspect of the survey. With that in mind, I have put together this guide to form the basis of the protocols for the MOS.

MOS collectors are urged to collect no more specimens than necessary to document a species in a given area, with the exception of exuviae. However, due to the number of similar-looking species, certain genera such as *Somatochlora*, *Gomphus*, *Aeshna*, *Argia*, *Enallagma*, and *Sympetrum* may require more extensive sampling. Species that are quite obvious such as *Calopteryx maculata*, require only a single specimen from a locality. The collection of adults and larvae should be limited to representative sampling, not depleting the population at the area being surveyed. Sight records, especially photographs or video, may be acceptable for certain species that are readily and unquestionably identifiable in the field by trained, reliable observers. Obviously, a majority of the species of Odonata require more careful examination, necessitating collecting.



Ischnura verticalis (from Morgan, 1930)

The Michigan Odonata Survey

What is the Michigan Odonata Survey (MOS)?

The MOS, centered at the UMMZ, consists of a group of people with a shared interest in the study of the distribution, biology and enjoyment of the Odonata found in Michigan. It is chaired by Mark O'Brien and Mike Kielb, of the Museum of Zoology, University of Michigan. The Museum of Zoology provides some support for the survey.

What are the goals of the MOS?

The eventual goal of the MOS is to catalog and distribute definitive information on the Odonata fauna of Michigan. In doing so, we want to:

1. Promote the study and appreciation of Odonata at various levels of understanding.
2. Identify and promote the security of critical habitats where species of limited distribution or threatened status are found.
3. Provide a clearinghouse of odonatological information for the Great Lakes, especially Michigan.
4. Cooperate with agencies and groups where we share common goals.
5. Expand our knowledge of poorly-known species.
6. Survey the state of Michigan as thoroughly as is both possible and practical to document the current status of all species of Odonata.
7. Provide an internet-based source of information (WWW and e-mail list) as well as printed resources.
8. Produce a catalog of the Odonata of Michigan by 2002 or sooner.

How can you assist the MOS?

We need many volunteers to aid in our various projects. The MOS is a volunteer organization. Field assistants, collectors, data entry people, collections assistants and catalogers are welcome and encouraged. Collaborators from other institutions and

agencies are welcome to share collection data, including biological and environmental data, as well as specimens from sampling activities.

Since we currently have no funding and no paid positions, we ask that people volunteer their time or services to help accomplish our goals. We certainly will accept monetary donations made out to the Museum of Zoology, and gifts in kind are also welcome. We are trying to procure external funding for projects from various public and private sources.

Contacts for further information:

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Check out our WWW pages at:

<http://insects.ummz.lsa.umich.edu/michodo/mos.html>



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The University of Michigan
Ann Arbor, MI 48109-1079

313-647-2199

The MOS is not affiliated with any state or federal agencies.

The MOS requests that participants follow the
Dragonfly Society of the Americas Guidelines for Collecting (1996):

Purposes of Collecting

- 1.1 To create a reference collection for study and appreciation.
- 1.2 To document regional diversity, frequency and variability of species, and as voucher specimens for published records.
- 1.3 To document faunal representation in environments undergoing, or threatened with, alteration by human or natural forces.
- 1.4 To participate in the development of regional checklists and institutional reference collections.
- 1.5 To complement a planned research endeavor.
- 1.6 To aid in dissemination of educational information.
- 1.7 To provide specimens for taxonomic studies.
- 1.8 To provide information for ecological studies.
- 1.9 To provide a resource for DNA material for genetic analyses.

Restraints As To Numbers

- 2.1 Collection of adults or immatures should be limited to sampling, not depleting the population concerned.
- 2.2 Numbers collected should be consistent with, and not excessive for, the purpose of the collecting.
- 2.3 Where the extent and/or fragility of the population is unknown, caution and restraint should be exercised when collecting.

Collecting Methods

- 3.1 Field collecting should be selective and should minimize harm to non-target organisms.
- 3.2 Field collecting should be conducted in such a manner so as not to cause undue damage to fragile habitats such as seeps and fens.

Live Odonata

- 4.1 Rearing to elucidate life histories and to obtain series of immature stages and adults is encouraged, provided that collection of the rearing stock is in keeping with the guidelines.
- 4.2 Reared individuals in excess of need should be released, but only in the region where they originated, and in suitable habitat.
- 4.3 Concerns about the introduction of disease and the dispersal of non-indigenous genetic material, makes it critical that release of excess individuals in areas removed from the larvae's origin be conducted only with a planned restoration program under the supervision of knowledgeable biologists.

Environmental and Legal Considerations

- 5.1 Protecting the supporting habitat must be recognized as essential to the protection of a species.
- 5.2 Collecting should be performed in a manner that minimizes trampling or other damage to the habitat.
- 5.3 Property rights and sensibilities of others must be respected (including those of nature photographers and observers).
- 5.4. All collecting must be in compliance with regulations relating to public lands (such as state and national parks, monuments, recreational areas, etc.) and to individual species and habitats.
- 5.5 Importation and movement of exotic species must be in compliance with international, national, or regional laws prior to importing or exporting live or dead material.

Responsibility for Collected Specimens

- 6.1 All specimens should be preserved with full data attached, including percentage of immatures when known.

- 6.2 All specimens should be protected from physical damage and deterioration, as by light, molds, and museum pests.
- 6.3 Collections should be made available for examination by qualified researchers.
- 6.4 Collections or specimens, and their associated written, electronic, photographic and other records, should be willed or offered to the care of an appropriate scientific institution, if the collector lacks space or loses interest, or anticipates death.
- 6.5 Type specimens, especially holotypes or allotypes should be deposited in appropriate institutions.

Related Activities of Collectors

- 7.1 Collecting should include permanently recorded field notes regarding habitat, conditions, and other pertinent information.
- 7.2 Recording of observations of behavior and other biological interactions are encouraged.
- 7.3 Fully documented photographic records are encouraged.
- 7.4 Education of the public about collecting and conservation, as reciprocally beneficial activities, should be undertaken whenever possible.

Traffic In Odonata Specimens

- 8.1 Collection of specimens for exchange should be performed in accordance with these guidelines.
- 8.2 Rearing of specimens for exchange should be from stock obtained in a manner consistent with these guidelines, and be so documented.
- 8.3 The sale of individual specimens or the mass collection of Odonata for commercial purposes (e.g. fish bait), and collection or use of specimens for creation of marketable artifacts, are not included among the purposes of the Dragonfly Society of the Americas.

For more information about the Dragonfly Society of the Americas, and how to join DSA, write:

DSA, c/o Thomas W. Donnelly
2091 Partridge Lane,
Binghamton, NY 13903

SPECIFIC COLLECTING INSTRUCTIONS

EXUVIAE

Exuviae are the cast larval skins of the penultimate instar of Odonata. They provide important information about where species live and where they emerge. Since the larval characters are quite evident, most exuviae are identifiable to species level. Since exuviae are not living, participants not wanting to collect and kill living odonates can make a great contribution just by conducting surveys for exuviae along rivers, streams, and ponds. Exuviae indicate the presence of larval populations and past breeding populations at a particular locale, and therefore, they are valuable records for the survey. MOS participants are encouraged to collect exuviae and note the location and conditions where they are found. The UMMZ has an outstanding collection of Odonata exuviae, and many are catalogued in a database. Exuviae should be documented with as much care as adults and larvae.



Figure 1. *Macromia illinoensis* exuviae

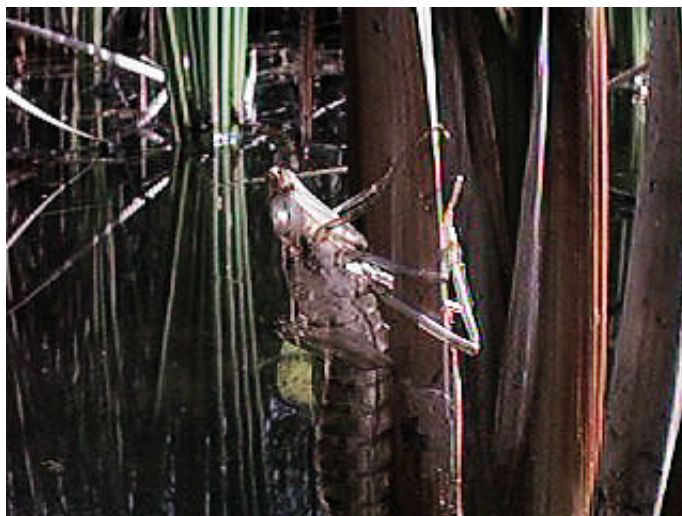


Figure 2. *Anax longipes* exuviae on cattail

LARVAE

Odonata larvae represent a different challenge for collectors. You must remember that most of the time they are unseen residents of aquatic systems. You will for the most part only catch them when sampling the streams, ponds, seeps, and lake bottoms. Try to minimize disruption to the site

where you are collecting. Unless you want to get wet, chest waders or hip boots are a good idea. Some aquatic situations are potentially dangerous to a lone collector, so it is advisable to collect with a companion. Bogs and fens and deep-muck-bottomed lakes can be places where a person collecting alone might get stuck for hours. Streams and rivers have their own potential dangers. If you can avoid collecting alone, please do so. Additionally, in or near metropolitan or mining areas, you might come into contact with polluted waters that are potentially hazardous. Waters might be polluted with organic matter or heavy metals or be very acidic. It is always wise to inquire with local health officials to see if any such warnings exist for streams, lakes, etc. that you may be working in. Again, a good set of chest waders provide a measure of safety.

For aquatic collecting, you will need a net designed for aquatic sampling, such as a D-net or similarly strengthened net with a long handle and strong nylon mesh (500-250 μ m mesh is best). You can also use standard kick-seines in riffle areas. Standard insect nets are not good for this purpose. (See the appendix for suppliers.) You can use a small dip net for pools and vegetated areas to catch species that are on the aquatic vegetation or near the surface. Seeps and very small streams are easily disturbed, so use as much care as possible when collecting in such habitats.

Samples can be dumped into a shallow pan (preferably a white enamel or plastic tray) to easily search for odonate larvae. Specimens to be preserved may be dropped into 70% ethanol or isopropanol. Specimens to be kept alive should be separated by size to avoid cannibalism and predation.

Make sure that data accompanies preserved specimens - use either pencil or India ink on rag paper (available from the MOS) to prevent fading or leaching of ink.

ADULTS

Collecting adult Odonata, especially the Anisoptera, can be quite challenging, and often, a lot of fun; it's a lot like fishing. Several hours spent on or in the water with only a few "keepers" to show for all the effort! Depending on where and what you collect, the techniques will differ. The most effective technique is to wait until the adult dragonfly is almost past you, and then swing the net from behind. A frontal assault nearly always end in favor of the dragonfly. Swinging the net at an oncoming dargonfly might also result in a headless or tailless specimen. The combined speeds are just too much for the exoskeleton if the specimen hits the ring of the net. Some species are more easily captured when they alight on a perch -- such as many Libellulidae. Others might be caught while they are basking on logs -- such as some gomphids and aeshnids. Corduliids are more easily captured early in the mornings or later in the afternoons to early evening. Again, a sweep from behind where the males are patrolling or female oviposition areas are more likely to succeed. Keep in mind that any observations you make on oviposition, mating, or predatory behavior are valuable, especially for some of the lesser-known species. Watching the habits of the species you see **before** you try to catch them will also yield greater success as well as useful behavioral information.

Several items are necessary for successfully collecting and preserving Odonata.

First, the major piece of equipment you'll need is an insect net with at least a 15" diameter hoop. Some collectors have been known to use a 36" diam. hoop with a 5 ft. handle! Most however, do fine with the standard 3 to 4 ft. handle. There is a debate on which color netting is best, - green, black, or white. This seems to be a matter of personal belief as to which is best.

Second, you'll need a supply of "paper triangles" (**Fig. 3**) to hold the captured adults. Place a single specimen per triangle, and record your field note number on the flap of the triangle (in pencil). Williamson (1916) recommended the use of newsprint to make the triangles.

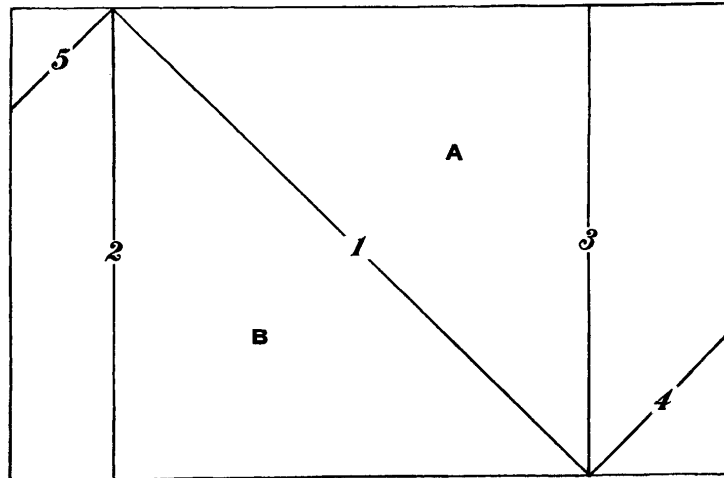


Figure 3. Template for making paper triangles (from Williamson 1916).

A porous paper is definitely recommended. You could also use the small kraft envelopes known as coin envelopes to store most specimens. If a collector just places the specimens in a killing jar, their wings may set in an unusual position after they die. Glassine envelopes such as those used by stamp collectors may also be used for field storage. (See Appendix 2 for more on this topic).

After the specimens have been placed in envelopes you'll need to kill and preserve them. One currently favored method is to place the entire paper triangle or envelope into a container of acetone. This method generally seems to remove some of the fats from the specimens and preserves many of the brilliant colors (except for the eyes). For very hairy specimens, such as some Corduliidae, this method may mat the hairs. Freezing specimens may also work, providing that they are air-dried afterwards as quickly and thoroughly as possible. Another common method of dispatching specimens is to place them in a killing jar, either charged with cyanide (the old-fashioned way), ethyl acetate, or acetone. Ethyl acetate is safer to use and easier to purchase, and is recommended for those not wanting to use the acetone method. There are some other more elaborate methods for killing and fixing specimens, and you can find out more on this topic via the Internet on the IORI web page (<http://www.afn.org/~iori/>). The most important aspect is to thoroughly dry your specimens as quickly as possible, without subjecting them to undue heat. Be sure to prevent the formation of mold on the specimens, as it will render them nearly useless. Perhaps even silica gel drying agents would be useful for this purpose. This is still a fertile field for experimentation on the best methods of color preservation.

In a nutshell, all you need to collect adult Odonata is a standard aerial insect net, a large peanut butter jar with an inch of plaster in the bottom soaked with ethyl acetate or acetone, a pencil, a field notebook, and many small paper triangles. The total cost for equipment should be less than \$20.00. The other thing you'll need is patience! That's priceless.

STORING SPECIMENS

Most large collections of Odonata are stored in a simple system using cellophane or plastic envelopes slightly larger than a 3 x 5 inch index card (**Fig. 4**). The card contains all of the data for the specimen arranged as illustrated. The specimen is placed in the envelope (well-dried, of course) and the flap is folded over on the right side. Specimens are stored either vertically on edge, or flat. There are tray systems in use that provide neat and organized methods of storage. For temporary

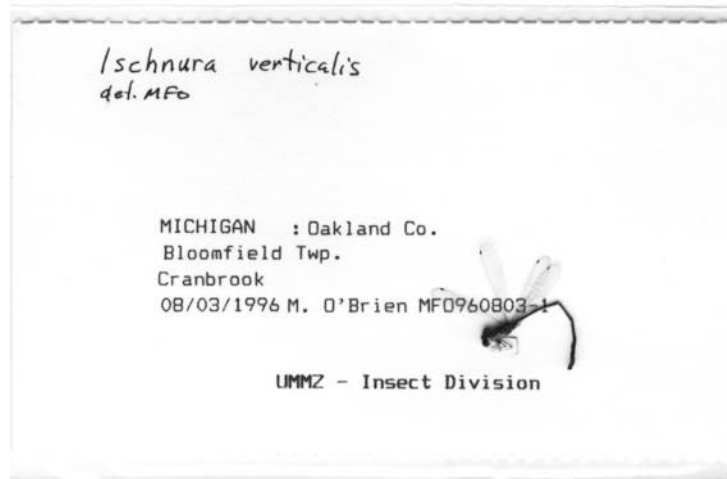


Figure 4. 3 x 5 card and envelope storage system.

use, plastic shoeboxes provide an easy and inexpensive storage system for the card and envelope method. Because they are not pest-proof, they are not a good idea for permanent storage. The plastic envelopes are very inexpensive and are available from the IORI in any quantity, and the MOS can make some available in small quantities to its participants. The MOS **does not** recommend pinning and spreading Odonata like butterflies. This method takes up too much space, makes the specimens harder to handle, and if any parts come off the specimen, they are easily lost. In fact, the 3x5 envelopes are cheaper than insect pins! If you are collecting for the MOS, it is acceptable to keep the specimens in paper triangles or envelopes and let the UMMZ take care of the transfer to the 3x5 system.

Storage of larvae and exuviae. At the UMMZ larvae are stored in glass screw-top vials and jars containing 70% ethanol (**Fig. 5**). However, if you cannot obtain ethyl alcohol, isopropyl may be used instead. **Never** use methyl spirits (wood alcohol). Rubber-stoppered vials may also be used. For the best protection of specimens, the use of Polyseal(R) caps are recommended on screw-top vials. Larger 2 and 8 oz. jars use polyethylene liners in the caps. These are available from a variety of sources. Labels are either handwritten in India-ink or printed with a multi-strike ribbon on a dot matrix or letter-quality printer. It is not advisable to use laser-printed labels because they are unreliable for long-term storage.



Figure 5. Larvae in stoppered vial.

Exuviae may be stored in a number of ways - inside vials, 3x5 envelopes, or pinned. Many collections store their exuviae in alcohol, or dry, in vials. The UMMZ has all three methods in use, depending upon the original condition and storage method of the exuviae. One practice is to place the exuviae into clear 3x5 envelopes with the data on the card. That way, all is visible, and if parts come loose, they stay inside the envelope. However, you have to be careful to avoid crushing the specimen. For this reason, storage in alcohol is a better method, especially if you want to manipulate the exuviae for measuring, etc.

FIELD NOTES AND DATA RECORDING

Of course, a specimen without data is practically worthless. At minimum, you should record the following for MOS purposes:

1. STATE and COUNTY

2. LOCALITY: nearest town, rd. miles from major intersection, State or National Forest, name of lake, etc.

3. TOWNSHIP, RANGE, & SECTION: If you have the MUCC map, this will be easy. You may also use geographic coordinates - latitude and longitude.

4. FEATURE: stream, bog, fen, river margin, etc.

DATE: Spell out or in this format: MONTH/DAY/YEAR - Spelling out or

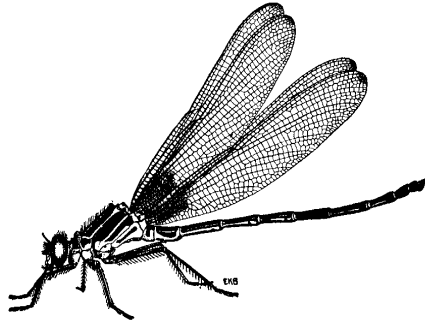
abbreviating the month is preferred. Roman numerals are also used for the month. Using a numeric for the month may confuse someone if the don't know that the US system is MM/DD/YYYY rather than the european preference of DD/MM/YY.

5. COLLECTOR: Your name or whoever collected the specimens.

6. COLLECTION NUMBER: It is a good idea to use a numbering system so that you can use a number in your field notebook that cooresponds to a specimen or lot of specimens from a single collecting event. My preferred system is simple: Your initials followed by the date in descending order and then a number or letter for the collecting event. Numbers are better for events if you anticipate more than 26 events in a day. Example: I have a specimen with the number MFO960702-1. This tells me that I collected it on July 2, 1996 and it was the first site I collected at that day. I may have collected 20 specimens representing 9 species from that collection. This way, all the specimens are correlated with that collection event. You may have another system, but I think the above is the most flexible and informative, if you apply it consistently.

7. NOTES: Provide any additional biological or ecological information here. Water temperature, stream or lake habitat type (riffle, undercut bank, pool, debris dam, etc.), substrate conditions (sand, muck, gravel, peat, boulder, woody debris, etc.), pH, turbidity, dominant vegetation, air temperature, cloud cover, wind, mating or oviposition behavior, etc.

This is basically all you need to provide for a rather complete set of data for a collection. Minimally, the basic locality and date information is quite acceptable.



Hetaerina americana (from Morgan, 1930)

FIELD NOTEBOOK

The above information should be kept in a field notebook. I like to use cloth-bound 5" x 8" notebooks with lined pages. "Blank books" are now often available from book outlets for less than \$4 each, and they have enough pages for a season's worth of collecting. Spiral-bound notepads don't hold up as well, but are acceptable. There are also waterproof field notebooks available from some vendors. Again, the Collection Number system described above works well, and is easy to use in your field notes. The above data should be kept in the journal, along with any other field notes that you think are worth recording.

IDENTIFICATION

Unfortunately, there are no Peterson-style field guides for Odonata. For many of the common species, this would probably work, but many Odonata require examination under magnification to determine the identity. In the past several years, there have been "local" field guides with excellent illustrations and photographs of living specimens. If you are just starting out, try to learn the identity of the common species first, then perhaps the species in a small area. As you work your way through the levels of identification, you'll gain more confidence in your ability to identify species on a larger scale. You do not necessarily even have to learn to identify specimens to the species level for the MOS, family and genus may be all that is needed so long as someone else does the determination of the species later on.

Larval Identification - The MOS will soon have a species-level guide for identification of larval Odonata found in Michigan. It will be available on the MOS WWW site. However, a printed guide will eventually also be available. For generic-level references, the following publications will be of use:

- Hilsenhoff, W. 1995. Aquatic Insects of Wisconsin: keys to Wisconsin genera and notes on biology, habitat, distribution and species. Natural History Museums Council, Univ. of Wisconsin-Madison, No. 3.
- Needham, J. G. and P.R. Needham. 1962. A guide to the study of freshwater biology. Holden-Day, San Francisco. 108 pp.
- Pennak, R.W. 1953. Freshwater invertebrates of the United States. Odonata, pp. 522-540. Ronald Press, NY. 769 pp.
- Westfall, M. J. and K. J. Tennessen. 1996. Odonata, pp. 164-211, *in*: Merritt, R. and Cummins. An Introduction to the Aquatic Insects of North America., 3rd. ed.. Kendall-Hunt Publishing Co., Dubuque, IA.

Adult Identification - The classic works on the Odonata of North America by Needham and Westfall (1955) and Walker (1953-1966) are hard to find, except in libraries. They rarely turn up in used book sellers. Unfortunately, this situation has made it difficult for amateurs to get authoritative works for identification of odonates. Recent efforts by various people have improved the

situation, especially the Westfall and May (1996) book on damselflies of North America. Guides to local faunas are also appearing and are aimed at the less-technically experienced audience.

The following recent (and in print) publications will be useful for identification of some of the adult Odonata that you'll encounter:

Carpenter, V. 1991. Dragonflies and Damselflies of Cape Cod. Cape Cod Mus. Nat. Hist. Series No. 4. 80 pp. Brewster, MA.
 Dunkle, S. W. 1989. Dragonflies of the Florida Peninsula, Bermuda and the Bahamas. Scientific Publishers, Gainesville. 155 pp.
 Dunkle, S.W. 1990. Damselflies of Florida, Bermuda, and the Bahamas. Scientific Publishers, Gainesville. 150 pp.
 Holder, M. 1996. The dragonflies and damselflies of Algonquin Provincial Park. Algonquin Park Tech. Bull. No. 11. 40 pp.
 Westfall, M. J. and M. L. May. 1996. Damselflies of North America. Scientific Publishers, Gainesville. 650 pp.

LITERATURE CITED

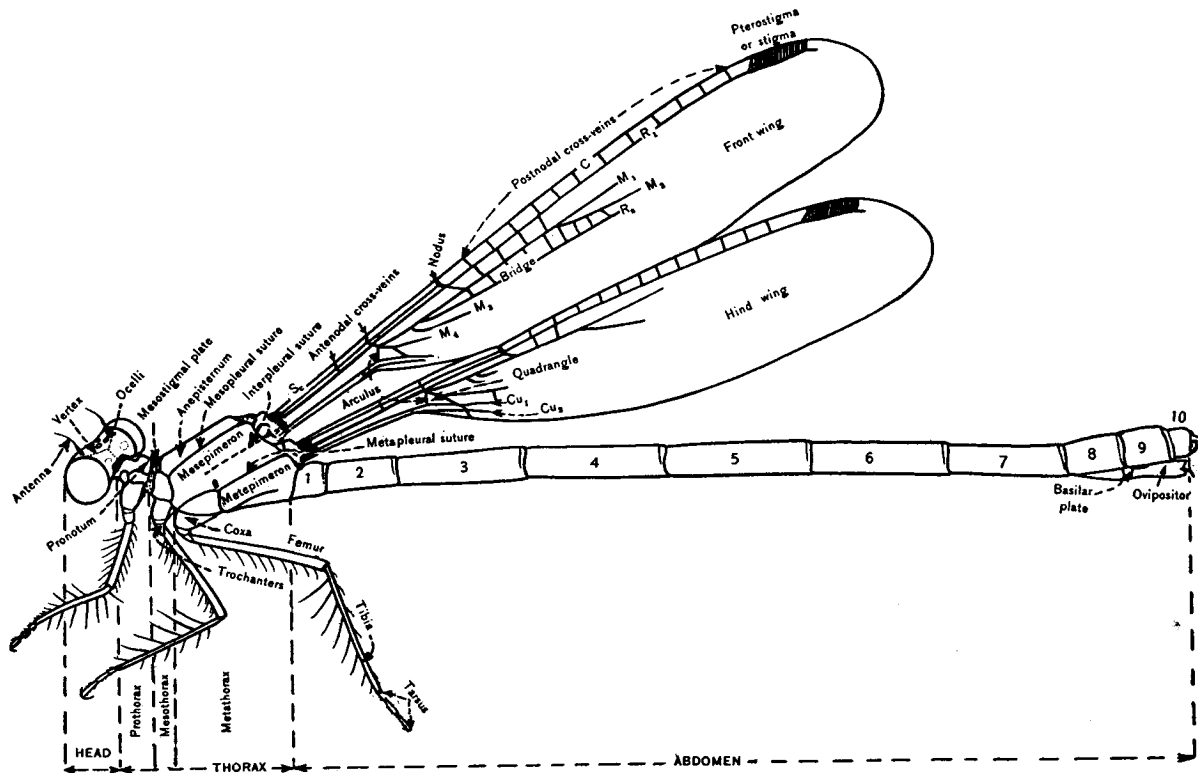
Garman, P. 1927. The Odonata or Dragonflies of Connecticut. Guide to the Insects of Connecticut, Part V. Hartford. 331pp.
 Morgan, Ann Haven. 1930. Field Book of Ponds and Streams. Putnam's and Sons, New York. 448 pp.
 Needham, J. G. and M. J. Westfall, Jr. 1955. A Manual of the Dragonflies of North America (Anisoptera). University of California Press: Berkeley, California. 615 pp.
 Walker, E. M. 1953. The Odonata of Canada and Alaska, Vol. 1. University of Toronto Press: Toronto, Ontario. 292 pp.
 _____ . 1958. The Odonata of Canada and Alaska, Vol. 2. University of Toronto Press: Toronto.
 Walker, E. M. and J. S. Corbet. 1975. The Odonata of Canada and Alaska, Vol. 3. University of Toronto Press: Toronto.
 Williamson, E.B. 1916. Directions for collecting and preserving specimens of dragonflies for museum purposes. Museum of Zoology, Univ. of Mich. Misc. Publ. No. 1, 16 pp.

Key to the Michigan Odonata Families

by Michael A. Kielb

Zygoptera

- 01** Antenodal crossveins numerous; postnodal crossveins not in line with the veins below them; anal vein at its base separate from posterior border of wing; quadrangle with several crossveins.....
 **Calopterygidae**
- 01a** Only two antenodals present; postnodals in line with the veins below them; anal vein joined with wing margin for a distance from the wing base; quadrangle never with crossveins..... **02**
- 02** Veins M₃ and R_s arising nearer the arculus than the nodus **Lestidae**
- 02a** Veins M₃ and R_s arising nearer the nodus than the arculus..... **Coenagrionidae**



Side view of adult Zygopteran body (from Garman, 1927).

Anisoptera

- 01** Triangles of fore wing less than twice as far from the arcus as those of the hind wing; triangles of both wings similar in shape, generally elongated in long axis of wing; two antenodal crossveins thickened, most of other antenodals of the 1st series not aligned with those of the 2nd series. **02**
- 01a** Triangles of fore wing at least twice as far from the arcus as those of hind wing; triangles of the fore wing generally elongated transversely, those of hind wing longitudinally; no thickened antenodal crossveins, those of the 1st series aligned with those of the 2nd series..... **05**
- 02** Eyes meeting for a considerable distance; stigma supported by an oblique brace crossvein at or very near its inner end; epiproct of male generally triangular, rarely forked or truncate; ovipositor well developed, both genital valves with a stylus..... **Aeshnidae**
- 02a** Eyes wide apart or barely meeting; stigma with or without a brace crossvein; epiproct of male usually forked or truncate, never triangular; ovipositor small or vestigial, rarely long; genital valves, if present, usually without stylus..... **03**
- 03** Eyes close together or barely meeting; labium with a medial cleft; stigma without a brace crossvein; ovipositor extending beyond tip of abdomen. **Cordulegastridae**
- 03a** Eyes wide apart; labium with or without a medial cleft; stigma with a brace crossvein; ovipositor never reaching tip of abdomen. **04**

Larval Morphology

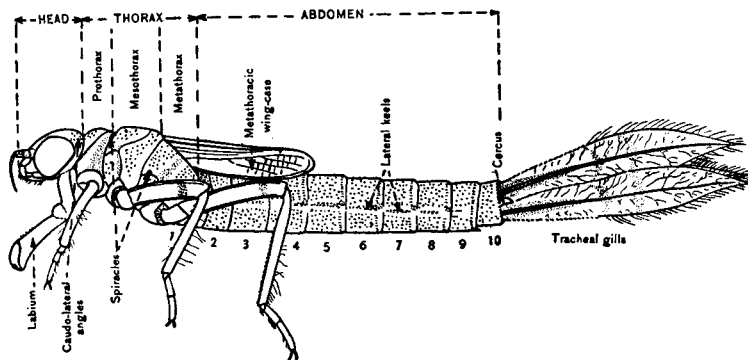


FIG. 5. Nymph of Zygopteron (*Ischnura verticalis*) showing parts.
from Garman, 1927

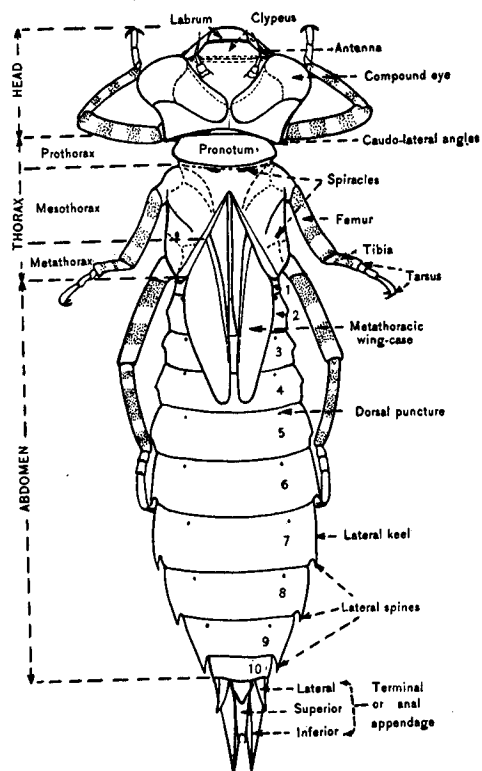


FIG. 6. Nymph of Anisopteron (*Basiaeschna janata*) showing parts.
from Garman, 1927

A KEY TO THE LARVAL ODONATA OF MICHIGAN (FAMILY LEVEL)

by Ethan Bright

This key is based on Ethan's key which will be on the internet. You can try it at:
<http://insects.ummz.lsa.umich.edu/michodo/test/home.htm>

1. External gills - three flat vertical plates-at tip of slender abdomen..... **Zygoptera**
2. Abdomen stout, usually with small spines at tip, gills are internal..... **Anisoptera**

Zygoptera - Damselflies

The 42 species of damselflies recorded in Michigan are divided into 3 families and 10 genera.

1. First antennal segment elongate, greater than segments 2-5 combined prementum with deep median cleft **Calopterygidae**

First antennal segment not so elongate, less than segments 2-5 combined; prementum with at most a tiny median cleft **2**

2. Basal half of labium greatly narrowed and elongate; folded labium extends back to mesocoxae or beyond (picture)..... **Lestidae**

Basal half of labium not greatly narrowed; folded labium extends back only to procoxae
..... **Coenagrionidae**

Anisoptera - Dragonflies

Recorded in Michigan are 109 species of dragonflies, which are divided into 6 families and 39 genera.

(Based on Hilsenhoff 1995; Westfall and Tennenssen 1996; Wright and Peterson 1944)

1. Mentum flat or nearly so , without dorsal premental setae **2**

Prementum and palpal lobes forming spoon-shaped structure **4**

2. Antennae 4-segmented, third segment often enlarged; fore- and middle tarsi 2-segmented; ligula without a median cleft **Gomphidae**

Antennae 6- and 7-segmented; fore- and middle tarsi 3-segmented; ligula with a median cleft **3**

3. Antennae segments (7) short, thick, and hairy; prementum with sides sub-parallel in distal three-fifths, abruptly narrowed near base; a pair of lateral-dorsal abdominal hair tuft present
..... **Petaluridae** (*Tachopteryx thoreyi*)

Antennal segments slender and bristle-like; prementum widest in distal half, then much narrower in basal half or more; lateral-dorsal abdominal hair tufts present **Aeshnidae**

4. Distal edge of lateral lobe with large, irregular teeth without associated setae; ligula with a median tooth-like cleft without dorsal premental setae **Cordulegastridae**, *Cordulegaster*

Distal edge of lateral lobe entire, or with even-sized dentations, with associated setae; ligula not as above 5

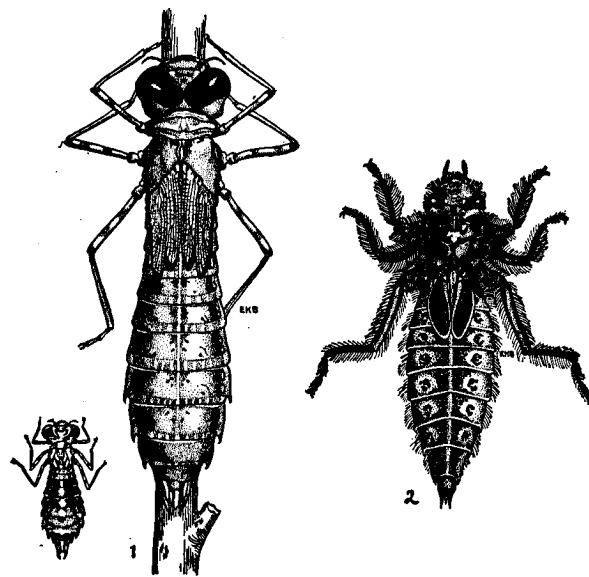
5. Head with thick, erect frontal horn positioned between antennae, and metafemur very long, reaching at least to apex of abdominal segment 8; metasternum with broad, median tubercle **Macromiidae**

Head without frontal horn positioned between antennae and metafemur not reaching apex of abdominal segment 8; metasternum without median tubercle..... **Corduliidae** and **Libellulidae**

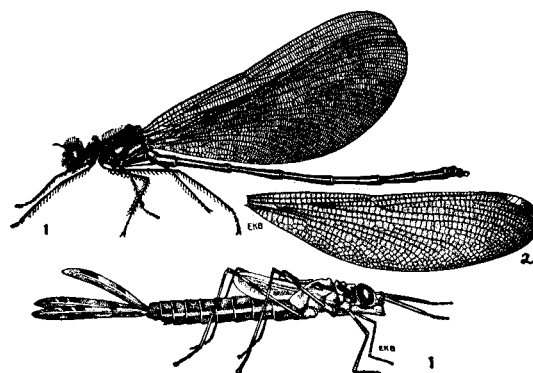
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From Morgan, 1930. *Anax* on left and *Gomphus* on right.



From Morgan, 1930. *Calopteryx maculata*

Michigan Odonata Survey

CHECKLIST OF MICHIGAN ODONATA

ZYGOPTERA

Calopterygidae

Calopteryx aequabilis Say
Calopteryx maculata (Beauvois)
Hetaerina americana (Fabr.)
Hetaerina titia (Drury)

Lestidae

Lestes congener Hagen
Lestes disjunctus disjunctus Selys
Lestes dryas Kirby
Lestes eurinus Say
Lestes forcipatus Rambur
Lestes inaequalis Walsh
Lestes rectangularis Say
Lestes unguiculatus Hagen
Lestes vigilax Hagen

Coenagrionidae

Amphiagrion saucium (Burm.)
Argia apicalis (Say)
Argia moesta (Hagen)
Argia sedula (Hagen)
Argia tibialis (Rambur)
Argia fumipennis violacea (Hagen)
Chromagrion conditum (Hagen)
Coenagrion interrogatum (Hagen)
Coenagrion resolutum (Hagen)
Enallagma antennatum (Say)
Enallagma aspersum (Hagen)
Enallagma basidens Calvert
Enallagma boreale Selys
Enallagma carunculatum Morse
Enallagma civile (Hagen)
Enallagma cyathigerum (Charp.)
Enallagma divagans Selys
Enallagma ebrium (Hagen)
Enallagma exsulans (Hagen)
Enallagma geminatum Kellcott
Enallagma hageni (Walsh)
Enallagma signatum (Hagen)
Enallagma traviatum (Selys)
Enallagma vesperum Calvert
Ischnura kelicotti Williamson
Ischnura posita (Hagen)
Ischnura verticalis (Say)
Nehalennia gracilis Morse
Nehalennia irene (Hagen)

ANISOPTERA

Petaluridae

Tachopteryx thoreyi (Hagen)

Cordulegastridae

Cordulegaster erronea Hagen
Cordulegaster maculata Selys
Cordulegaster obliqua (Say)
Cordulegaster (Zoraena) bilineata
 Carle
Cordulegaster (Zoraena) diastatops
 (Selys)

Aeshnidae

Aeshna canadensis Walker
Aeshna clepsydra Say
Aeshna constricta Say
Aeshna eremita Scudder
Aeshna interrupta Walker
Aeshna juncea Linn.
Aeshna mutata Hagen
Aeshna sitchensis Hagen
Aeshna subarctica Walker
Aeshna tuberculifera Walker
Aeshna umbrosa Walker
Aeshna verticalis Hagen
Anax junius (Drury)
Anax longipes Hagen
Basiaeschna janata (Say)
Boyeria grafiana Williamson
Boyeria vinosa (Say)
Epiaeschna heros (Fabr.)
Gomphaeschna furcillata (Hagen)
Nasiaeschna pentacantha
 (Rambur)

Gomphidae

Dromogomphus spinosus Selys
Gomphus brevis Hagen
Gomphus cornutus Tough
Gomphus exilis Selys
Gomphus fraternus (Say)
Gomphus furcifer Hagen
Gomphus graslinellus Walsh
Gomphus lineatifrons Calvert
Gomphus lividus Selys
Gomphus quadricolor Walsh
Gomphus spicatus Hagen
Gomphus submedianus
 Williamson
Gomphus vastus Walsh
Gomphus ventricosus Walsh
Gomphus villosipes Selys
Gomphus viridifrons Hine
Hagenius brevistylus Selys
Ophiogomphus aspersus Morse
Ophiogomphus carolus Needham
Ophiogomphus colubrinus Selys
Ophiogomphus howei Bromley
Ophiogomphus rupinsulensis
 (Walsh)

Progomphus obscurus (Rambur)
Stylogomphus albistylus (Hagen)
Stylurus amnicola Walsh
Stylurus laurae Williamson
Stylurus notatus Rambur
Stylurus plagiatus Selys
Stylurus scuderi Selys
Stylurus spiniceps (Walsh)

Macromiidae

Didymops transversa (Say)
Macromia illinoensis Walsh
Macromia taeniolata Rambur

Corduliidae

Cordulia shurtleffi Scudder
Dorocordulia libera (Selys)
Epicordulia princeps (Hagen)
Epitheca canis MacLachlan
Epitheca cynosura (Say)
Epitheca spinigera Selys
Neurocordulia obsoleta Say?
Neurocordulia yamaskanensis
 (Provancher)
Somatochlora cingulata (Selys)
Somatochlora elongata (Scudder)
Somatochlora forcipata (Scudder)
Somatochlora franklini (Selys)
Somatochlora incurvata Walker
Somatochlora kennedyi Walker
Somatochlora linearis (Hagen)
Somatochlora minor Calvert
Somatochlora tenebrosa (Say)
Somatochlora walshi (Scudder)
Somatochlora williamsoni Walker
Williamsonia fletcheri Williamson

Libellulidae

Celithemis elisa (Hagen)
Celithemis eponina (Drury)
Celithemis monomelaena
 Williamson
Erythemis simplicicollis (Say)
Leucorrhinia frigida (Hagen)
Leucorrhinia glacialis Hagen
Leucorrhinia hudsonica (Selys)
Leucorrhinia intacta (Hagen)
Leucorrhinia proxima Calvert
Libellula cyanea Fabr.
Libellula incesta Hagen
Libellula julia (Uhler)
Libellula luctuosa Burmeister
Libellula lydia (Drury)
Libellula pulchella Drury
Libellula quadrimaculata Linn.
Libellula semifasciata Burm.
Nannothemis bella (Uhler)

Pachydiplax longipennis (Burm.)
Pantala flavescens (Fabr.)
Pantala hymenaea (Say)
Perithemis tenera (Say)
Sympetrum ambiguum (Rambur)
Sympetrum corruptum (Hagen)
Sympetrum costiferum (Hagen)
Sympetrum danae (Sulzer)
Sympetrum internum Montgomery
Sympetrum obtrusum (Hagen)
Sympetrum occidentale Bartenev
Sympetrum rubicundulum (Say)
Sympetrum semicinctum (Say)
Sympetrum vicinum (Hagen)
Tramea carolina (Linn.)
Tramea lacerata Hagen

**Species to look for but not yet
 recorded from Michigan:**

LESTIDAE:
Archilestes grandis (Rambur)
 CORDULIIDAE:
Somatochlora hineana Williamson

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Some Dragonfly Terms

Dennis R. Paulson

- abdomen** - last segment of body, the long and slender one, with 10 segments
- Anisoptera** - suborder to which "true" dragonflies belong
- appendages** - structures at the end of abdomen: 2 in females, 3 (2 superior, 1 inferior) in male dragonflies, 4 (2 superior, 2 inferior) in male damselflies; distinctive of species
- caudal lamellae** - three leafy appendages at rear of abdomen in damselflies, for respiration and locomotion; also called caudal gills
- copulation** - act of fertilization, may be brief or lengthy; male holds female as in tandem, but tip of female's abdomen swings up to contact second segment of male, where accessory genitalia located (sperm transfer already accomplished) cuticle - outermost covering of insect
- damselfly** - member of suborder Zygoptera, characterized by narrow wings, fore and hind identical, usually folded over abdomen; small, widely separated eyes; and small size and slender body
- dragonfly** - member of suborder Anisoptera, characterized by broad wings, fore and hind different, held out from body; large eyes, touching in most groups; and typically large size and bulky body; also used to refer to the entire order
- emerge** - to leave water and undergo metamorphosis into an adult; emergence is thus both from water and from exuviae
- endophytic oviposition** - laying eggs into plant tissue
- exophytic oviposition** - laying eggs onto water or land
- exoskeleton** - outer hard part of an insect, including legs & wings
- exuviae** (sing. & pl.) - cast skin from any larval molt (including transformation into adult)
- femur** - first (basal) large leg segment
- flight season** - period during which adults occur
- guarding** - behavior by male to keep other males away from ovipositing female he has fertilized
- hamules** - paired structures that project from pocket under second segment and hold female abdomen in place during copulation
- imago** - sometimes-used term for adult stage
- immature** - adult past teneral stage but still not with mature coloration; usually not at water
- in copula** - collected while copulating
- instar** - a larval stage; most larvae go through 10-13 of them
- interspecific** - between different species
- labium** - lower "lip" of larva that is extended during prey capture
- larva** (pl. larvae) - immature stage of Odonata
- maiden flight** - first flight of teneral away from water
- mandibles** - what dragonfly bites with
- mature** - of reproductive age, fully colored
- metamorphosis** - process of changing from larva to adult; happens within larval exoskeleton
- molt** - each time exuviae is shed; larval growth can take place only when larva is briefly soft at this time
- naiad** - another term for larva, not much used
- nymph** - another term for larva, commonly used
- ocellus** (pl. ocelli) - simple eyes between the large compound eyes
- Odonata** - order to which dragonflies/ damselflies belong
- odonate** - another term for dragonfly/ damselfly
- ommatidium** (pl. ommatidia) - one division of compound eye
- oviposit** - to lay eggs
- oviposition** - act of laying eggs

ovipositor - complex structure at posterior end of female damselflies, darners, and petaltails that functions in endophytic oviposition

pruinose - with powdery bloom that exudes from cuticle and turns it light blue, gray or white

pruinosity - powdery bloom deposited on mature odonates of some species

pterostigma - thickened structure at front of wingtip in most Odonata; function still in question; often called "stigma," but that is the term for the holes in the side of an insect through which aerial respiration takes place

sexual patrol flight - characteristic flight of male odonates at water; either actively searching for females or defending a territory into which females may enter

sperm transfer - when male transfers sperm from tip of abdomen to accessory genitalia on 2nd abdominal segment

tandem - when male and female are linked either in flight or at rest; male damselfly's 4 appendages clasp female at junction of prothorax and thorax; male dragonfly's 3 appendages clasp female at back and top of head

tarsus - third leg segment, made up of several short segments

teneral - adult after it has just emerged, soft and not definitively colored

territoriality - active defense of a small area

thorax - second section of body, bulky part bearing wings and legs

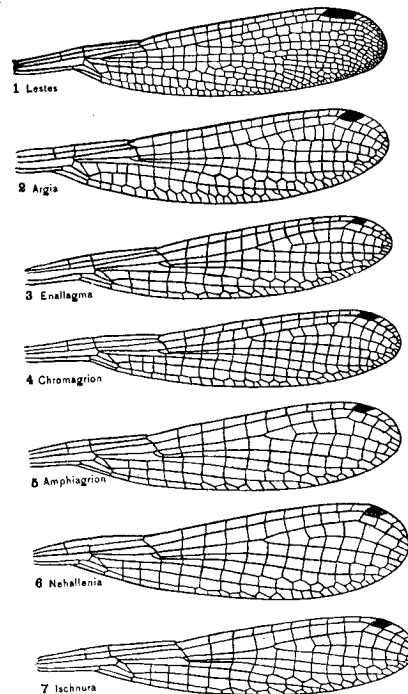
tibia - second leg segment, usually longer and thinner than femur

vulvar lamina - plate under 9th abdominal segment of female odonates that serves to hold eggs in place during exophytic oviposition; distinctive of species

wing sheaths - contain developing wings on thorax of larva; swollen when emergence is near

"wheel" - term often used for the copulation position

Zygoptera - suborder to which damselflies belong



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Appendix 2. More on Collecting Techniques (from Terry Morse's summary on the IORI web site.)

Sid Dunkles Method (from "Dragonflies of the Florida Peninsula ..", p. 13–14):

- 1) The most important aspect of collecting is recording detailed information on the day of collection. Include the state, county, nearest town, nearest water body, nearest highway, date, time of day, weather, name of collector, habitat notes, and behavior notes. [See Herman (1986) for a useful method of scientific note-taking—TM] Of particular scientific interest are pairs associated in tandem or in wheel (be sure to record this fact), and adults associated with the larval skin from which they emerged. In the latter case, let the adult harden for a day in a brown paper bag before preserving.
- 2) Choice of net: It should be large, strong, and as light-weight as possible. Sid uses a modified fish-landing net with ca. 50cm hoop, 2m handle, and a net bag with mesh size a few mm across for reduced air resistance. Dark material is probably best for the net, and it should be tough enough to resist thorns.
- 3) Approaching dragonflies: Approach slowly, then swing the net as fast as possible from the insect's rear. Don't make a shallow swing: follow through until the dragonfly is deep in the bag, then twist the handle to close the net entrance. [For dragonflies which rest on the ground, e.g. some *Sympetrum*, I approach slowly from behind, sit down perhaps 4' behind the dragonfly, and **slowly** edge the net over the ground (at ground level) toward it. When the net is 1–2" behind the 'fly, I rest it on the ground for a few moments, so the dragonfly will get over any nervousness caused by my approach and the movement of the net. When I think the dragonfly has relaxed its guard, I quickly swoop the net a few inches up and over the dragonfly, almost always catching it. When I miss, it's usually due to impatience: either I tried to pounce when the net was too far away from the dragonfly or without giving it adequate time to settle down before striking—TM]
- 4) Removing the dragonfly from the net: Hold the wings together over its back and keep the jaws from clamping onto the net (a dragonfly will often hold the net with its jaws until the head is pulled off). Place the dragonfly in an envelope with its wings over its back and the abdomen straight (if it can curl its abdomen to its mouth, it will chew the end of the abdomen off). Place the envelope in a crush-proof container and protect it from excessive heat until you get home. Killing it in the field is not advisable, since it will begin to lose color before you can commence preservation.
- 5) Killing methods: Dropping the dragonfly in a wide-mouthed bottle filled with acetone works in a few seconds. Freezing also works, but blue and green colors will darken, and the specimen will decompose rapidly when thawed.
- 6) Preservation: After killing, arrange each specimen with wings above the back and abdomen straight in a paper envelope. Write collection data on the envelope, then place the entire envelope in a jar of acetone. Beware of acetone's flammability! Don't breathe the fumes! Soak for 24 hours, remove from acetone, and dry as described previously. Some fading of colors will occur, and eye color will not be preserved, so make notes on coloration before preserving. [Smithe, 1975 is useful for standardizing color descriptions.—TM]
- 7) Storage: Store specimens in paper or glassine envelopes or, better, in transparent cellophane envelopes containing a 3"x5" data card. Store envelopes on edge like cards in a cardfile in tightly closing plastic containers, with a few naphthalene moth balls (paradichlorobenzene dissolves some kinds of plastic) for pest control. Do not pin dragonflies because the head and abdomen are prone to breakage if not supported.
- 8) Mailing specimens: Place the specimens in their envelopes in a small box with padding to prevent them from sliding against each other. Nest this box in a large box with padding, and label the outer box "Fragile" and "Contents: Dead Insects, No Commercial Value."
- 9) Be sure to state in your will who gets your collection when you go to that great acetone bottle in the sky [:) —TM]

In 1995, I put a follow-up question to Entomo-l: "In looking at older collections of pinned dragonflies, I notice that color seems to be better preserved in many of them than in the specimens I treat in acetone. Is acetoning really the best method of preservation?"

Dennis Paulson replied:

"I suspect the dryness of the surrounding air is critical in air-drying odonates. Anything placed in a rather rigorous desiccating atmosphere probably dries quite well, and some of my air-dried odonates look pretty good. Most of them, however, haven't held their color as well as acetone-preserved specimens. Nor are they as strong (structural integrity), which I consider an equally valid reason for using acetone. That's even if you discount the (somewhat?) lower likelihood of acetone-dried specimens to be subject to pest attack. Acetone supposedly also takes some of the fat out of specimens, and fat definitely causes deterioration. This may be a problem only in migratory species. I've had *Pantala* and *Tramea* and some others ooze grease until they discolored much of the index card on which they were stored. It wasn't until I had been replacing these cards for some years that it dawned on me that they represented some sort of index to fat content, which probably related to migratory status! When we prepare birds we always record the amount of fat, which is highly correlated with migratory status. With acetoning, we're now probably losing that information. It may be that pinned specimens experience a lower humidity than those stored in glassine envelopes, which of course is the way I've always collected mine. Thus pinned specimens on the average may look better than those originally stored in envelopes.

I think there is a great variation in the way acetoned specimens turn out, depending certainly on the amount of use the acetone has already had, but perhaps even on the acetone itself. A friend of mine bought some in a hardware store this summer, and I was amazed that his specimens didn't look all that good. When he got a new supply, all of a sudden they were fine.

I have had some acetoned specimens in which the eye color preserved moderately well, in most the eyes turn dark, in still others they become very pale. Perhaps again either age or type of acetone. I am pretty consistent in how long I leave specimens in it, ca 24 hours, although I've found I can leave them in for only 12 and they still preserve all right. Might be worth experimenting more with this."

And Bill Mauffray wrote:

"The whole key is drying the Odonate specimen as quickly as possible. Before Acetone, I used to place my newly collected dragonflies in pastic cases in the direct sunlight on my dash board. I would remove from the sunlight before they got crispy. Then I would allow them to air dry in PDB fumes for a few days. The air drying method worked for most of the greens,browns and yellows as well as powdery pruinose colors like bleus and whites. It did not wor at all for the blues in *Argia* and other zygopts. The key here is beng able to dry the specimens rapidly. In the humid parts of the US and the tropics, the moist air works against this method. You have to use acetone to preserve color paterns here in Florida and in the tropics. It is true that Acetone destroys the eye color, and pruinose colors (if the acetone is not clean). I still hold out the pruinose specimens for air drying. It would be nice if we could find another substance that would allow for quick drying, color preservation in both eyes and body, and be readily available no mather where you go (Acetone is hard to get in some countries)."

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Appendix 3. Databasing Collections

Instructions for DOS users of the MOS Database Template

by Mark O'Brien, 10/31/1996

The DOS version of the MOS database template may be downloaded from our web site, or we can send a disk in the mail to you. For MOS people without an internet connection, the disk in the mail is the easiest solution.

The DOS version of the MOS database is a zipped file, with all of the files needed except the database engine, which in the case of this template, can be any dBase version or a clone. dBase III was used to generate the entry screen and format, since it requires very little in the way of computing "power." If you have a DOS-based PC laptop or simple 640K 8088-class desktop machine, you will be able to use dBase III or a variant of it. The template has fields for all of the basic information that is needed. Macintosh users can also use the Shareware DABase desk accessory which allows one to access .dbf files on a Mac. Contact Mark O'Brien if you would like a copy.

1. Insert the 3.5" disk into your drive (usually A)
2. At the C: prompt, type DBASE (the following instructions assume dBase III)
3. After the initial dbase III screen, type ASSIST at the . prompt.
4. Under the Set Up Menu:
 - Select Database file, Search the A: drive
 - Select SPPLIST.DBF for your database.
 - Hit Return when it asks if the file is indexed.
 - Select Format for Screen (Again, the A: drive)
 - Select ODOENTRY.FMT
5. Under the Update Menu:
 - Select Append
 - You are now ready to enter records into the MOS Database Template.

Entering Data

Try to be as concise as possible, retaining as much data as possible, without losing that which is important. In other words, be brief but accurate.

FAMILY Field:

Here are the recognized families in Michigan:

Calopterygidae
Lestidae
Coenagrionidae
Petaluridae
Cordulegastridae
Aeshnidae
Gomphidae
Corduliidae
Macromiidae
Libellulidae

GENUS Field - Enter the Genus

SPECIES Field - Enter the species name, if known

AUTHOR Field - author of the species, if known.

DET_BY Field - Who identified the specimen, and in what year?

Locality Data:

STATE: (should be MICH)

COUNTY: (required, add Co. after name)

LOCALITY: (name of nearest town, river name, Hwy, etc.)

TWP/RGE/SEC: Enter the Township, Range and Section Numbers, Please! Get a good map, if necessary.

COORD: Use UTM Coordinates, if possible, but conventional lat. & long. okay.

DATE: MONTH DAY YEAR, as in Aug 12, 1996 is: 08/12/96

COLLECTOR: Name of Collector(s).

FIELD_NO: If there is a field note number, enter it.

Other Data:

STAGE: AD for ADULT; LA for LARVAE; EX for EXUVIAE; EX+AD for EXUVIAE and ADULT.

SEX: M for Male, F for Female

NO. OF SPECIMENS: How many?

CATALOG NO.: Enter the MOS##### if there is one, or the other institutions' specimen number if present. (NOTE: To use MOS numbers, you must obtain them from Mark O'Brien)

ENTERED BY: your initials

COLL FROM: Where did the specimens come from?

UMMZ - Univ. of Michigan Museum of Zoology

MSUC - Michigan State

ISMC - Illinois State Museum

CSCB - Cranbrook Science Center, Bloomfield

ACIC - Adrian College

MNFI - Michigan Natural Features Inventory

WMUC - Western Michigan Univ.

UMBS - Univ. Michigan Biological Station

NDIC - Notre Dame

NMUC - Northern Michigan University

EMUC - Eastern Michigan University

UMDC - Univ. of Michigan Dearborn

UMFC - Univ. of Michigan Flint

ALBC - Albion College

ALMA - Alma College

OSUC - Ohio State Univ. Collection

ODSC - Ohio Dragonfly Survey Collection

HRWC - Huron River Watershed Council

FMNH - Field Museum of Natural History

___ - enter the last name if a private collection

NOTES: Whatever else is pertinent; time, behavior, in copula, etc.

Once you have entered the data, you'll go to the next record.

NOTE: ENTER KEY TAKES YOU FROM FIELD TO NEXT FIELD

If you have any questions, call Mark O'Brien or email: mfobrien@umich.edu

MOS Distribution Maps

The following pages of maps are derived from our “working copy” of the distribution maps. The maps are based on what we knew as of the end of 1996, so any recent county records are probably not included. The master copy in the MOS files will be continually kept up-to-date.

Feel free to use these maps as a basis for determining where you should collect and plan for future surveys. As you might expect, most of the county records are based on Kormondy, 1957. Any additions have been marked with an “X”. Hopefully, within a short time, we’ll see a lot more records represented by an X. Just because a species is not marked on a county, does not mean it is not known from there. It may be in our database as a new record, or, more likely, it is not yet known from that county due to incomplete collecting. Look at the overall range of a species to get a feel for its possible distribution in Michigan.

