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Stephen D. Gaimari, editor
Plant Pest Diagnostics Branch
California Department of Food & Agriculture
3294 Meadowview Road
Sacramento, California 95832, USA
Tel: (916) 738-6671
FAX: (916) 262-1190
Email: stephen.gaimari@cdfa.ca.gov

Welcome to the latest issue of *Fly Times*! You may have noticed in the newsletter information above that I've changed the issue-timing to "Fall" instead of "October" (and will similarly change the "April" issue to "Spring" from now on). Recognizing that issues are rarely (if ever) actually published in October or April, I thought it was high time to lose the pain of being late every time! I will still be soliciting articles on the same schedule, and shooting for April/October, but there will be a bit broader window for me to work with. But rest assured, I WILL NOT produce Fall issues after 20 December! Nor Spring issues after 20 March!

Thank you to everyone for sending in such interesting articles! I encourage all of you to consider contributing articles that may be of interest to the Diptera community, or for larger manuscripts, the Supplement series. *Fly Times* offers a great forum to report on research activities, to make specimen requests, to report interesting observations about flies or new and improved methods, to advertise opportunities for dipterists, to report on or announce meetings relevant to the community, etc., with all the digital images you wish to provide. This is also a great place to report on your interesting (and hopefully fruitful) collecting activities! Really anything fly-related is considered. And thanks very much to Chris Borkent for again assembling the list of Diptera works since the last *Fly Times*!

The electronic version of the *Fly Times* continues to be hosted on the North American Dipterists Society website at http://www.nadsdiptera.org/News/FlyTimes/Flyhome.htm. Also note, the *Directory of North American Dipterists* is constantly being updated, so please check your current entry and send all corrections (or new entries) to Jim O'Hara – see the form for this on the last page.

Issue No. 64 of the *Fly Times* will appear next Spring. Please send your contributions by email to the editor, aiming for the end of April 2020. I will send a reminder. And articles after April are OK too!

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NEWS

Residual Fragments of Past Lives (Patagonia's Untold Stories)

R. Isaí Madriz

Casilla 367 Correos Coyhaique, 5950000, Region de Aysén, Chile; rimadriz1@gmail.com

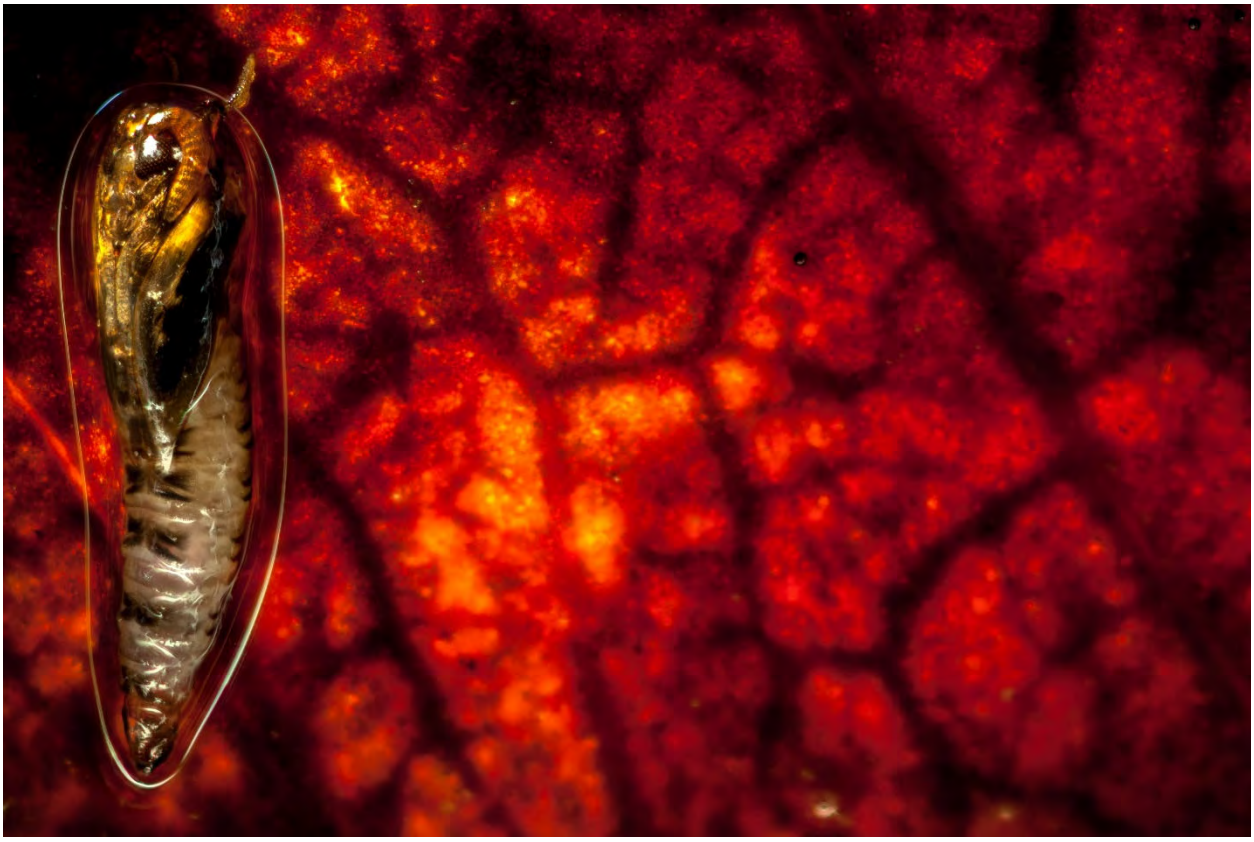

Figure 1. Stacked image of *Feuerborniella* sp. (Psychodidae) pupa. Photo by R. Isaí Madriz.

Decaying fragments hang from the cavity's roof as streaks of viscous dark red liquid meander down its sides.

The peculiar scent of extracellular digestion from fungal mycelia impregnates the stale air.
I force myself into the cramped cavity to further investigate. With only half of my body inside, I sit motionless, looking up, in awe. The sound of silence is overwhelming.

The bifurcating trunk bears equally tall galleries. Water percolates high above, saturating the substrate.

Insects feeding on decayed organic matter have turned the surface cellulose into a pulp. Partially digested body segments of forgotten generations provide vital nutrients for those in current development.

Under the dim light passing through the large crack, I observe predatory beetles moving across my body, in search of tender larvae inhabiting the collapsed fragments from above. I cover most of the small aperture to the outside, trapping myself in darkness. As I breath shallowly to fit in the claustrophobic space, the sound of intermittent water droplets impacting my forehead break the silence.

Inches away, a secret is being unveiled before my eyes. A new species of fly inhabits the tree cavity. Their unknown anatomy and behavior is revealed for the first time through the green hue of my night vision goggles.

Mature larvae 3mm long move below the organic matter, consuming the moist pulp. Their body segments are decorated with exquisite intricate patterns. Rows of hairs arranged throughout their dorsal side help trap the moist substrate.

Figure 2. *Feuerborniella* sp. (Psychodidae) mature larva (front) moving on top of decaying organic material, with pupae (back) ready to emerge. Photo by R. Isaí Madriz.

Predatory slugs and spiders patrol the vertical surface. Below, hidden by the soft mass on their backs, larvae use their posterior respiratory siphons to acquire vital oxygen from above.

Lethargy signals the initiation of metamorphosis. Their heads soon become an empty shell as their organs retreat inside their thorax to rearrange one last time.

As the insects develop inside their pupal skin, they outgrow their old larval skin, splitting it down its sides. They slowly migrate to the surface, where they are most vulnerable. The thin layer of moist

substrate on their backs is their only defense against predators. There, they remain face down until their metamorphosis is complete.

Figure 3. *Feuerborniella* sp. (Psychodidae) pupae breaking out of their larval skin (left & right). Mature larva (middle). Photo by R. Isaí Madriz

An individual is ready to emerge. The pupa slowly pushes itself out of the old larval skin. With its exposed head capsule and thorax, the emergence commences. The skin splits down the middle, from its forehead down its back, tearing its face off. Its larval head capsule and front half of its thorax hang perpendicular to its body.

With controlled movements from its abdomen, it pushes itself free. Its legs spread for the first time. It uses its tender limbs to pull its compressed abdomen from its old skin. It is a female. She hangs on to the vertical wall until her skin hardens. Her larval and pupal skins are now part of the cryptic biological memoir inscribed in the lugubrious facade. Soon after her emergence, a male begins to court her. The pair quickly disappear into the dark gallery above.

Down below, through the crack in the bark, daylight beckons those curious enough to investigate. With a short and sharp flight pattern, a female slowly migrates down towards the glare. As she moves downward, the moist substrate becomes dry and brittle, a metaphor of what is to come in her life.

Near the entrance, residual fragments of past lives hang from abandoned spider webs. The female reaches the gateway to the exterior, stopping at the divisory line between light and darkness. Her antennae are gently swayed by the soft breeze caressing the bark.

I can only wonder how she perceives the world outside. Is she aware of the exterior's expanse? If she only knew hers is one of the few remaining old-growth trees in the area. Can she grasp the magnitude of the decision she is about to make?

At that very instant, she leaps onto a wind gust and leaves behind the only world she has ever known.

This story was originally posted on the National Geographic Blog.

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Unidentified Central and South American Diptera in the Oxford Museum of Natural History (OUMNH), or Awesome flies and where to find them

Zoë M. Simmons[1] & Martin J. Ebejer[2]

[1] Oxford University Museum of Natural History, Parks Road, Oxford, OX1 3PW, England; zoe.simmons@oum.ox.ac.uk

[2]Hon. Research Fellow, National Museum of Wales, Cathays Park, Cardiff, CF10 3NP, UK; martin.ebejer@btinternet.com

A species of *Somatia* (Somatiidae) that until recently was “lost” in the collections.

In late 2017, prompted by the increase in fly fervour for the then incoming 9th International Congress of Dipterology (Windhoek, Namibia) and 2019 Year of the Fly, an overview of the Diptera collections held at OUMNH was undertaken with a view to forecast and plan collections work over the next decade. One of the strands for this plan encompassed the enigmatic goal of ‘tackling the specimen backlog’. Like most major museums OUMNH has an accumulation of both wet and dry by-catch material awaiting processing after various field expeditions. Whilst these samples are well stored, they remain relatively inaccessible due to the nature of that storage and lack of identification even to family level. It was felt however that a portion of staff time should be devoted to unlocking at least some of its research potential, capitalising on the buoyant mood of the dipterological community. Thankfully the museum, whilst not currently employing a dipterist, has a long tradition of dipterological work and links remain strong with researchers around the world. Coupled with the timely advent of the OUMNH Fellowship scheme, launched in spring 2018 this provided a hitherto unprecedented opportunity to secure expert help.

The following lists some of the material available from expeditions that the OUMNH organised or participated in. This is by no means an exhaustive list but rather reflects that which was most easily accessed at the commencement of MJE’s tenure:

Bolivia

i) August – November 2003; A.C. Hamel
ii) November – December 2003; A.C. Hamel and D.J. Mann
iii) November – December 2004; A.C. Hamel, D.J. Mann and Z.M. Simmons
iv) January 2005; A.C. Hamel, S. Herzog and D.J. Mann

Capture techniques: Malaise, MV light, blue light, pitfall, sweeping
Bait: human faeces; pig dung; chicken dung; fish carrion

Venezuela
March 1991; G. McGavin
Capture technique: Malaise

Honduras
July 2007; J. Nuñez-Mino
Capture technique: Flight intercept

Costa Rica
July 2009; Cowburn, Yu, Mehrabi and Coals
Capture technique: Pig dung PF [pitfall trap]

Malaise and pitfall traps were baited with various dungs and formed by far the major part of sampling methods. The main objective for most of these expeditions was to collect dung beetles (Coleoptera: Scarabaeidae) but other insect orders were retained. The Diptera was by far the greatest component of this by-catch and this brief account deals with this alone. The chosen sites, the bait utilised, and the collecting methods inevitably skewed the representation of families and diversity of species. For example, hardly any Agromyzidae and only a few Chloropidae (both large families) were collected, whereas Drosophilidae, Micropezidae, Phoridae, Sphaeroceridae and Sepsidae, unsurprisingly, were taken in large numbers. Nevertheless, the diversity of material is of significant value.

To give some context for this, a few brief details are provided below about each of the collecting localities.

Venezuela and Costa Rica. These two collecting trips were of limited scope. The former was undertaken by Dr George McGavin who collected sparingly and across limited insect Orders; the latter was an Oxford University student expedition. Neither of these trips generated large quantities of material. Sampling had a strong propensity towards easier collecting methods and larger sized insects within the Coleoptera, Hemiptera and Hymenoptera. The Diptera collected here was largely incidental but at least has been kept in reasonable condition through preservation in a freezer and alcohol since its arrival in Oxford.

Honduras. This material represents the greatest potential pool of unsorted ‘mystery’ material. Specimens from 2007 were made available for study through a volunteer led sorting project that aimed to upskill general museum volunteers in behind-the-scenes entomological work, teaching handling and basic identification skills. However, this project has only covered one tenth of the available material. The museum holds samples ranging across a 10-year sampling period (2007-2017) at the Cusuco Research site. Projects undertaken there are overseen by Operation Wallacea and have been both numerous and diverse in their collecting methods. There are varying quantities of Diptera, with later years influenced by the heavy bias towards dung-baited pitfall trapping. As many

of the on-site workers were volunteers, much of the smaller fauna was overlooked. Even so the mass of samples represents a deep reservoir of untapped material for researchers interested in Central America.

Bolivia represents by far the largest portion of South American Diptera to have been received into the collections at OUMNH in the last two decades. The main Refugio Los Volcanes research site was established by A.C. Hamel and S. Herzog, and much of the early work undertaken to survey the site for insects was done by A.C. Hamel. Numerous trapping techniques were employed for this faunal inventory work including, but not limited to, Malaise, flight interception, light (UV and mercury vapour), pitfall and aerial traps baited with a wide variety of materials including dung from cattle, pigs, chickens and humans, fish, various fruits and red meats, and mushroom. Later projects were more targeted, looking to collect specific groups within the Coleoptera (mainly coprophagic Scarabaeidae) but a variety of trapping methods continued to be employed and collecting was not limited by order. Work expanded from the Refugio Los Volcanes site and took in much of the south-eastern region of Bolivia, covering numerous habitat types. Some of this material remains unsorted, but the majority of material has now been processed to the point where it is split to order and accessible for research.

The collections also have some material from **Chile, Argentina and Brazil**.

During a week's visit in April 2018 to the Oxford Natural History Museum (OUMNH), MJE sorted out to family the dry mounted specimens resulting from the above expeditions. These were housed in eight full drawers. A great majority of specimens was mounted by non-specialist volunteers. At that time, it was apparent that many groups were poorly represented, contrary to expectations. Given that an even larger sample still remained unsorted in alcohol, it was highly desirable to have a dipterist examine this in order to select other species/specimens to supplement the main collection.

During tenure of a one month visiting fellowship in February – March 2019, MJE examined about 10-12,000 specimens, from which, several hundred were dry mounted to supplement the collections. For most families, but especially in the acalyptrates, most specimens were sorted out to genus, many to morpho-species and a handful to species. For MJE's personal benefit and interest, the following acalyptrate families received special attention: Chyromyidae, Clusiidae, Lauxaniidae, Micropezidae, Richardiidae and Ulidiidae. The families Chloropidae and Sphaeroceridae were specifically excluded because other workers had a specialist interest in these, and it was considered not economical of time to work on these. Among the lower Brachycera only the Stratiomyidae were numerous, other families being represented by very small numbers of species/specimens. Nevertheless, most of these were sorted out to family. Most calyptrates had already been partially sorted out and few specimens

UFO: Unidentified fly [belonging to the family] Odiniidae.

Table of listed material. A '+' sign indicates only a small number of additional genera/species.

Family	Number of genera	Number of species	Number of specimens	Country of origin
1. AGROMYZIDAE	5+	10+	60+	Argentina, Bolivia
2. ASTEIIDAE	2	2	2	Bolivia
3. CANACIDAE	1	2	12	Argentina
4. CARNIDAE	1	1	1	Venezuela
5. CHAMAEMYIIDAE	3	3	4	Argentina, Venezuela
6. CHYROMYIDAE	1	1	7	Venezuela
7. CONOPIDAE	4	24	81	Argentina, Belize, Bolivia, Brazil, British Virgin Islands, Chile, Costa Rica, Mexico, Tobago
8. CHLOROPIDAE	-	-	-	-
9. CLUSIIDAE	5	30+	130	Argentina, Bolivia
10. CURTONOTIDAE	1	4	6	Bolivia, Guyana, Mexico
11. DROSOPHILIDAE	10+	50+	250+	Argentina, Bolivia
12. EPHYDRIDAE	14	29	220	Argentina, Bolivia, British Virgin Islands, Costa Rica
13. HELEOMYZIDAE	1	3	18	Argentina, Venezuela
14. INBIOMYIIDAE	1	1	1	Bolivia
15. LAUXANIIDAE	14+	47+	234	Argentina, Belize, Bolivia, British Virgin Islands, Costa Rica, Venezuela
16. LONCHAEIDAE	2+	6	12	Bolivia, British Virgin Islands, Venezuela
17. MICROPEZIDAE	10	44	510	Bolivia, Brazil, British Virgin Islands, Costa Rica, Guyana, Honduras, Jamaica, Panama
18. MILICHIIDAE	6+	14	25+	Argentina, Bolivia, Venezuela
19. NERIIDAE	2	8	18	Bolivia, British Virgin Islands, Costa Rica
20. ODINIIDAE	3	3	3	Bolivia, Venezuela
21. PERISCELIDIDAE	1	1	1	Bolivia
22. PLATYSTOMATIDAE	2	5	13	Bolivia
23. PSEUDOPOMYZIDAE	2	3	9	Bolivia
24. PSILIDAE	1	1	2	Bolivia
25. PYRGOTIDAE	2	2	2	Bolivia
26. RICHARDIIDAE	12	26	104	Bolivia, Costa Rica, Honduras
27. ROPALOMERIDAE	3	3	5	Bolivia
28. SCIOMYZIDAE	3	3	7	Argentina
29. SEPSIDAE	?	?	370+	Bolivia
30. SOMATIIDAE	1	1	1	"Amazon"
31. SPHAEROCERIDAE	?	?	720+	Argentina, Bolivia
32. SYRINGOGASTRIDAE	1	2	3	Costa Rica,
33. TANYPEZIDAE	1	1	2	Bolivia
34. TEPHRITIDAE	5	7	10	Bolivia
35. ULIDIIDAE	14	46	120	Bolivia, Brazil, British Virgin Islands, Costa Rica, Mexico

were extracted from alcohol because of limited time. What was selected was based on those with very low numbers of representation (the common and numerous species had examples already dry mounted). This exercise yielded 11 acalyptrate families new to the collections and numerous species new to the collections in most families, but particularly in the Clusiidae, Lauxaniidae, Micropezidae and Ulidiidae.

All the examined and sorted material has been listed in an excel sheet with the number of specimens for each entry and their country of origin. In order to complete the picture, material from the Neotropics already incorporated in the collections (in accessions or in the main collection) and belonging to the families sorted out has been added to the list. The complete list is available to interested persons from ZMS, but a summary of the acalyptrates is provided in the Table on the previous page. It must be noted that more specimens remain in alcohol, especially where large numbers of particular genera or species were taken.

It is hoped this will encourage persons studying any group to contact ZMS to arrange a loan. It may also facilitate a loan to anyone willing to identify some of the material.

The fellowship scheme at the OUMNH continues to run on an annual basis with applications opening in around April every year. Information about the scheme can be found on the museum's website or by contacting the relevant collections manager. Approximately six fellowships are awarded each year and projects are welcomed on any area of collections-based research.

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Observation of the water snipe fly *Atherix lantha* Webb (Anthericidae) in Québec, Canada

Robert Loiselle[1] & Marjolaine Giroux[2]

[1] retired from Université du Québec à Chicoutimi (UQAC), Canada; robert_loiselle@uqac.ca;

[2] Division collections entomologiques et recherche, Insectarium de Montréal / Espace pour la vie, Québec, Canada; marjolaine.giroux@ville,montreal.qc.ca

On September 5, 2016, a curious picture was sent to the first author who was working for a pest control firm in Chicoutimi district of *Ville de Saguenay*. A young woman wanted to know the nature of a cluster of insects (wasps or flies?) glued together in a compact mass on the ceiling of a culvert (Fig. 1). Some weeks before, she and family members were afraid to approach the site (48° 44' N, 70° 84' W) with these insects flying around. Perhaps they could bite people?

Figure 1. A (left). Cluster of dead flies glued to the ceiling of the culvert; B (right). Bulk of flies showing a piece of wood on which the first females began to aggregate. Photos by Jacques Desbiens.

Figure 2. A (left). The brook downstream from the culvert; B (right). The culvert under the road 172. Photos by Jacques Desbiens.

We were intrigued at the first sight because all these insect bodies were barely recognizable, and some fungi did not help. However, according to the wing venation, it was clear that these insects were flies. On September 16, we decided to reach the brook (Fig. 2A) and then the culvert (Fig. 2B) where the bulk of flies was glued to the ceiling (Fig. 1A), some centimeters from the south opening. We noticed a small piece of wood on which the first flies grasped (Fig. 1B). The mass of flies was almost 6 cm wide and was the only one we could find.

The family of these flies, Athericidae, was identified by looking at the pattern of the veins (Fig. 3). Thereafter, the literature review allowed us to discover the special biology of the members of the genus *Atherix* (Madsen 2010, 2012, Haupt & Haupt 2000, Webb 1977) and to understand the bizarre behaviours of *Atherix lantha* Webb (Marshall 2012, Hunter & Maier 1994, Lauzon & Harper 1993).

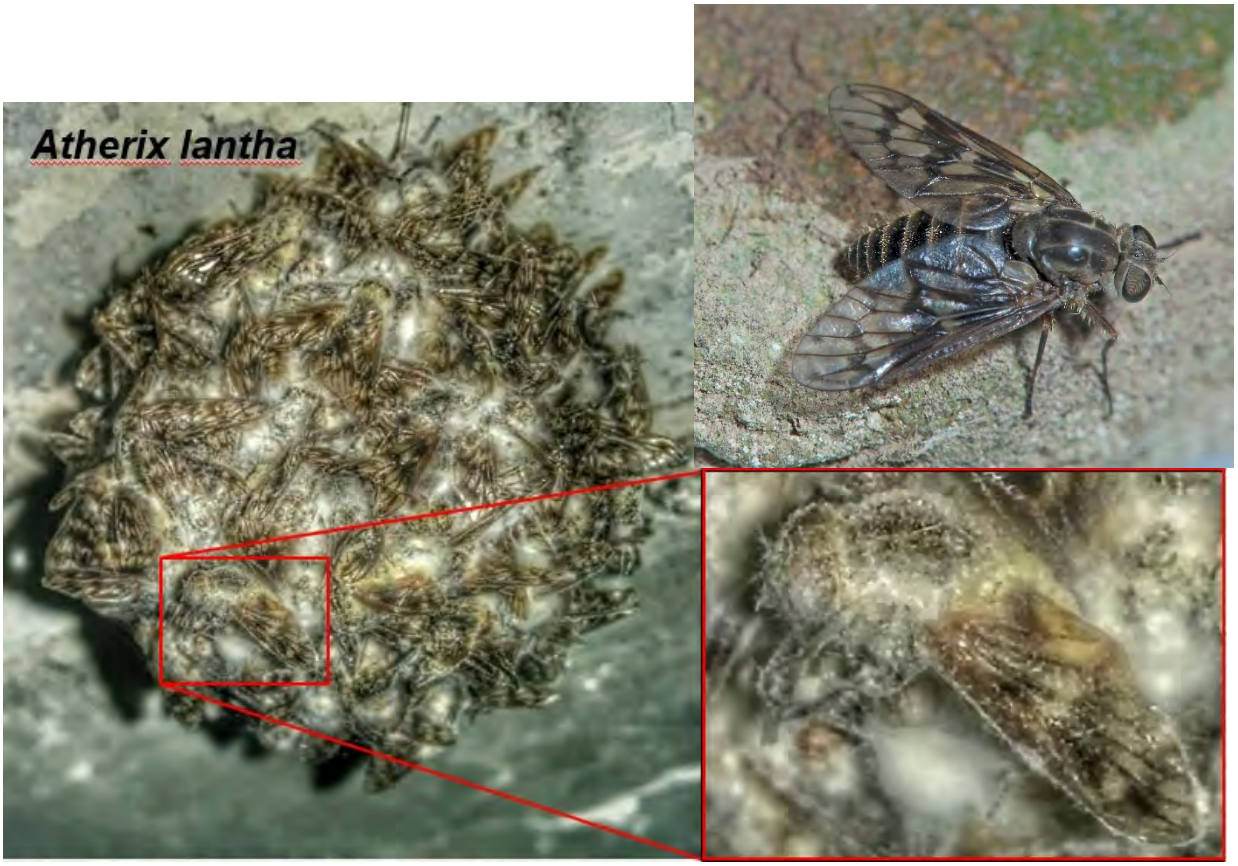

Figure 3. *Atherix lantha*. A (left). Cluster. B (lower right). Close-up of one fly, showing the left wing. C (upper right). Close-up of a living fly, showing the wings. Photos by Catherine Riverin (A, B) and Steve Marshall (C).

Females of this genus usually aggregate in large clusters at the tip of a branch situated over the water (Madsen 2010) or under bridges or other structures above the surface of streams (Madsen 2012, Marshall 2012). Females hang on the cadavers of previously installed females, lay their eggs and die. The larvae will soon go out the egg envelope and fall in the water of the brook. In Québec, Lauzon & Harper (1993) have shown that *A. lantha* has a univoltine life cycle with five larval instars, overwintering in the fifth and final instar.

How many of these flies were glued together?
On the upside-down bulk of flies, we first recognized some egg masses (Fig. 4). Afterward, to determine the total number of individuals, the bulk was put in 70 % alcohol. We then used an EZ4W Leica stereomicroscope with integrated digitizer and an ASUS laptop. It was very difficult to separate complete bodies; every cadaver was almost glued with its neighbors. Most of the time, it was impossible to detach them without breaking the body parts (Fig. 5). So we had to count the thorax present in the "soup". Doing this, we found some larvae cadavers here and there (Fig. 6).

We finally counted about 620 cadavers (± 10) in an approximate 110 cm^3 volume, a number that we did not expected at all! The biggest cluster Madsen (2012) had found had 290 cm^3 with *Atherix ibis* 1320 cadavers.

Figure 4. Bulk of flies (almost 6 cm wide) upside down, with close-up of an egg mass.

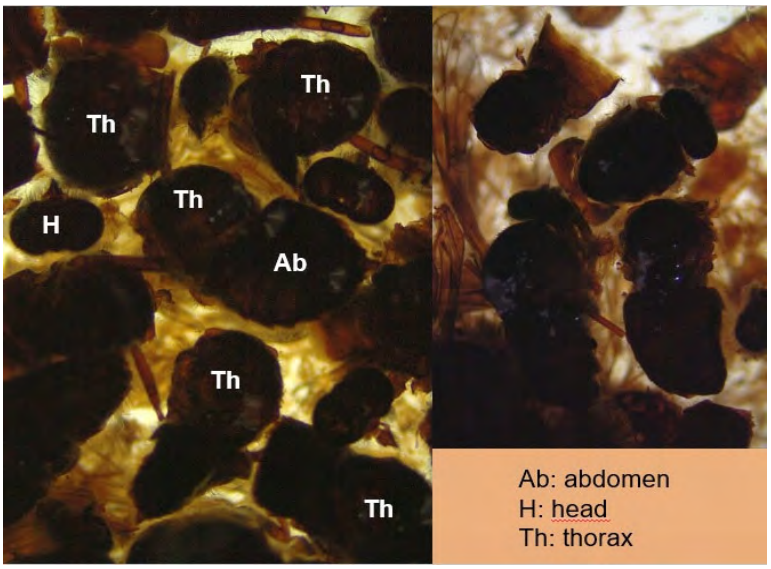

Figure 5. Counting of the number of flies in the "soup".

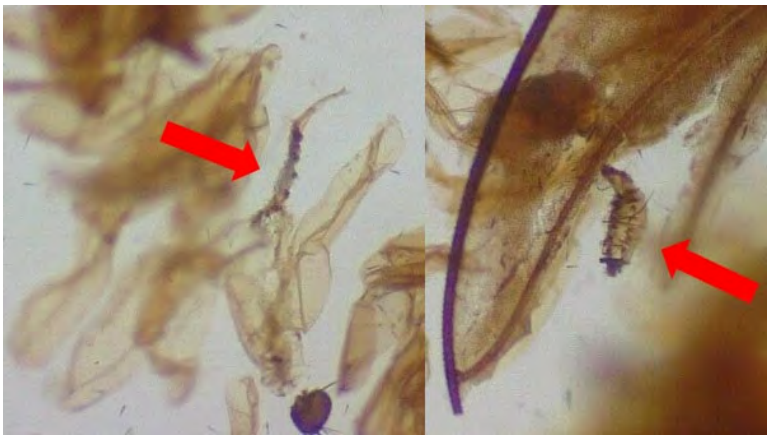

Figure 6. Dead larvae and egg envelopes.

Some interesting facts about of *Atherix* behaviours

With 22 species, the genus *Atherix* is mostly Holartic and the major north temperate group of water snipe fly (Marshall 2012). It appears that adults imbibe honeydew or water (Webb 1981), while aquatic larvae feed on other stream invertebrates by sucking their body fluids (Hunter & Maier 1994, Lauzon & Harper 1993). There are three Nearctic species (Lauzon & Harper 1993, Webb 1981) including *A. lantha* whose feeding mode and types of prey consumed, resembles its Palearctic relatives, notably *A. ibis*.

Atherix ibis, is a European species for which Madsen (2012) described the behaviour:

1. The first attraction to a good spot for the females seems to be visual; afterward, pheromones are used. A fresh bulk of flies would have a characteristic odour.
2. The gathering of females has probably a negative impact on egg predation. And perhaps their size creates confusion with wasps? It works with people…
3. Some parts of the first texts written about *Atherix* were wrong: the first larvae do not eat the dead body of the females. They rapidly (48 hours or less) fall in the water.
4. Finally, the larvae would drop during the night to avoid predation.

For the two authors and many of their friends, this very interesting behavior of *Atherix lantha* was completely new. Since Septembre 2016, when we cross a brook, we try to have a look under the bridge or the culvert, just to check if there would be a pack of flies. We didn't see any new ones… yet.

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Observations on how some very interesting flies benefit from ant behaviour to rob their resources

Ana Rita Gonçalves

Computational Biology and Population Genomics Group, cE3c, Faculdade de Ciências, Universidade de Lisboa, Campo Grande, 1749-016 Lisboa, Portugal; &
Operation Wallacea, Wallace House, Old Bolingbroke, Lincolnshire, PE23 4EX, United Kingdom; anagoncalvesm@gmail.com

During my stay in Calakmul Biosphere Reserve (Yucatán Peninsula, Mexico) as a Lepidoptera surveyor for Operation Wallacea, I came across interesting fly behaviours which do not seem to be very well known yet. I took the opportunity to take notes and, when possible, videos. Thus, I had the chance to witness the association of Tachinidae and Conopidae flies with ant army raids, as well as Chloropidae stealing the extrafloral nectar from an ant-plant mutualistic relationship. Despite the efforts to try to obtain a collecting permit, it was not possible to obtain one within the available timeframe I had. Consequently, I could not collect any of the insects here mentioned to identify them to species level.

Calakmul Biosphere Reserve is characterized as a semi-deciduous tropical forest, which is dry in the north but becomes more humid towards the south. The association of the flies with the ant army raids was observed on 18 July 2019, at Dos Naciones (approximate coordinates: 17.967, -89.357), in a deep valley, while the kleptoparasitic Chloropidae were observed on 01 August 2019 near a temporary pond by the Museo de la Naturaleza y Arqueología de Calakmul (18.365374, -89.889834).

Rettenmeyer (1961) documented for the first time the association of *Calodexia* van der Wulp, 1891 and *Androeuryops* Beneway, 1961 (Tachinidae) and *Stylogaster* Macquart (Conopidae) with army ant raids (Formicidae: Ecitoninae), noticing their presence at the front of swarm raids where they chased fleeing insects (acting as parasites on crickets and cockroaches). Thousands of these flies can be found accompanying the raids. Other fly families like Calliphoridae, Phoridae and Sarcophagidae have also been documented (Rettenmeyer et al. 2011). *Calodexia* are reported to be parasites of both cockroaches and crickets, and they seem to ignore all other arthropods. On the other hand, *Stylogaster* has never been reared, but by observing their behaviour it is hypothesised that they are also parasites on cockroaches and possibly on other arthropods (Rettenmeyer et al. 2011).

I was made aware of the presence of the army ants by the sound of the dry leaf litter crackling under the fleeing insects and their pursuers. The army ant species I observed probably belongs to the genus *Labidus*. I have seen a very large number (hundreds) of *Calodexia* flies, mostly females, perched on the vegetation, the vast majority of which were right at the front of the swarm raid, where they swiftly changed position likely to avoid contact with the ants. I have noticed several mating attempts among *Calodexia* and two specimens were already mating. *Stylogaster*, both males and females, were also very common over the front of the raid, hovering and switching position in sudden movements. Females had their very elongated abdomen and oviscape curved forwards. I have not noticed any mating attempts in this case.

Whenever an arthropod would flee from the ants, it would be immediately followed and checked by both the tachinids and conopids but, as previously reported, only cockroaches and/ or crickets were further pursued, while others were quickly ignored. At one point, a cricket tried to flee and instantly half a dozen *Calodexia* females surrounded it and chased it under the leaf litter. I have seen many

similar attempts, for which success of oviposition I cannot confirm, also with cockroaches. Regarding *Stylogaster*, I have only seen the females darting at cockroaches but ignoring the crickets and other arthropods. The females would also chase the cockroaches under the leaf litter.

Recently, the behaviour of a chloropid species, *Notaulacella octicola* Sabrosky, 1994, was described for the first time robbing the extrafloral nectar, a resource used and protected by *Pseudomyrmex* Lund, 1831 ants, which have an obligate mutualistic relationship with the acacia *Vachellia* Wight & Arn (Barrantes et al. 2018). I have witnessed this behaviour and I was able to record a (shaky) video (youtube link: https://youtu.be/UIxl8N1hK5M).

I've seen what looked like many hundreds of chloropids trying to obtain the nectar from the extrafloral nectaries of a *Vachellia* tree, while the *Pseudomyrmex* ants tried to repel the flies by rapidly moving towards them. The flies always escaped very quickly (walking backwards of flying away) before the ants could touch them. As reported by Barrantes et al. (2018), the chloropids would often land right behind the ants. Afterwards, the flies would again walk towards the nectaries and sometimes a line of flies was formed, as each one waited for a chance.

When they landed far away from the nectaries, the chloropids moved very quickly to reach it but if finally approaching an ant, they would continue to move, but now carefully, often stopping as if to avoid being detected. Sometimes, the ants would rotate around a nectary, possibly trying to protect it from the chloropids which could come from any direction. I have also witnessed chloropids feeding on the nectar and not being detected by an approaching ant, which turned around before getting close enough.

Summarizing the information, the chloropids were very numerous, moved very fast and seemed to be careful to avoid detection, while the ants were displeased by their presence, trying to repel them. The ants did not seem to make a special effort to actually attack and sometimes failed to notice the robbers.

Acknowledgements
I'm very thankful to Operation Wallacea for supplying the logistics that allowed me to stay in Calakmul, as well as to a large number of people who contributed to a crowdfunding aimed at covering the travel costs.

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Seeking records of rare or declining northeastern syrphids

Bruce E. Young

NatureServe, 2550 South Clark Street, Suite 930,
Arlington, VA 22202; bruce_young@natureserve.org

In 2007, the National Research Council released a report on the status of pollinators in the United States (NRC 2007). In it, they were unable to say much about the status of native pollinators because of a lack of monitoring and status information. Since then, NatureServe in collaboration with the US Forest Service, the Xerces Society and others has systematically assessed the conservation status of many native North American butterfly, moth, and bee groups.

Rhingia nasica, a syrphid that occurs in the northeastern US. Photo by John Klymko.

With the exception of bumble bees, most bee groups reach their diversity peak in desert regions of the southwestern U.S. In the Northeast, by contrast, other insects take on important pollinator roles. One group that has been largely overlooked by conservationists is the Syrphidae (flower flies). Flower flies are particularly diverse in moist Northeastern habitats (Skevington et al. 2019). Both male and female adults pollinate many plant species, including crops such as cranberries, as they forage for nectar and pollen. Unlike bees, in which young are provisioned by their mothers or colonies, flower fly larvae forage for themselves and often serve as decomposers in their ecosystems. Thus, flower flies play important but very different ecological roles in native habitats during different phases of their life cycle.

In a pilot study, we found that some flower flies are less common now than they were historically. The same threats that imperil bees and other insects, including intensified agricultural practices, habitat loss and pesticides, could also be affecting flower fly populations. In addition, some syrphids are known to inhabit old-growth forest. With little old-growth forest remaining in the Northeast, these species may have declined. Yet no broad scale assessment has been completed, and therefore conservation agencies do not have a basis for directing any conservation efforts toward the group.

Request for Assistance

To broaden our knowledge of the many challenges faced by native insect pollinators and continue catalyzing conservation funding, NatureServe is initiating a conservation status review of all native Northeastern syrphids (recognizing that some do not visit flowers). We began by assembling a list of target species from New York and New England that are rare or thought to have declined (Table 1). We have or plan to visit natural history collections from Washington, DC, to Vermont to gather data

Table 1. Syrphid species targeted for conservation status assessment and for which submissions of records is solicited.

Subfamily	**Species**
Eristalinae	*Blera pictipes* (Bigot, 1883)
Eristalinae	*Blera umbratilis* (Williston, 1887)
Eristalinae	*Brachyopa daeckeri* Johnson, 1917
Eristalinae	*Brachyopa diversa* Johnson, 1917
Eristalinae	*Callicera erratica* (Walker, 1849)
Eristalinae	*Ceriana abbreviata* Loew, 1864
Eristalinae	*Ceriana willistoni* (Kahl, 1897)
Eristalinae	*Chalcosyrphus anomalus* (Shannon, 1925)
Eristalinae	*Chalcosyrphus aristatus* (Johnson, 1929)
Eristalinae	*Chalcosyrphus depressus* (Shannon, 1925)
Eristalinae	*Chalcosyrphus metallifera* (Bigot, 1883)
Eristalinae	*Chalcosyrphus sacajaweae* (Shannon, 1926)
Eristalinae	*Cheilosia leucoparea* Loew, 1863
Eristalinae	*Cheilosia pontiaca* Shannon, 1922
Eristalinae	*Cheilosia primoveris* (Shannon, 1915)
Eristalinae	*Cheilosia wisconsinensis* Fluke and Hull, 1947
Eristalinae	*Chrysosyrphus latus* (Loew, 1863)
Eristalinae	*Cynorhinella longinasus* Shannon, 1924
Eristalinae	*Helophilus latifrons* Loew, 1863
Eristalinae	*Lejops* (*Anasimyia*) *distinctus* (Williston, 1887)
Eristalinae	*Orthonevra anniae* (Sedman, 1966)
Eristalinae	*Parhelophilus divisus* (Loew, 1863)
Eristalinae	*Parhelophilus integer* (Loew, 1863)
Eristalinae	*Pelecocera pergandei* (Williston, 1884)
Eristalinae	*Sericomyia bifasciata* Williston, 1887
Eristalinae	*Sericomyia slossonae* Curran, 1934
Eristalinae	*Sericomyia transversa* (Osburn, 1926)
Eristalinae	*Temnostoma trifasciatum* Robertson, 1901
Eristalinae	*Teuchocnemis bacuntius* (Walker, 1849)
Eristalinae	*Tropidia calcarata* Williston, 1887
Eristalinae	*Volucella evecta* (*bombylans*) Walker, 1852
Eristalinae	*Xylota ejuncida* Say, 1824
Eristalinae	*Xylota ouelletti* (Curran, 1941)
Microdontinae	*Microdon laetus* Loew, 1864
Microdontinae	*Microdon megalogaster* Snow, 1892
Microdontinae	*Microdon ocellaris* Curran, 1924
Microdontinae	*Mixogaster breviventris* Kahl, 1897
Microdontinae	*Mixogaster johnsoni* Hull, 1941
Pipizinae	*Neocnemodon pisticoides* (Williston, 1887)
Pipizinae	*Neocnemodon squamulae* (Curran, 1921)
Pipizinae	*Trichopsomyia banksi* (Curran, 1921)
Pipizinae	*Trichopsomyia pubescens* (Loew, 1863)
Syrphinae	*Dasysyrphus amalopsis* (Osten Sacken, 1875)
Syrphinae	*Eupeodes confertus* (Fluke, 1952)
Syrphinae	*Parasyrphus semiinteruptus* (Fluke, 1935)
Syrphinae	*Platycheirus parmatus* Rondani, 1857
Syrphinae	*Platycheirus scamboides* Curran, 1927
Syrphinae	*Sphaerophoria longipilosa* Knutson, 1973

from specimen records of these species. To enhance the information base available to perform the assessments, we seek additional records of these species in private or academic collections. If you have or know of records of these species from anywhere in their ranges and are willing to share them,

please let us know. The minimal data we need is simply year of collection or observation, county, and state. Full dates and notes on floral associations or other natural history information are also helpful. Please also let us know if you think a declining or rare species from our study area is missing from the list. You can contact me at bruce_young@natureserve.org. Thank you.

Past Success

NatureServe's past work highlighting the conservation plight of insect pollinators has demonstrated that conservation status assessments can mobilize conservation action. For example, our work on bumble bees (Schweitzer et al. 2012) and hawk moths (Young et al. 2017) inspired a bumble bee atlas program conducted by the state of Maine and a $1M, five-year initiative in New York to census pollinators. Using NatureServe status data as partial justification, partners in state agencies are adding pollinating insects to their State Wildlife Action Plans and therefore paving the way for public funds to be invested in their conservation. Through the current effort, we hope to publicize conservation needs of any Northeastern syrphids that we find to be imperiled.

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Cautionary notes about the use of lactic acid for clearing Diptera genitalia

Stéphanie Boucher

Lyman Entomological Museum, Macdonald Campus, McGill University,
Ste-Anne-de-Bellevue, Quebec, Canada; stephanie.boucher@mcgill.ca

The use of potassium or sodium hydroxide (KOH; NaOH) in clearing fly genitalia has been associated with a few problems including the destruction of delicate membranous structures, immediate over-clearing and a process of clearing that persists over time despite the use of acetic acid for neutralization (Cumming 1992; Lonsdale 2017; Sinclair 2008; Skevington and Marshall 1998; Wheeler 1994; Yau and Marshall 2015).

To overcome these problems, lactic acid for macerating fly genitalia was proposed as an alternative to KOH/NaOH (Cumming 1992). Lactic acid had the advantage of removing soft tissue without over-clearing the sclerotized portions of the genitalia and offered better long-term stability as the reaction did not persist once the soft tissues were dissolved (Cumming 1992; Skevington & Marshall 1998). The disadvantages of lactic acid include the corrosive and dangerous fumes produced when heated and the possible expansion and deformation of soft tissues (Cumming 1992; Yau and Marshall 2015). The suggested procedure for macerating fly genitalia using lactic was to gently heat up the 85% lactic acid with the fly abdomen for approximately 10-15 min depending on the size of the specimen. The genitalia can then be observed and stored in glycerin (Cumming 1992).

Although many dipterists are still using KOH/NaOH as a clearing agent, some (e.g. Grootaert and Shamshev 2012; Henriques and Krolow 2013; Solecki and Wheeler 2015) have switched to subsequent use of lactic acid following the procedure described in Cumming (1992). Others are using lactic acid but with higher temperatures and/or longer exposure times (e.g. abdomen boiled in lactic acid for about 30 min (Skevington 1999); genitalia cleared by heating in lactic acid overnight (Skevington et al. 2019).

A faster way of preparing fly genitalia involves clearing the abdomen in 85% lactic acid heated in a microwave oven in 30 second intervals separated by cooling periods (Wheeler 2000). This procedure was later adopted with exposure times varying from 15–30 seconds, separated by cooling periods (Barrie and Wheeler 2016; Boucher 2002; Boucher 2003; Boucher 2004, Gilbert & Wheeler 2007; Gregoire Taillefer and Wheeler 2011, Brooks and Cumming 2018; Wheeler 2003), or 10 second intervals separated by cooling periods (Mlynarek and Wheeler 2018; Wheeler and Solecki 2013).

Unfortunately, I have noticed that the genitalia of many male Agromyzidae cleared in the microwave with lactic acid became paler over time, including some holotype specimens shown in Figures 1–8. These figures show the progression in clearing over only three years (from 2010 to 2013) (Figs. 1–2) and over nine years (Figs. 3–8). Unfortunately, the genitalia of these holotypes have now become almost invisible. Because of this transparency, the current state of the holotype of *Cerodontha angela* Boucher cannot be shown here as it is invisible in the microvial. This dramatic over-clearing has occurred in many Agromyzidae dissected by the author since 2000. Figs. 9–10, show the progression of clearing over nine years on the genitalia of a specimen of *Cerodontha colombiensis* Spencer. This over-clearing problem is also noticeable in some Chloropidae (J. Mlynarek and A. Solecki pers.com.). that have been cleared in the microwave. On the other hand, empidoids that have been cleared this way, are not or very little affected with possibly only slight fading over time (S.Brooks and J. Cumming pers. com.).

Figures 1–2. Male phallus of *Cerodontha angela* (Holotype) (Ecuador: El Angel, 1903) (NMNH). 1 (left). Dissection and picture 2010. (From Boucher and Wheeler, 2014). 2 (right). Picture 2013. No view possible in 2019, as specimen is transparent

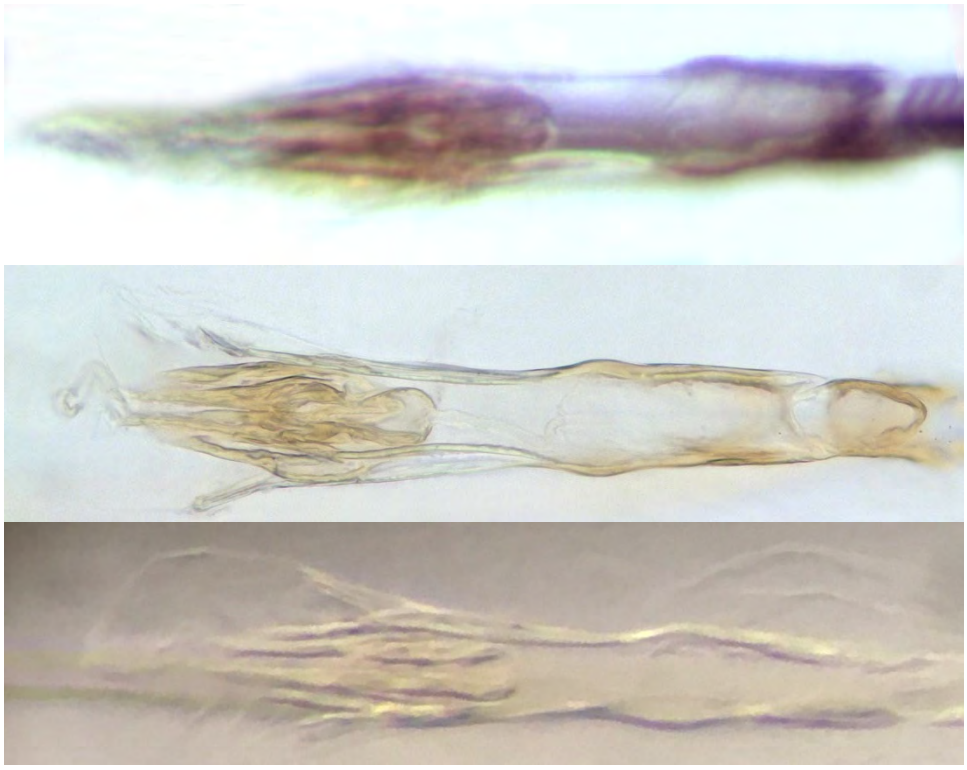
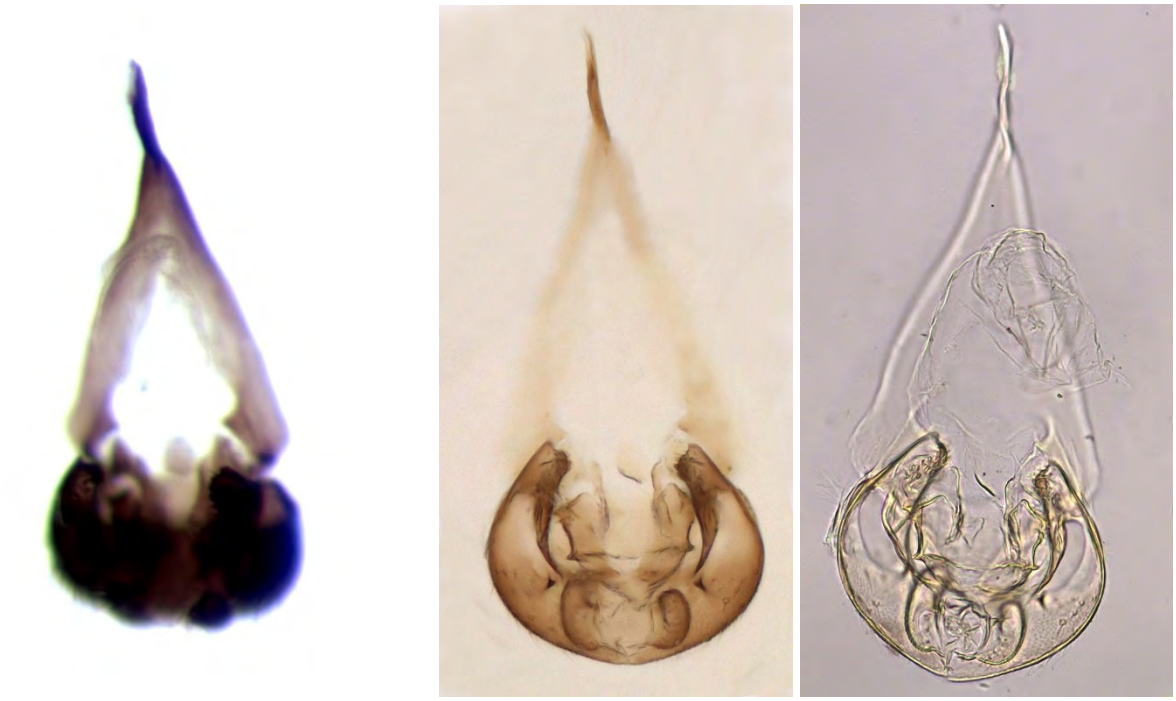
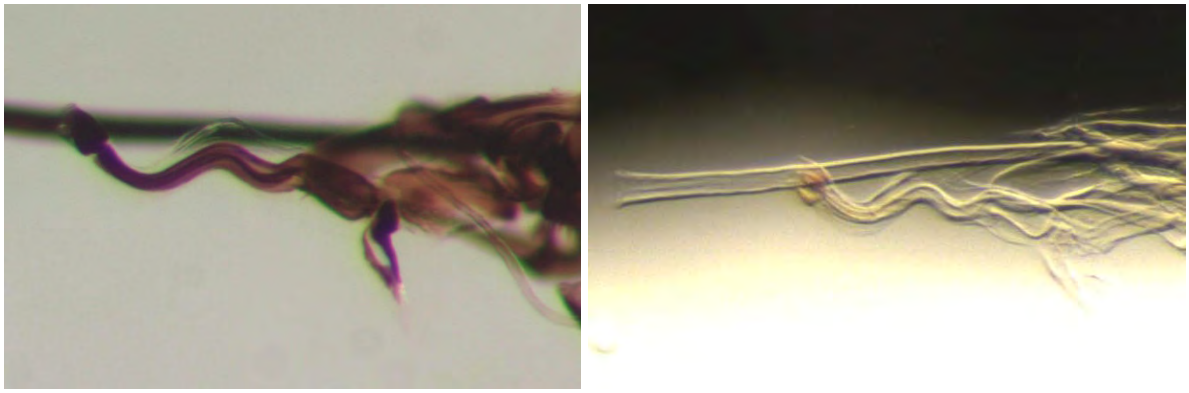


Figure 3–5. Male phallus of *Melanagromyza pontis* (Holotype) (Ecuador: La Rinconada, 1903) (NMNH). 3 (top). Dissection and picture 2010 (from Boucher and Wheeler, 2014). 4 (middle). Picture 2013. 5 (bottom). Picture 2019.

It is not yet known if over-clearing has also happened in specimens heated on a hot plate or in a water bath. Most Pipunculidae and Syrphidae that have been cleared with lactic acid years ago on a hot plate, are still in good condition, except for some very pale yellow flies that clear too much with lactic acid (J. Skevington pers. com.). Small, yellow and weakly sclerotized agromyzids (e.g. some *Liriomyza* or *Phytoliriomyza*) appear to be more rapidly and frequently affected by over-clearing, although some agromyzids with heavily sclerotized genitalia have been similarly affected as well.

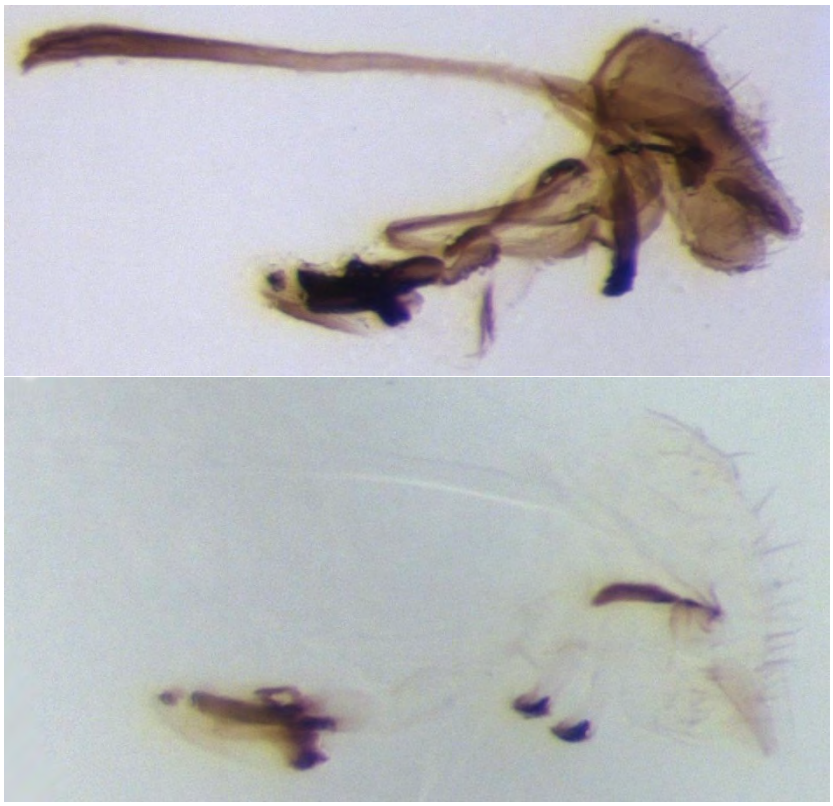


Figures 6–8. Genitalia (epandrium and hypandrium) of *Melanagromyza pontis* (Holotype) (Ecuador: La Rinconada, 1903) (NMNH). 6 (left). Dissection and picture 2010. 7 (middle). Picture 2013. 8 (right). Picture 2019.

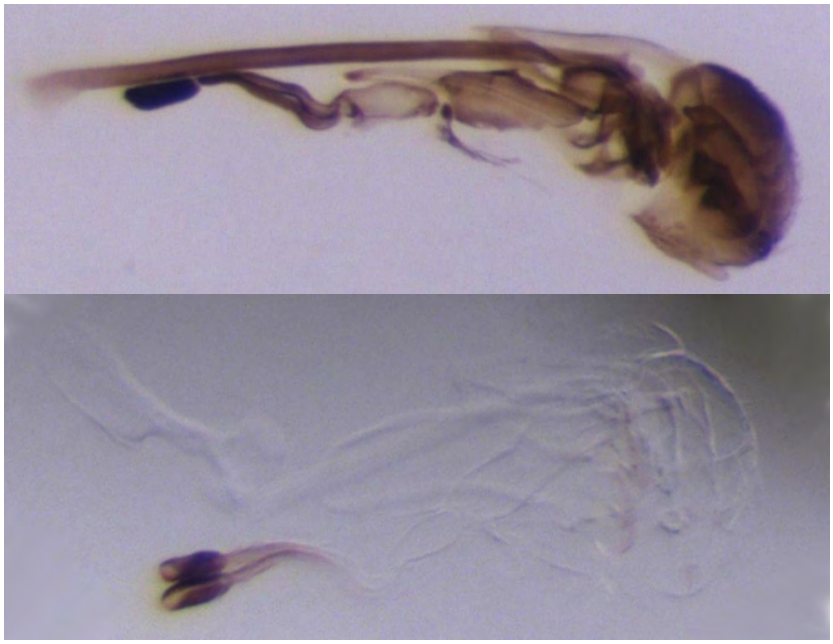


Figures 9–10. Male phallus of *Cerodontha colombiensis* (Peru: Camacani, 1955) (CNC). 9 (left). Picture taken after dissection in 2010. 10 (right). Picture year 2019.

Agromyzid specimens that have been affected by over-clearing belong to different insect collections (LEMQ - Ste-Anne-de-Bellevue, CNC - Ottawa, NMHM - Paris, NMNH – Washington, DC and DEBU - Guelph). Most were probably collected in ethanol. Some have probably been air-dried, others chemically dried using Hexamethyldisilazane (HMDS) (LEMQ specimens) or critical point dried (some CNC specimens). The age of the specimens varies from the early 1900's (e.g. *Cerodontha angela* and *Melanagromyza pontis* Boucher, Figs.1–4) to the early 2000's. Some LEMQ agromyzid specimens collected approximately the same year and belonging to the same species, and that have received the same chemical (ethanol, HMDS, lactic acid) and microwave treatments show very different stages of clearing today, with some in very good condition 19 years after the treatment (Figs. 11, 13) while others are almost completely transparent (Figs. 12, 14).



Figures 11–12. Male phalluses of *Pseudonapomyza europaea* Spencer. 11 (top). QC: Ste-Anne-de-Bellevue, 2000 (LEMQ), dissected in 2001, picture in 2019. 12 (bottom). QC: Pointe-des-Cascades, 2002 (LEMQ), dissected in 2002, picture in 2019.



Figures 13–14. Male phalluses of *Cerodontha dorsalis* (Loew). 13 (top). NS: nr Pictou, Caribou Prov. Park. 1999 (LEMQ) dissected in 2000, picture in 2019. 14 (bottom). MB: 20 km E. of Anola. Brokenhead R. 1999 (LEMQ) dissected in 2000, picture in 2019.

It is still unknown if cold lactic acid (e.g. residual lactic acid remaining on the genitalia) is responsible for the clearing process persisting once in storage, but it seems unlikely, as lactic acid is sometimes used as a long-term storage fluid for Lepidoptera genitalia (J-F Landry pers.com.) and has also been used for storage of Diptera genitalia by D.M. Wood (J. Cumming pers.com.).

This difference in clearing stages may be explained by a variation in temperature received by the specimens. One of the problems associated with microwave preparations is the difficulty in monitoring the temperature of lactic acid. Furthermore, the time of heating (normally suggested as 10-30 seconds intervals) should be modified based on the size and the pigmentation/sclerotization of the specimen. In addition, the freshness of the lactic acid should be considered. As mentioned by Cumming (1992), one advantage of using lactic acid is that it can be reused multiple times, up to about 10 times on average before changing it for a new preparation. But after multiple uses of the same preparation of lactic acid, water evaporates, and the preparation becomes thicker and takes longer to heat up. A freshly changed preparation of lactic acid can take as little as 10 seconds in the microwave to reach near boiling stage (although this may vary depending on the power of the microwave). A preparation of lactic acid that has been heated up multiple times could take more than twice as long to reach the same temperature. Fly genitalia should probably not be boiled in lactic acid.

To avoid boiling the genitalia, I have started heating the lactic acid in the microwave without the genitalia. Although there is still some testing to do, here are my recommendations for clearing fly genitalia using lactic acid in the microwave (obviously this microwave should only be used for this purpose!):

1) Heat up a small amount of lactic acid (I use a small ceramic pot with glass cover) in the microwave for 10 to 20 seconds depending on the freshness of the lactic acid (see above). Do not include abdomen/genitalia.
2) Transfer the abdomen/genitalia into the warm lactic acid. Leave for about 30 seconds (this step should be performed under the fume hood to avoid dangerous fumes from hot lactic acid (see above).
3) Transfer the abdomen/genitalia into a depression slide containing glycerin.
4) Inspect under the microscope to see if further maceration is necessary. Use fine probes to push remaining macerated tissue inside abdomen.
5) If further maceration is necessary, repeat steps 1-4.

For small, weakly sclerotized specimens probably no more than one or two rounds should be necessary to examine the genitalia effectively without causing over-clearing. I also recommend rinsing the genitalia in fresh glycerin or possibly in ethanol to remove any residual lactic acid, in case this could affect the specimens over the long-term. Dissection can then be stored in a microvial containing fresh glycerin.

For specimens that have been over-cleared it is sometimes helpful to look at the genitalia partly over a black background (Fig. 10). Over-cleared specimens can also be stained. Although I haven't personally stained specimens, the method described by Nartshuk and Anderson 2013 for Chloropidae (adapted from Wilkey 1962) might be appropriate, although there are possibly better options (J.-F. Landry pers. com.).

Lactic acid may remain a good option for clearing Diptera genitalia when done properly. But another method that I am starting to explore is the one described by Yau and Marshall (2015), using contact lens enzymatic solution. I have been using "Boston one step liquid enzymatic cleaner (5ml)" (that is already mixed into a solution), and obtained good results (Figs. 15, 16). The fly abdomen is put directly into a glass microvial with one or two drops of enzymatic solution and the microvial is placed into a sonicator with water. The sonicator vibrates and the water is heated gently, but it takes 1-2 hours to get to a nicely cleared genitalia preparation for a small specimen.

Unfortunately the methods used for clearing Diptera genitalia are not always included in systematic papers (including my agromyzid papers published after 2004 - all genitalia were cleared in lactic acid in a microwave for 2-3 times 20 seconds). It should be considered important to describe the method more clearly so that a better follow up on a specimens' condition could be made at a later date and different methods of clearing (KOH, NaOH, lactic acid, enzymatic contact lens solution, hot plate, water bath, microwave, etc.) compared.

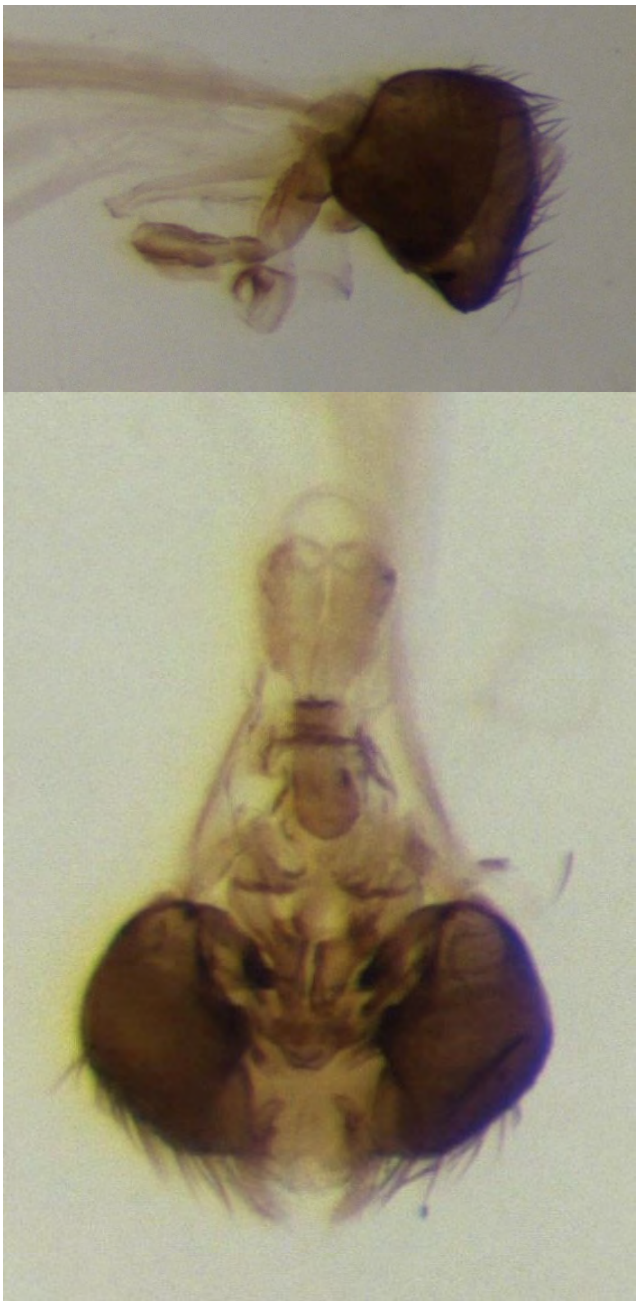


Figure 15–16. Male phallus of *Liriomyza marginalis* (Malloch) (Venezuela: Aragua 1998) (LEMQ) dissected in 2019 and macerated using contact lens enzymatic solution.

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I would like to thank Jeff Cumming, Scott Brooks, Anna Solecki, Julia Mlynarek, Jean-François Landry and Jeff Skevington, for answering my questions about their experience with lactic acid. I would also like to thank Jeff Cumming for his comments on this article.

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Untapped dipteran treasures in the Cornell insect collection

William L. Murphy

Research Collaborator, Smithsonian Institution, 7835 Tufton Street,
Fishers, IN 46038 USA; murph3000@gmail.com

This is to document and update a longtime sciomyzid project and to provide a roadmap to possible research activities for graduate students.

In the 1960s, live specimens of Sciomyzidae collected in South America by Clifford O. Berg and Jay Abercrombie were sent back to Cornell University to be reared in the laboratory by Abercrombie and Jan Zuska. The goal was to elucidate the life cycles of as many species as possible and to describe all immature stages. Zuska reared about 12 species of *Ditaeniella*, *Parectinocera* (known then as *Dichrochirosa*), *Pherbellia*, and *Shannonia* as well as several as-yet undescribed species of several genera and possibly members of one or two as-yet undescribed genera. Abercrombie concurrently reared some of the same species from the same field collections. Also involved in the Cornell rearings were Albertus Bratt, Victor Kaczynski, and Karl Valley.

Abercrombie, Berg, Bratt, Kaczynski, Zuska, and possibly others kept day-by-day records of individual laboratory rearings (Fig. 1). The custom in Dr. Berg's Cornell lab was for all the graduate students to have a specific number that identified the data being collected about a fly (or maybe a pair of flies); thus the concept of biological note (BN) numbers. For example, "6901" was the first rearing of 1969, with a prefix added to identify the student responsible for the rearing: “A6901” for Al Bratt, "B6901" for Berg, "J6901" or "JA6901" for Jay Abercrombie, “KV6901" for Karl Valley, and "V6901" for Kaczynski. According to Zuska, some BN numbers, such as Z6757A (Fig. 2), which includes an appended letter, probably denoted subcultures started from progeny of Z6757—eggs, larvae, and/or puparia transferred from female sciomyzid Z6757 to a separate plastic box and then maintained for detailed observation such as duration of immature stages or food preferences.

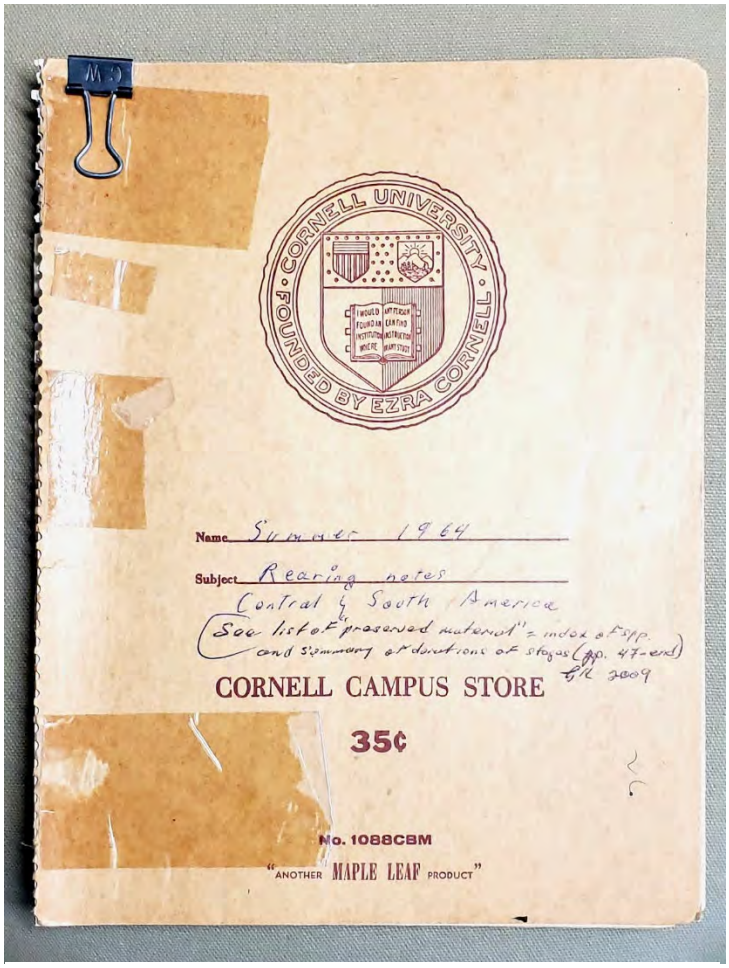

Figure 1. Al Bratt’s rearing notebook.

Figure 2. Typical box of pinned reared material, showing many with the BN number starting with Z.

Currently the Cornell collection holds about 1000 adults (parents and progeny, some pinned with puparia and/or shell of the host snail) from these rearings, stored at the end of each series of the respective genera, where the identity is known. Most specimens bear a single label with only a biological note number. The species name associated with each BN number can be found in one of several notebooks/journals that I have uploaded to the Internet (see below); the originals will be deposited at Cornell. Once the species name has been obtained, then the next step is to locate data about the field trip on which the specimen or parent specimen was collected. In most cases, that information can be found in one of the notebooks maintained by the person who reared the organism. I have uploaded to the Internet as much of that information as I have been able to locate.

Additionally, the Cornell collection contains many specimens of immature stages from these rearings, preserved in small vials submerged in ethanol in more than 65 Mason jars. The jars bear labels as to their content, and the small vials are labeled with BN numbers and possibly with species data, in some cases. Eventually all of these specimens should be identified and labeled with full date/locality labels and rearing information.

A few years ago, the late Lloyd Knutson (Gaeta, Italy) obtained from Jan Zuska (Czech Republic) several thousand pinned adults from the Cornell rearings, intending to label every specimen. Zuska still had his 500+ biological note cards from the rearings. The cards were written in English with some Czech. Knutson tried without success to obtain funding to get Zuska's biological notes translated and compiled so the data could be used to produce labels to be pinned with each specimen. With financial aid from Knutson, Zuska's notes were translated, transcribed, and burned onto a CD. Zuska merged the biological notes/note numbers with the field collection data to produce documents from which labels could have been printed, but Knutson died before beginning that task. I have uploaded Zuska's list of biological notes with field-collection data to the Internet.

The Zuska specimens at Knutson's home were shipped last September to the Smithsonian Institution in Washington, DC USA. Last month I retrieved those specimens and all associated journals and paperwork. Using Zuska's documents, I prepared labels for all specimens that had BN labels, but I left for a future student the task of matching the labels with the specimens. In early August I will hand-carry all of the specimens, journals, and other material to Cornell, where Karl Valley (Harrisburg, PA) and I will re-curate it into the collection.

Of note are long series of pinned specimens, one labeled *Dichrochirosa iacobi* and the other labeled *Dichrochirosa kusheli*. These species have never been described. Zuska described the names as provisional, originally coined by Berg. Because they were never used in any manuscript, they are not manuscript names, strictly speaking. Zuska suggests that they be labeled *D. iacobi* nom. prov. and *D. kusheli* nom. prov. These species, if verified to be valid new species, currently would be placed in the Sciomyzini genus *Parectinocera*, which contains only three species, all Neotropical.

The specimens from the Cornell rearings represent a fantastic opportunity for students of Diptera to expand our knowledge of the family with a minimum of effort. All of the date/locality data, multiple specimens of each life stage, and all rearing data await the attention of future researcher. I hope this background will assist them in that task.

Files to download (temporarily on my Trinidad Birding web space)

1) Jay Abercrombie's Cornell Rearing Data: http://www.trinidadbirding.com/wp-content/uploads/Cornell/AbercrombieCornellRearingData.pdf
2) Jay Abercrombie / Jan Zuska List of Biological Notes on Zuska CD: http://www.trinidadbirding.com/wp-content/uploads/Cornell/AbercrombieZuskaListOfBiologicalNotesOnZuskaCD.pdf
3) Albertus Bratt/ Vic Kaczynski Rearing Notes: http://www.trinidadbirding.com/wp-content/uploads/Cornell/BrattKaczynskiSciomyzidaeRearingNotes.pdf
4) Cliff Berg's Field Journal, 1964, 1967: http://www.trinidadbirding.com/wp-content/uploads/Cornell/BergFieldJournal1964_1967.pdf
5) Lloyd Knutson's Biological Note Numbers and Field Trip Records: http://www.trinidadbirding.com/wp-content/uploads/Cornell/KnutsonBiologicalNoteNumbersAndFieldTripRecords.pdf
6) Jan Zuska's List of Biological Notes (integrated Biological Note Numbers and Date/Locality: http://www.trinidadbirding.com/wp-content/uploads/Cornell/ZuskaListOfBiologicalNotes.pdf

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The blowfly *Chrysomia megacephala* (Fabricius, 1794) (Calliphoridae) aggregation on drumstick, *Moringa oleifera* pods in India

R. Mini Shaily[1], S. Chandra kumar[2] and M. Jayashankar[3]

[1] Maharani's Science College for Women, Bengaluru-12, India

[2]St Aloysius PU College, Bengaluru-05, India

[3] Assistant Professor, Department of Zoology, St. Joseph's College (Autonomous), Bengaluru-27, India; jay81zoology@gmail.com

The blowfly *Chrysomia megacephala* (Fabricius, 1794) is a calliphorid of forensic importance (Rozane & Martin, 2018), pollinator and crop pest. The adults are attracted to and breed in carrion, garbage and feces, hence the name Oriental latrine fly. The species is also attracted to nectar and is reported as a sapromyophilous agent in *Sterculia foetida*, *Euphorbia antiquorum* and *Rafflesia* (Bai 2008). There are multiple records of the species as pollinator of different crops in India (Mitra 2010). *C. megacephala* was found to be effective pollinator in mango ecosystem (Rajan & Reddy 2019) and the authors suggest augmenting the populations of *C. megacephala* in mango orchards during flowering period to reap better yields considering the ease and economy compared to beekeeping. On the contrary, it is reported as a pest of Cashew in India (Technical bulletin 2015).

The present report is on the occurrence of *C. megacephala* on drumstick pods in India during the month of June 2019. The unusual aggregation of the synanthropic flies was observed in one of the trees planted in the locality in Doddanna Nagar of Bengaluru city (12.9716° N, 77.5946° E) (Fig.1).

Fig. 1. Aggregation of *Chrysomyia megacephala* on drumstick pods, necrosis of pods can be observed (arrow), *Bactrocera* sp. can also be spotted (encircled)

Pod length, percent damage based on visual estimate and number of adult flies were recorded on seventeen pods on the tree (Fig.2). The average pod length was 28.47 cm, percent damage 38.53% and 14.06 flies/pod. Observations on seven more trees in the same lane revealed flies on one more tree but with minimal incidence. Drumstick tree ('miracle tree') is known for its medicinal and nutritional attributes, with leaves, pods and flowers consumed in India, its native country. Although a horticultural crop cultivated and harvested on a large scale in the southern states of the country, it is a common practice to plant drumstick, neem and curry leaf plants in backyard or in front of residences in urban areas. The flies aggregating in clumps were found sucking sap causing necrosis of the tender immature pods. Adult blowflies are known to feed on nectar, honey dew and other sweet liquids or liquid products of organic decomposition (Mitra 2010). The incidence of flies continued for more than a week affecting the pod development until the tree was pruned. Incidentally, during the observation, a *Bactrocera* species (not identified) was spotted near the pod with *C. megacephala* aggregation. The other dipteran causing drying and splitting of fruits from tip and oozing of gummy exudate from pods is the Drumstick Pod-fly, *Gitona distigma* Meigen (Drosophilidae: Diptera) with crop losses as high as 75 percent (Sivagami & David 1968). Considering lack of literature in this regard, the present observations are first of its kind of *C. megacephala* on drumstick pods. Is this a case of exploitative feeding on an ephemeral resource like the pod?. From a chemical ecology perspective, reasons for this behaviour and mechanism of sap sucking need to be probed.

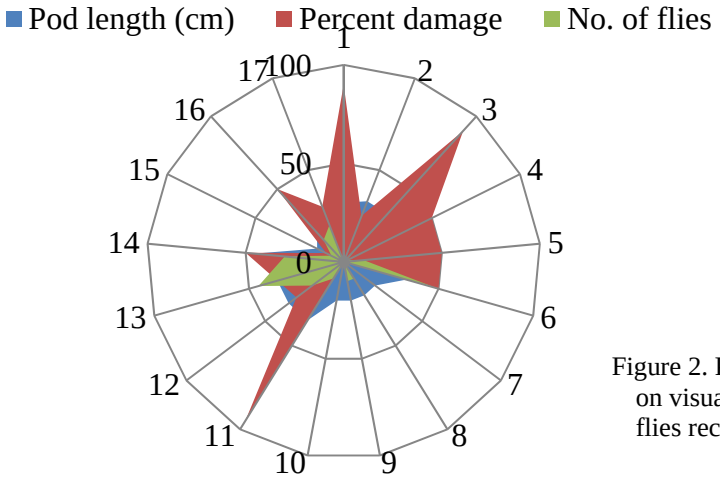

Figure 2. Pod length, percent damage based on visual estimate and number of adult flies recorded

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Ménage à trois **in a laboratory culture of crane flies**

Arthur Forer

Prof. Emeritus, Biology Department, York University,
Toronto, Ontario M3J 1P3, Canada; aforer@yorku.ca

FIGURE 1

I have reared *Nephrotoma suturalis* (Loew) in the laboratory since 1961, in a stock that started from a small number of larvae. (As described in a previous issue of *Fly Times*. I still am happy to help people start a laboratory culture of them before the stock disappears when I really retire not too many years from now and my lab gets decommissioned.) We put pupae in a moist dish in large cages [essentially a cube of transparent plexiglas, approximately 50 x 50 x 50 cm, containing arm holes covered with cheesecloth (muslin) to allow arms to enter the cage]. The adults emerge in the cage. After mating they lay eggs in a dish of moist paper. In the cages I often see pairs mating. I often see males try to mate with females, sometimes successfully, sometimes not. I often see males try to mate with males, always unsuccessfully. I often see males try to mate with the female of a copulating pair though I have never seen them succeed: invariably, after several attempts (in a short time) the single male always stops trying and separates from the mating pair. I never have seen females attempt to mate with males. And I never have seen females attempt to mate with females [drawings in the literature to the contrary notwithstanding – F. H. Stich, *Canadian Journal of Zoology* **41** (1963) 99-109]. Figure 1, typical views of adult *Nephrotoma suturalis* resting on the wall of a cage and on the cheesecloth covering an unused arm hole, illustrates single individual flies as well as several copulating pairs.

Last year I worked in a lab not equipped with large cages for the adult crane flies to emerge into, so I had to improvise. Instead of a cage I used small empty blue-plastic boxes that originally held pipette tips (for use with Pipetman or Eppendorff pipettes). I didn't measure the box dimensions but estimate that they are maybe 14 x 9 x 10 cm. One day I saw in the pipette box that two males were mating with the same female. I was able to see well enough to be sure that two indeed were attached to the one female, and that they stayed attached for the several minutes I looked at them and as I was taking pictures with my cell phone (Figure 2). In our large cages I have never seen two mails mated with one female. I can only presume that in cramped quarters two males were alerted to a female and arrived at the same female at the same time. I did not see this again in the small boxes, I have not seen it in our cages, ever, and I have no idea how they did it, but I thought the observation might be of interest to others.

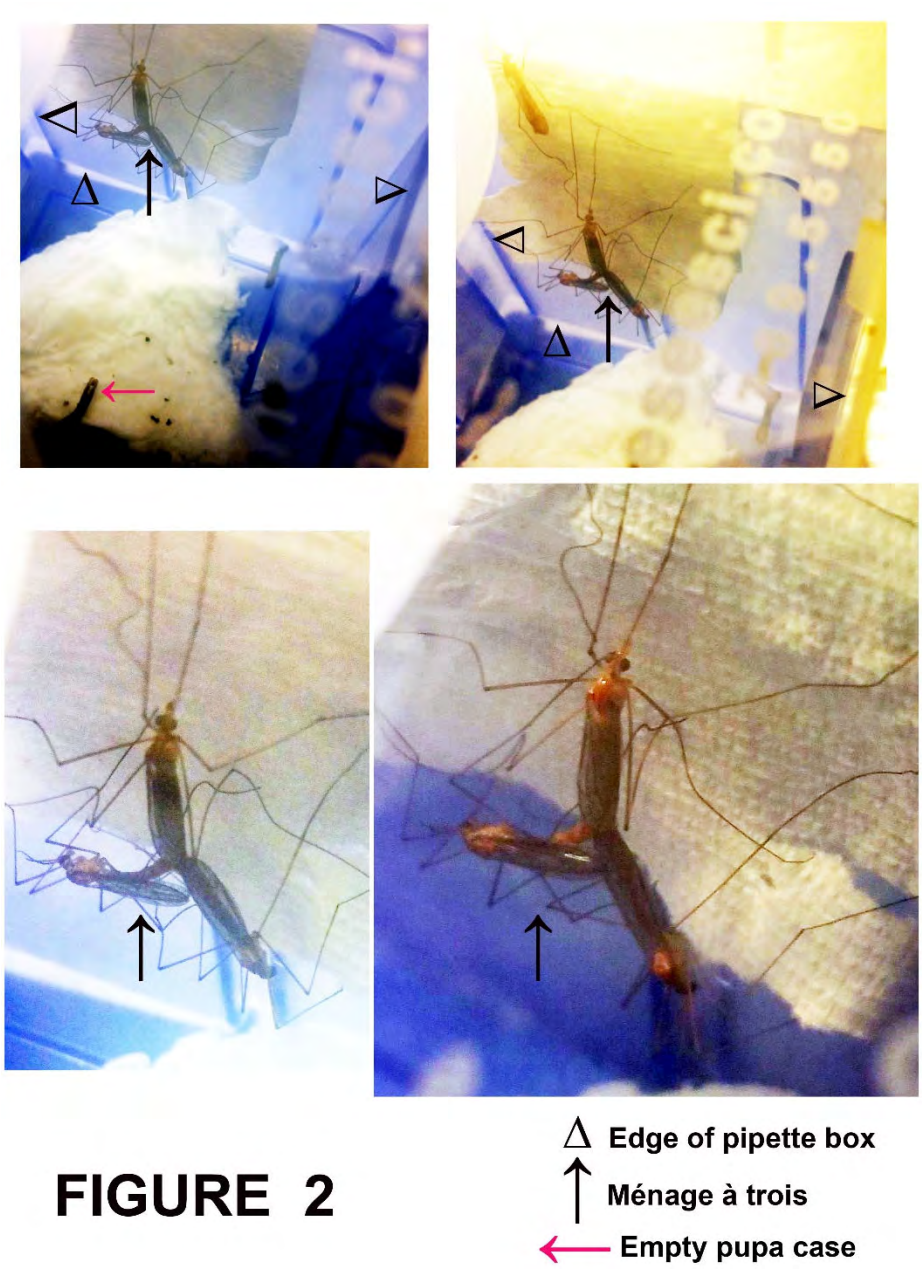

FIGURE 2

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A Florida Keys record for the African fig fly, *Zaprionus indianus* Gupta (Drosophilidae)

Lawrence J. Hribar & Michael Boehmler

Florida Keys Mosquito Control District, 503 107th Street, Marathon, Florida 33050 USA

On 29 May 2019 a notably patterned fly was collected in a carbon dioxide-baited light trap set for mosquito surveillance on Upper Matecumbe Key, Monroe County, Florida. The striking coloration of the fly ignited a desire to identify the specimen; it was determined to be the African fig fly, *Zaprionus indianus* Gupta (Fig. 1).

Figure 1. Point-mounted fig fly from Upper Matecumbe Key, Florida.

Previous records in Florida include 34 counties: Baker, Bay, Brevard, Broward, Collier, Dade, Desoto, Duval, Escambia, Flagler, Franklin, Gadsden, Hardee, Highlands, Hillsborough, Indian River, Jefferson, Lee, Leon, Levy, Madison, Manatee, Martin, Nassau, Okaloosa, Okeechobee, Orange, Osceola, Palm Beach, Polk, Pasco, St. Lucie, Wakulla, Walton (G.J. Steck, FDACS, pers. comm). This record from Monroe County increases the number of county records in Florida to 35, which is over half of the state.

Only two members of the genus *Zaprionus* are found in the United States, the other being *Z. ghesquierei* Collart in Hawaii (Yassin & David 2010). *Zaprionus indianus* was first collected in Florida in 2005 (Steck 2005, van der Linde et al 2006). Since then it has spread into other southeastern states and has been collected in states further north (Pfieffer et al. 2019). This species is a pest of fruits and as such is a species of concern (van der Linde et al. 2006, Pfieffer et al. 2019).

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"*Systema Dipterorum* is dead! Long live *Systema Dipterorum*!"

Neal L. Evenhuis[1] & Thomas Pape[2]

[1] J. Linsley Gressitt Center for Research in Entomology, Bernie Pauahi Bishop Museum, 1525 Bernice Street, Honolulu, Hawaii 96817-2704; NealE@bishopmuseum.org

[2] Natural History Museum of Denmark, Zoological Museum, Universitetsparken 15, 2100 Copenhagen, Denmark; TPape@snm.ku.dk

A year ago, there was a short note in *Fly Times* by Chris Thompson that he had turned over *Systema Dipterorum* (SD) to Thomas Pape and myself and was relinquishing himself from the names game – except flower flies. Not long before that, the link to the *Systema Dipterorum* database at www.diptera.org died.

In the year that has passed, we have been doing "clean up" of the data and making a few updates as well in preparation for setting up a web portal for crowdsourcing input of new records and updating others – much the same as how ZooBank currently works. Until that web portal is set up (some time in the near future), *Systema Dipterorum* is indeed been alive and kicking, but now resides (albeit temporarily) at a new url: http://www.diptera.dk

When you go there, you will see the url refresh to http://sd.zoobank.org. That is because the owner of the domain that is home to ZooBank has graciously allowed us to use that space until we can set up

www.diptera.org again or something else. Rich Pyle, who administers ZooBank has also been helping us with our new online presence and we are making some improvements and enhancements to the display and search results screens. There are still a lot of things we intend to do to make your searching adventure an exciting and pleasant one, but in the meantime we are making SD as accurate as possible, albeit slowly, and thank everyone for their patience.

We are currently posting updates every two months – and have been since February 2019. Unfortunately, there are not many taxa from recent papers that have been added because virtually all the time has been spent cleaning up the existing data, e.g., removing a few thousand duplicate entries for species; removing a few hundred duplicates for references; correcting misspellings; wrong pages; adding diacritics to author names, place names, etc. (not present in the previous SD); correcting items in wrong fields; fixing bugs to searches; updating correct synonyms; and daily tending to errors pointed out by users (there is a button on every search results page that sends an email to Thomas and me noting that there is an error to be fixed). There are a number of updates that have been made here and there as well as getting some families as up-to-date as possible: e.g., the Culicidae are updated based on Ralph Harbach's 2018 *Culicipedia*; all that remains is to add the new taxa not currently in SD; and the Afrotropical Tachinidae are up-to-date based on the 2016 Afrotropical Catalogue by Pierfilippo Cerretti and Jim O'Hara.

That said, the latest posting is Version 2.5 (28 October 2019) and has the following stats:

192,724 available species-group names [159,921 valid*; = 83.0% valid taxonomically]
19,818 available genus-group names [12,045 valid*; = 60.8% valid taxonomically]
4,316 available family-group names
33,725 references
*note that taxonomically valid names are in part subjective and will change over time

As a treat for those who are interested, we can give *Fly Times* readers some interesting stats that can be used in professional papers and trivia games.

Top 15 Dipterists in Species Described.

Author	total spp.	Valid spp.	% valid	yrs wrkd	sp/yr
Alexander, C.P.	11474	10977	95.67	72	159.4
Walker, F.	4090	2695	65.89	42	97.4
Loew, H.	3708	2840	76.59	41	90.4
Macquart, P.-J.-M.	3543	2079	58.68	37	95.8
Kieffer, J.J.	3526	2608	73.96	47	75.0
Malloch, J.R.	3323	2731	82.18	33	100.7
Robineau-Desvoidy, J.B.	3201	1488	46.49	37	86.5
Meigen, J.W.	2966	1695	57.15	36	82.4
Edwards, F.W.	2540	2296	90.39	34	74.7
Becker, T.	2520	2006	79.60	45	56.0
Curran, C.H.	2476	1903	76.86	46	53.8
Hardy, D.E.	1867	1783	95.50	66	28.3
Bezzi, M.	1795	1517	84.51	39	46.0
Wiedemann, C.R.W.	1698	1338	78.80	14	121.3
Enderlein, G.	1598	1073	67.15	43	37.2

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Investigations on the Mycetophilidae in North Central Nevada

Robin Gray

Seven Valleys LLC, Winnemucca, Nevada; sevenvalleysent@gmail.com

Since the beginning of 2017 I have been investigating the Mycetophilidae of Northern Nevada, attempting to see what species are here and learn as much as possible about their biology. I made some progress on this in 2019, and in this article would like to summarize what I have done since the early spring of 2019.

I started very early to put both emergence and Malaise traps out in the area to try to catch adult mycetophilids. I also began to collect material to run through the two Berlese funnels I made — moss, and various kinds of leaf litter from various habitats made up the majority of these materials, but I also ran anything that I thought might have even a chance of finding any mycetophilid larvae. Early in March I found mycetophilid adults in various kinds of leaf litter. I identified these as *Acomoptera*, but on reading my description of these adults in my last article in Fly Times, Dr. Scott Fitzgerald wrote to me and told me that they did not seem to fit into that genus. I sent him a male specimen which he identified as an unknown or indeterminate species of *Boletina*. The heaviest concentration of these adults was around the mosquito abatement shed in Winnemucca (Fig. 1), and they seemed to move in and out of the leaf litter with the weather.

Figure 1. Leaflitter at mosquito abatement shed in Winnemucca.

There were thousands of Burr buttercups (*Ceratocephala testiculata*) in bloom at this time only a few yards away from where I was catching these flies, and though many other insects were feeding at them, I never saw a single *Boletina* at these flowers. During the latter part of March, mushrooms of various types began to emerge. I collected a lot of these and put them in rearing chambers — no *Boletina* emerged from them. The adults of this mycetophilid were gone by the beginning of May — I felt the larvae had to be developing somewhere near, so I looked at every kind of leaf litter, dry grass, debris in the bottom of a nearby gulley, and under the bark of fallen trees, but I found no larvae. I have run a Malaise trap, beginning in March, in the area where the adults were present and will keep it up until at least the end of October. My intent is to see whether or not they have more than one generation per year. I have caught no adults in this spot or anywhere else since April. I am thinking that there may be only one generation a year, and that the larvae and pupae are in the soil; when the adults emerge toward the end of winter, they come out of the ground and take up residence in leaf litter until the weather gets warmer. But I have not actually found them doing this, I base this speculation on their apparent absence everywhere else I have looked.

Mushrooms began to appear in northern Nevada towards the end of March and were more abundant both in number and species than they were in 2018, apparently because of the wet winter and spring. When I saw a mushroom, I waited a couple of days to let the mycetophilids find it, then I dug it out, being careful to get all of it, including the bulb-like bottom of the stalk, to be sure I had the entire mushroom the mycetophilids might feed on. I put these in rearing chambers, along with the date collected and its location. The containers were either canning jars with cloth lids or freezer containers with cloth-lined holes in the sides to provide ventilation (Fig. 2). As a substrate, I used a 50/50 mix of peat moss and sand. I kept them moist by spraying them with a little water each week. These rearing chambers were kept outside on a table on the north side of a shed, away from direct sunlight.

Figure 2. Rearing chambers for mycetophilids, including canning jar (left) and freezer container (right)..

On March 31, a mushroom was seen coming up, and it was collected on April 3 and put into a rearing jar. This mushroom was tentatively identified as *Tricholoma* sp. by people on the mushroom identification page of Facebook (Figs 3–5).

Figures 3 (left), 4 (middle), 5 (right). *Tricholoma* sp.

On May 3, a number of mycetophilid adults were seen active in the jar and they were collected. Twelve of these adults were collected and identified as *Rymosia* sp. After the capture of these adults, I had a look at the mushroom and found that the cap had been seriously eaten out (Fig. 6). I examined the substrate and found 23 cocoons (Fig. 7). They were made of a fine open netting of silk covered in sand and peat moss, glued on somehow. I found one dead adult in the process of emerging (Fig. 8), and when I dissected another cocoon, I found an adult that had not emerged. I reared this particular mycetophilid out of a number of mushrooms of this genus, and out of one other species, identification uncertain.

During April, *Docosia* sp. began to emerge. One place I caught a number of them was in the emergence trap I set up over an animal burrow in February. In early May, a mating pair was taken in this trap (Fig. 9), having evidently emerged from the animal burrow over which the trap was placed. Numerous adults were taken in Malaise traps, and dozens were caught in a rain barrel I had set up. I think most of the animal burrows around here have more than one entrance, as a great variety of

insects and other arthropods turned up in this trap — mutillids, Spider wasps, bethylids, a great variety of small flies and Hymenoptera, beetles, moths, Collembola, mites, spiders, scorpions, pseudoscorpions to name a few. During the first week in October, there was an emergence of hundreds of tiny wasps. So I am not sure what the *Docosia* were doing in such animal burrows.

Figure 6 (left). *Tricholoma* sp. cap eaten out. 7 (middle). Mycetophilid cocoons. 8 (right). Adult mycetophilid on cocoon, dead while emerging.

During the first week of June, the mushrooms faded away and were no longer seen. Later in June, I hiked up into the Dun Glen range and set up a trap in a Juniper forest — I caught one mycetophilid, as yet unidentified. I also collected a large quantity of juniper needle leaf litter, which was filled with mycelium. I also collected puffballs growing out of these needles. No mycetophilids emerged from the puffballs or were found in the Juniper needles. I collected other types of leaf litter along nearby Raspberry Creek, which yielded no mycetophilid larvae either. In July, after the snow had largely receded, I hiked up into the Santa Rosa Mountains and put out three Malaise traps, ranging in elevation from 8450 feet to 9400 feet elevation. One was an island Aspen thicket in a swampy area, the second was under a lone Limber Pine (Fig. 10), and the third was in a subalpine setting at 9400 feet. I left them out eight days, then went to pick them up. The two lower traps had caught a great many insects, few of which were mycetophilids, as yet unidentified. When I got to the upper elevation, I found the trap was gone. Nothing was left but the four tent stakes that held it to the ground. I looked around for it, but found it nowhere. A big thunderstorm came through two days before I made my second trip up there; I suspect violent winds swept the trap away. I didn't like losing the trap, but my biggest regret is that I don't know if any mycetophilids were caught in that habitat. My efforts in getting there were for nothing.

Figure 9. Mating pair of *Docosia* sp. taken from an emergence trap.

During the remainder of the mosquito season, I caught a few mycetophilids in Malaise traps and EVS traps, and nothing in the samples I ran through Berlese funnels. During the latter part of September I caught a few *Mycetophila* sp, in Malaise traps. Also at that time, due to rain, some mushrooms came up, which I collected and put in rearing chambers. Nothing has emerged from them yet.

Figure 10. Malaise trap under a lone Limber Pine.

I pursue mycetophilids in my spare time; work and personal responsibilities compete heavily with this activity. I am conscious that I am missing out on things because of this. Next March when the ground thaws, I plan on digging up an animal burrow and all its branches, retrieving any nesting material and maybe soil, and running some of it through a Berlese funnel and some of it in rearing chambers, to see if the *Docosia* I am collecting from such sources breed there. Even in the best years, the mushroom season doesn't really last more than a couple of months before everything dries up. What do the *Rymosia* that develop in mushrooms do when there are no more mushrooms? And what are the adult mycetophilids feeding on? I have closely examined every type of flower in bloom when I know there are adult mycetophilids around, and have yet to see even one of them on flowers that other insects find very attractive. I think I am going to to look in the soil to find any *Boletina* larvae, but will they be there? There are habitats I have not investigated yet which I plan to hike to in 2020. I have many mycetophilid specimens to identify this winter, and I think I will have to travel to a university with a good entomology library to search the literature for papers that have keys and information on the genera I have so far uncovered here — it's impossible to get everything I need on the internet.

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MEETING NEWS

A report from the NADS 2019 Field Meeting at Bull Shoals Field Station, Ozark Plateau, Missouri (June 3–7)

Greg Courtney[1] & David Bowles[2]

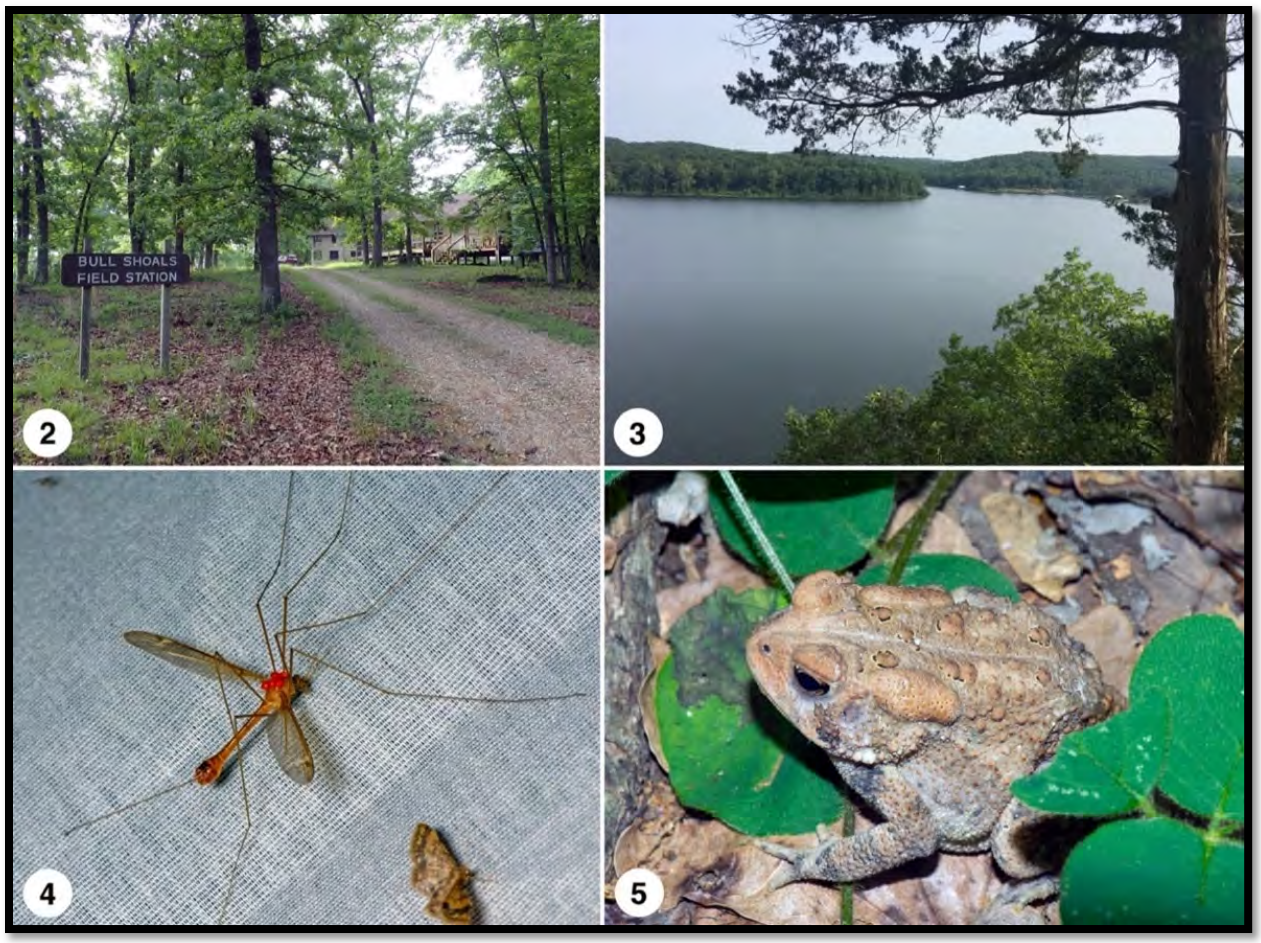
[1] Department of Entomology, Iowa State University,
Ames, Iowa, 50011, USA; (515-294-4017); email: gwcourt@iastate.edu

[2] Department of Biology, Missouri State University,
Springfield, Missouri 65897, USA; (417-836-4702); email: david_bowles@nps.gov

An intimate group of 18 Dipterists assembled at Bull Shoals Field Station (BSFS) for the 2019 NADS Field Meeting. The meeting was in the heart of the Ozark's in southwestern Missouri, only about 10 miles from Branson, a popular tourist destination. Despite the almost daily threat of rain and thunderstorms, the weather remained mostly dry, collecting was productive, and the meeting was a wonderful opportunity for colleagues to interact. Meeting participants enjoyed collecting excursions to Buffalo National River in northern Arkansas and to many "local" sites inside the Drury-Mincy Conservation Area and nearby parts of Mark Twain National Forest.

Figure 1. Participants in the 2019 NADS Field Meeting. Left to right: David Bowles, Greg Dahlem (back), Andrew Fasbender, Tristan McKnight (back), Jim Hogue, Sarah Workman (back), Juan Manuel Perilla-Lopez, John Stireman (back), Jim O'Hara, Jon Gelhaus, Brad Sinclair (back), Greg Courtney. Not pictured: Jeff Cumming, Susan Cumming, Greg Curler, Nathan Dorff, Cameron Cheri, Kornelia Skibińska.

Although the first official day of the meeting was Monday, June 3, many delegates arrived at BSFS on Sunday, June 2. Most early arrivers took advantage of good weather on Sunday to set Malaise traps and familiarize themselves with the general surroundings. That evening was a good opportunity to socialize and to hang out by a black light and sheet that Greg Courtney set at the pavilion adjacent to Drury House. The light attracted a diversity of insects, including a medium-sized moth that unfortunately found its way into the ear of Juan Manuel Perilla-Lopez. After attempting to extricate the lepidopteran interloper for about an hour, Juan Manuel and the rest of the Stireman lab loaded their vehicle and headed to the hospital in Branson. Fortunately, the emergency room staff was able to remove the moth (albeit not without some difficulty), and Juan Manuel would be back in the thick of the swarming insects during subsequent black light sessions.



Figures 2–5. 2, Bull Shoals Field Station; 3, Bull Shoals Lake; 4, Male *Tipula* (*Lunatipula*) *translucida* Doane with mites (ID by Jon Gelhaus); 5, American Toad (*Bufo americanus* Holbrook) (Figs. 2, 4, 5 by Greg Courtney, Fig. 3 by Greg Dahlem).

On Monday, meeting co-organizers David Bowles and Greg Courtney devoted part of the day to organizing the BSFS classroom / laboratory and confirming that other logistical matters were in order. Meanwhile, most early arriving delegates stayed "close to home" to investigate habitats at BSFS and adjacent parts of the Drury-Mincy Conservation Area. Sampled habitats included shorelines of Bull Shoals Lake, nearby ponds and springs (e.g., Buttonbush Pond), and various forest and glade habitats near Drury House. This also provided delegates an introduction to the local hematophagous arthropod fauna; namely, the numerous species of ticks. Although some delegates might beg to differ, ticks were actually less abundant than usual during the week. After

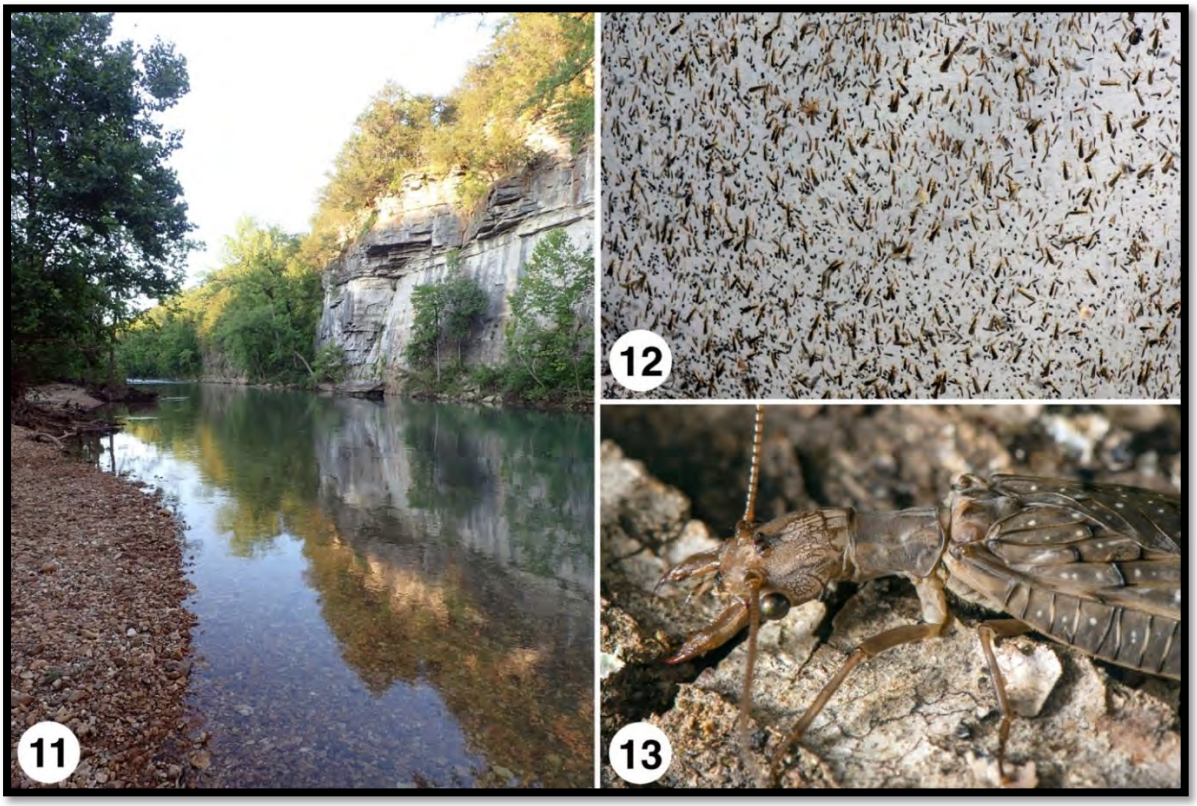
dinner at Drury House or in Branson and a brief check-in session Monday evening, Greg Courtney officially opened the meeting, Janice Greene, Director of Bull Shoals Field Station, Missouri State University, welcomed the delegates to the station, and David Bowles provided an overview of the natural history of the Ozark Plateau and an introduction to many sites that would be on the itinerary in the days to come. The night concluded with a group black-light session and informal discussions at Drury House.

Figures 6–9. Buffalo National River @ Mt. Hersey. 6, Greg Dahlem; 7, Nathan Dorff and Cameron Cheri; 8, Davis Creek; 9, Box Turtle (*Terrapene carolina* (Linnaeus)) (Fig. 6 by Jim O'Hara, Fig. 7 by David Bowles, Figs. 8, 9 by Greg Courtney).

On Tuesday, most delegates travelled to Buffalo National River in northern Arkansas. Morning activities focused on the Mt. Hersey area southeast of Harrison (Figs. 6–9), where delegates collected from a variety of riparian and terrestrial habitats along the river and along Davis Creek and Mill Creek, two small tributaries of Buffalo River. Among the collection highlights: Greg Dahlem, for the second time ever, caught a male *Cistudinomyia cistudinis* Aldrich (monotypic sarcophagid known to parasitize box turtles). In the afternoon, a few vehicles returned to BSFS, but most travelled downriver to Lost Valley Trail near Ponca. Lost Valley Trail provided a nice opportunity to sample Clark Creek, another small tributary of the Buffalo River. Habitats along the creek included several wetted rock faces and productive collecting for larvae of various "madicolous" flies (Fig. 10). Late in the day, a black light and sheet was set at Buttonbush Pond, and a black light and sheet (and mercury vapor light) was set at Drury House.

On Wednesday, some delegates remained at BSFS, focusing especially on Buttonbush Pond and Bee Creek, Mincy Conservation Area. Others ventured into the Brushy Creek watershed in nearby Mark Twain National Forest. Among the day's highlights: Brad Sinclair recorded *Trichoclinocera ozarkensis* Sinclair, a new state record (Missouri), at Bee Creek. Late in the day, and despite the threat of thunderstorms in the area, Courtney, Bowles and Sinclair set a black light trap at Bee Creek, stopped in Harrison, Arkansas, for a quick dinner, then continued to Grinder's Ferry on the Buffalo River to run a black light and sheet. Although not hugely productive for odd Diptera (e.g., too late in season for Tanyderidae), the Buffalo River BL session provided what might have been the largest number of insects Courtney has ever seen at a sheet (including BL's in the tropics). At one point, in addition to thousands of mayflies, stoneflies and caddisflies, we recorded 11 dobsonflies (*Corydalus*) on the sheet at the same time (see Figs. 12, 13)!

Fig. 10. Brad Sinclair searching for madicolous flies, Lost Valley Trail, Buffalo National River (Fig. by Greg Courtney)



Figures 11–13. Buffalo River, insects at black light, and female dobsonfly (*Corydalus cornutus* Linneaus) (Figs. by Greg Courtney).

On the last full day of collecting (Thursday, June 6), the weather forecasters finally called it. Despite the cool, wet & windy weather, a few hardy souls went out in the field... if for no other reason than to empty Malaise traps! The day was noteworthy because of a loss of power at Drury House, which impacted work in the laboratory and plans for the evening presentations. It also led some delegates to improvise in their processing of samples (e.g., Greg Dahlem used a headlamp to work on his microscope)! A delicious barbeque dinner was had under the pavilion and all seemed to enjoy that fine dining experience. The untimely power outage at Drury House forced us to move the presentation session to Mincy House, also part of Bull Shoals Field Station but just outside the conservation area boundary.

Fig. 14. Greg Dahlem & John Stireman sorting samples in BSFS classroom (Fig. by David Bowles).

The evening session included four presentations, as follows:

Andrew Fasbender "A new rheophilic genus of Orthocladiinae (Chironomidae) from the western Nearctic.

Brad Sinclair "*Trichoclinocera* Collin of the Ozark Mountains (Diptera: Empididae: Clinocerinae)"

Tristan McKnight "The hardest part is letting go: lessons learned from mark-resighting studies on robber flies (Diptera: Asilidae)"

Greg Courtney "Overview of *An Introduction to the Aquatic Insects of North America. 5th Edition* (Merritt, Cummins & Berg, eds.)"

The session concluded with a discussion of possible venues for the 2021 NADS Field Meeting. Jon Gelhaus generously offered to investigate a couple options near Philadelphia: (1) Franklin Parker Preserve in the New Jersey Pine Barrens and (2) Lake Lacawac Sanctuary in the Pocono Mountains, Pennsylvania.

Among other reported highlights:
(1) Jim O'Hara recorded close to 100 species of tachinids, which represented "probably the best ever for a NADS meeting". Much of this diversity came from the large Malaise traps set at BSFS.
(2) Several delegates caught specimens of a small hippoboscid, which Greg Courtney tentatively identified as *Lepoptena mazamae* Rondani, an ectoparasite of white-tailed deer. Unfortunately, both of Courtney's specimens died before he could capture an image to include in this report.

Friday, June 7, consisted mostly of participants packing up and travelling home or to other destinations. The field station was vacated by noon. In closing, we enjoyed hosting the meeting and greatly appreciate everyone who participated.

Figures 15–18. Miscellaneous images from BSFS. 15, Jim O'Hara preparing for field work; 16, Greg Dahlem, Juan Manuel Perilla-Lopez and Sarah Workman discussing the day's catch; 17, Greg Courtney editing images; 18, Tristan McKnight, Jon Gelhaus, Jim Hogue and Andrew Fasbender after a successful trip to Bee Creek (Figs. 15–17 by David Bowles, Fig. 18 by Jim Hogue).

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North American Dipterists Society Organized Meeting Wrap-up (St. Louis, MO, USA)

Matthew Bertone[1] & Torsten Dikow[2]

[1]North Carolina State University, Raleigh, NC, USA; matt_bertone@ncsu.edu

[2]National Museum of Natural History, Smithsonian, Washington, DC, USA; DikowT@si.edu

This year's annual NADS meeting was organized by Matt Bertone and Torsten Dikow. The below report was put together by Matt, who chaired the meeting.

The Organized Meeting of the North American Dipterists Society (NADS) was held Tuesday, November 19 from 7:00–9:00 pm during the Annual Meeting of the Entomological Society of America, in St. Louis, Missouri (USA). It was well attended by both fly enthusiasts and entomologists working on other groups. The meeting program included three 15-minute talks and four ten-minute talks.

Matthew Bertone was moderator, and encouraged brief introductions from the audience, showed some of his recent interesting fly finds, presented a recap of the NADS field meeting (from Greg Courtney), and made an announcement about funding through the Williston Diptera Research Fund (https://naturalhistory.si.edu/research/entomology/opportunities/williston-diptera-research-fund)

Figure 1. Justin Runyon's specimens.

After his introductions, the presentations began (see list below). Justin Runyon spoke about a group of wonderful little Dolichopodidae and their amazing morphology and biology (Fig. 1 – "specimens" – are they even there?). After that, Erica McAlister (who was also the ESA meeting's plenary speaker) gave a more intimate talk about exercises in extracting DNA from historical mosquito material in the NHM London, using methods that would not destroy those important specimens. Michael Skvarla (Fig. 2) then spoke about his studies on North American deer keds (Hippoboscidae), pulling together a wide variety of resources and combing lots of dead deer to summarize their distribution (he is also interested in the diseases they carry). Alex Baranowski presented some encouraging (?) data on non-target effects of the tachinid *Compsilura concinnata* affecting giant silkworm moths; perhaps these moths are rebounding in areas where the fly is present (let's hope so)? Robin Gray presented rearing studies on fungus gnats (Mycetophilidae) in North Central Nevada – surprisingly almost none were reared from mushrooms! Greg Dahlem presented musings on whether single-specimen descriptions are acceptable, giving examples from the history of flesh flies (Sarcophagidae) and highlighting his own hopes to name species for which he may not be able to find any more specimens. Lastly, Karl Magnacca (Fig. 3) showed us some disheartening data on declines in native Hawaiian drosophilids, due to loss of native plants and habitat (for which remedies are being assessed in the beautiful Pacific state).

Figure 2. Michael Skvarla discussing deer keds.

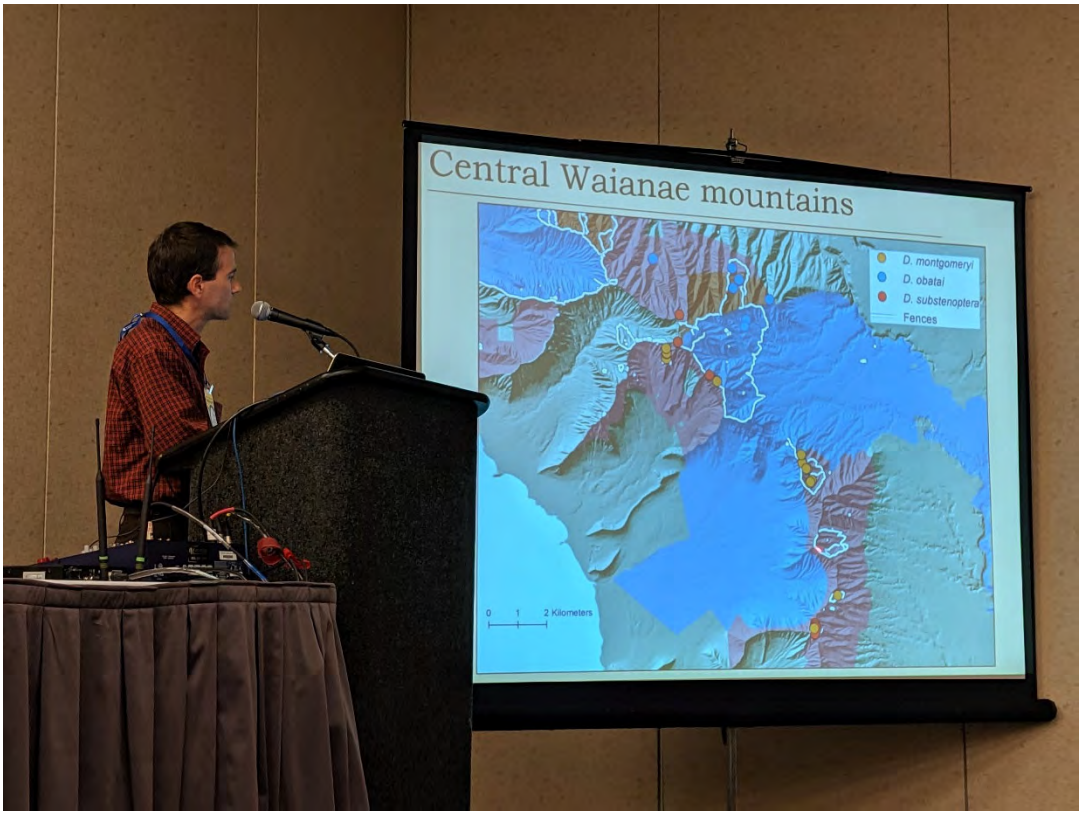

Figure 3. Karl Magnacca discussing Hawaiian flies.

All in all I think the meeting was a great success! Thank you to all who presented and attended. Please look out for next year's meeting and calls for presentations. Until then, happy fly hunting and keep advancing dipterology!

The obscure and overlooked 'micro-Dolichopodidae'
by Justin Runyon, USDA - Forest Service, Bozeman, MT

Neandersquitoes: Back to the future
by Erica McAlister, Natural History Museum, London, United Kingdom

Preliminary results of a tick-borne pathogen survey of deer keds in Pennsylvania
by Michael Skvarla, Pennsylvania State University, University Park, PA

Reduced silkmoth parasitism by *Compsilura concinnata* in New England
by Alex Baranowski, University of Rhode Island, Kingston, RI

Investigations of the Mycetophilidae of North Central Nevada
by Robin Gray, Seven Valleys LLC, Winnemucca, NV

When is describing single unique specimens acceptable? A historical perspective from the calyptrate Diptera
by Gregory A. Dahlem, Northern Kentucky University, Highland Heights, KY

Flying in the dark: Conservation of Hawaiian flies with limited data
by Karl Magnacca, Army Natural Resources Program, Schofield Barracks, HI

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Fly School II

Giar-Ann Kung

Natural History Museum of Los Angeles County, Entomology Section,
900 Exposition Blvd., Los Angeles, CA 90007, USA; gkung@nhm.org

Fly School II, organized primarily by the Entomology Section of the Natural History Museum of Los Angeles County, took place early this summer in Los Osos, Calfornia. Twenty-five students representing fourteen countries attended. Fourteen participants were students; one undergraduate and thirteen graduate students. The other eleven students ranged from museum curators and other collection professionals, researchers, technicians, to professors and teachers.

Greg Dahlem (Northern Kentucky University; USA) and Brian Wiegmann (North Carolina State University; USA) joined returning instructors Dalton de Souza Amorim (Universidade de São Paulo, Riberão Preto, Brazil), Brian Brown (Natural History Museum of Los Angeles County; USA), David Grimaldi (American Museum of Natural History; USA), and Jim Hogue (California State University, Northridge; USA) to teach the course. We were thrilled to have special guests Erica McAlister (Natural History Museum; UK) and Michelle Trautwein (California Academy of Sciences; USA) present talks. Courtney Richenbacher (American Museum of Natural History; USA) and Estella Hernandez (Natural History Museum of Los Angeles County; USA) provided invaluable support in preparation for, and during, the course.

Figure 1. Group photo.

Our base was Camp KEEP, in Montaña de Oro State Park, located on the central coast of California. From there, we could conveniently access numerous California State Park sites, the Los Padres National Forest, and other locally managed sites. We were able to sample in a variety of habitats, including the beaches, sand dunes, riparian areas, hilltops, and oak forests.

The two-week course covered the majority of Diptera families, focusing on the biology, phylogeny, economic importance, and identification of families and major genera. Students learned proper specimen preparation, field techniques, and focused collecting methods. There were also special lectures, demos, and discussion topics including the Natural History of Flies, basics of systematics, biogeography, Diptera phylogeny, and scientific illustration. During the two weeks, students collected seventy-one families. Interesting finds included Atelestidae, lots of acrocerids, some oestrids, and overall excellent hilltopping in the area.

Although the days were packed with lectures, labs, and field trips, there was also opportunity to unwind and recharge. We were lucky to have ping pong tables, a volleyball court, and other diversions at our disposal at camp. There was also a road trip up to San Simeon to see the elephant seals, and a 4th of July barbecue.

A few people felt the 7.1 magnitude Ridgecrest earthquake, although most of us did not.

The Williston Diptera Research Fund generously provided grants to help defray part of the costs to five students traveling from abroad: Heloísa Flores (Universidade de São Paulo, Riberão Preto; Brazil), Latoya Foote (University of the West Indies; Jamaica), Ana García-Ruilova (Escuela Polytécnica Nacional; Ecuador), Silvia Gisondi (Sapienza University of Rome/University of Copenhagen; Italy/Denmark), and Khutzy Munguía-Ortega (Centro de Investigación Cientifica y de Educación Superior Ensenada; Mexico). BioQuip Products and Sakura Color Products of America generously donated supplies.

We are extremely grateful to Chris Thompson and Terry Whitworth for again sponsoring Fly School. The course could not have happened without their support.

If you are interested in future course offerings, send an email to dipteracourse@gmail.com to be added to the mailing list. Hope to see you in 2021!

Figure 2 (left). “The Dome”, where labs and lectures were held. Figure 3 (right). Dave Grimaldi and Greg Dahlem working with students in the lab.

Figure 4 (left). Michelle Trautwein helping students key out specimens in the lab. Figure 5 (right). Aquatic field trip.

Figure 6 (left). Brittany Wingert and Alan Mata searching for hilltopping flies at Black Hill. Fig 7 (right). Dalton de Souza Amorim giving a quick lecture on collecting methods.

Figure 8 (left). Dave Grimaldi, Janis Matsunaga, and Jingli Xuan look for parasitic drosphilids on spittlebugs. Figure 9 (right). Erica McAlister, Denise Knapp, and Brittany Wingert enjoying the day in the field.

Figure 10 (left). David Grimaldi giving another fantastic demo on scientific illustration. Figure 11 (right). Students playing volleyball.

Fig 12. Brian Brown and Dalton de Souza Amorim playing ping pong. Fig 13. Brian Brown presenting Jim Hogue with a Certificate of Appreciation on the last day of Fly School.

Figure 14. Instructors Dalton de Souza Amorim, Brian Brown, Greg Dahlem, David Grimaldi, and Jim Hogue. (Missing: Brian Wiegmann).

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OPPORTUNITIES

New opportunities for Diptera research at McGill University

Jessica P. Gillung

Department of Natural Resource Sciences, McGill University,
Sainte-Anne-de-Bellevue, QC H9X 3V9, Canada

It is with immense joy that I announce I will be joining McGill University's Department of Natural Resource Sciences (NRS) in January 2020 as Assistant Professor and Director of the Lyman Entomological Museum. I'll be the newest addition to an already productive and interdisciplinary team of dipterists pursuing outstanding research on fly diversity, taxonomy, evolution, and ecology.

Both NRS and the Lyman Entomological Museum have a long and impressive track-record of dipterological research, ranging from taxonomy and phylogenetics to pollination biology and ecology of Arctic systems. Some of the remarkable research currently being conducted on site include Linley Sherin's M.Sc. work on fly succession in post-burn wildfire sites; Anthony Zerafa's M.Sc. research on high arctic field ecology, a fauna comprised mostly of flies; Vinko Culjak Mathieu's M.Sc. research on pollination ecology in the Canadian Arctic; Sarah Loboda's Ph.D. research on the effect of climate change on Arctic Muscidae in Greenland; Anna Solecki's Ph.D. work on the community structure and diversity of flies in Northern Canada at varying spatial scales; Stephanie Boucher's Ph.D. research on fly diversity, ecology and systematics (with a focus on Agromyzidae); and Morgan Jackson's Ph.D. and postdoctoral research on fly taxonomy and systematics (with a special focus on Micropezidae).

The Lyman Entomological Museum is one of the largest insect collections in Canada, housing approximately 3 million specimens and being a major research and training centre in insect systematics, biodiversity, and ecology. Highlights of the collection include Diptera (over 400,000 specimens, worldwide in scope), Coleoptera (250,000 specimens, strong on the Nearctic and west African faunas), Lepidoptera (worldwide collection of butterflies), Orthopteroid orders (150,000 specimens, worldwide in coverage), and soil microarthropods (large slide collection, especially rich in mites).

Lasia sp. (Acroceridae). Photo by Shaun Winterton.

My initial motivation to become an entomologist was inspired by my desire to be a taxonomist, and I feel fortunate to join the Lyman Museum. I completed my Ph.D. at the Bohart Museum of Entomology at the University of California Davis. My dissertation research focused on the evolution and taxonomy of spider flies (Acroceridae, see figure), a remarkable group of

parasitoids specialized in spiders. My work combined the study of fossil evidence with molecular and morphological phylogenetics to gain insights into key evolutionary adaptations, relationships, and origins of spider flies.

I will soon be recruiting students for both M.Sc. and Ph.D. positions in Entomology to join my lab. Students will have the opportunity to conduct research in the highly collaborative and interdisciplinary NRS Department, and to take advantage of the resources and infrastructure of the Lyman Museum. My research at McGill University will focus on discovering and documenting fly biodiversity while resolving the evolutionary patterns and processes that underlie speciation, diversification, and trait evolution, and I am particularly interested in parasitoid flies. For more information about the Master's and Ph.D. programs at the NRS Department, check out https://mcgill.ca/nrs/graduates/graduate, and for general information about McGill University's graduate program admissions, see https://mcgill.ca/gradapplicants/apply/prepare.

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DIPTERA ARE AMAZING!

We had several great submissions from R. Isaí Madriz (four photos) and Robert Parks (one photo), presented here. Coincidentally both Isaí and Bob made their photos in Patagonia, but different ones! After the photos, there is a comic used with permission of the artist, Joshua Barkman (False Knees).

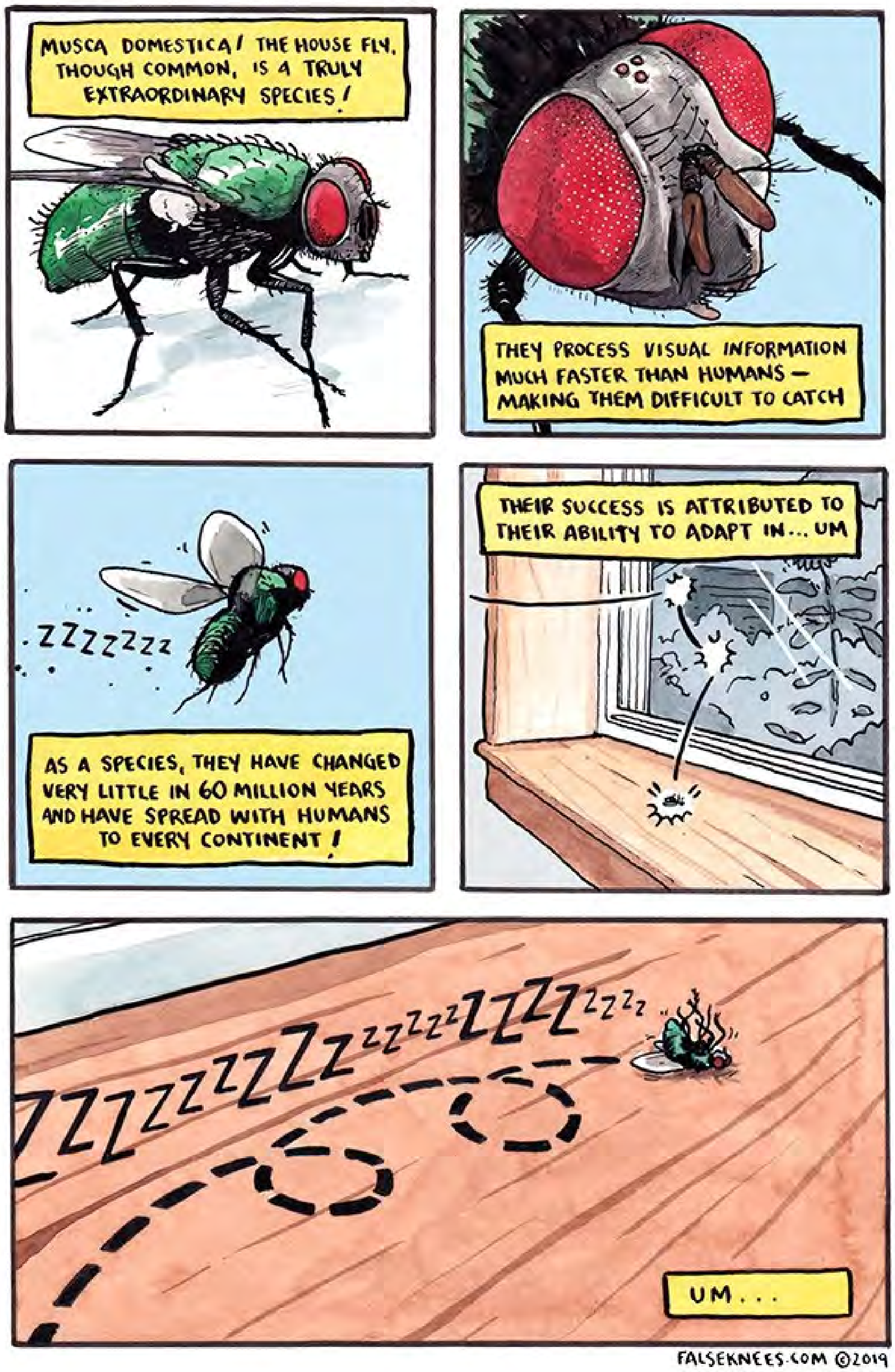
A female *Mydas arizonensis*, photographed by Bob Parks in Harshaw Road about 6 miles south of Patagonia, Arizona last September.

Auraucoderus gloriosus. (Tanyderidae) and *Chiasognathus grantii* (Lucanidae) from Patagonia (Aysén, Chile). Photo by R. Isaí Madriz.

Nothodixa sp. (Dixidae) from Patagonia (Aysén, Chile). Photo by R. Isaí Madriz.

Shannonomyia sp. (Limoniidae) from Patagonia (Aysén, Chile). Photos by R. Isaí Madriz.

Disclaimer so dipterists don't start a riot! Joshua Barkman has learned after-the-fact that this is *Lucilia sericata*, not *Musca domestica*, and that the diversification of oestroid flies didn't even begin until 50 million years ago!



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BOOKS AND PUBLICATIONS

Once again here is your biannual serving of dipterological readings. We hope they prove useful in whiling away the winter hours (assuming you're in the northern hemisphere).

One item that deserves particular mention is the newly released 'Field Guide to the Flower Flies of Northeastern North America' by Jeff Skevington, Michelle Locke, Andrew Young, Kevin Moran, William Crins and Steve Marshall. It is a beautiful piece of work (and is already winning awards!) and should be a required addition to any dipterologists library. Further details on the book can be found here: https://press.princeton.edu/books/paperback/9780691189406/field-guide-to-the-flower-flies-of-northeastern-north-america

As usual if we have not included a paper that you think should have been here please feel free to pass it along to Chris (chris.borkent@gmail.com) and we will include it in the next issue. Unfortunately, the online resources do not always catch everything and are a couple of months behind. We also apologize for the missing diacritics in some author's names, unfortunately this is a product of searching in Zoological Record and Web of Science, where they are removed.

Acurio, A.E., Rhebergen, F.T., Paulus, S., Courtier-Orgogozo, V. and Lang, M. 2019. Repeated evolution of asymmetric genitalia and right-sided mating behavior in the *Drosophila nannoptera* species group. Bmc Evolutionary Biology **19**: 14. doi:10.1186/s12862-019-1434-z.

Adler, P.H., Srisuka, W., Van Lun, L.L., Takaoka, H. and Saeung, A. 2019. High-elevation chromosomal diversity of black flies (Diptera: Simuliidae) in Thailand. Insect Systematics and Diversity **3(3)**: 1-U11. doi:10.1093/isd/ixz004.

Adler, P.H., Takaoka, H., Sofian-Azirun, M., Chen, C.D. and Suana, I.W. 2019. Evolutionary and biogeographic history of the black fly *Simulium wayani* (Diptera: Simuliidae) on the island of Timor. Acta Tropica **193**: 1-6. doi:10.1016/j.actatropica.2019.02.017.

Al Sayad, S. and Yassin, A. 2019. Quantifying the extent of morphological homoplasy: A phylogenetic analysis of 490 characters in *Drosophila*. Evolution Letters **3(3)**: 286-298.

Ale-Rocha, R. and Pollet, M. 2019. First records of Ropalomeridae (Diptera, Acalyptratae) from French GuianaIn Touroult J. (Ed.). "Our Planet Reviewed" 2015 large-scale biotic survey in Mitaraka, French Guiana. Zoosystema **41(1)**: 1-5. https://doi.org/10.5252/zoosystema2019/41/1.

Amancio, G., Hernandez-Ortiz, V., Aguirre-Jaimes, A., Guevara, R. and Quesada, M. 2019. Feeding specialization of flies (Diptera: Richardiidae) in aroid infructescences (Araceae) of the Neotropics. Journal of Insect Science (Tucson) **19(3)**: 28.

Ambrozio-Assis, A., Lopes, B.C., Amaral, A.P., Pinho, L.C., Peeters, E. and Necked-Oliveira, S. 2019. Preferences for anuran calls in hematophagous corethrellids (Diptera: Corethrellidae) from Southern Brazil. Austral Entomology **58(3)**: 622-628. doi:10.1111/aen.12376.

Aragao, A.D., Neto, J.P.N., Cruz, A.C.R., Casseb, S.M.M., Cardoso, J.F., da Silva, S.P. and Ishikawa, E.A.Y. 2019. Description and phylogeny of the mitochondrial genome of *Sabethes chloropterus, Sabethes glaucodaemon* and *Sabethes belisarioi* (Diptera: Culicidae). Genomics **111(4)**: 607-611. doi:10.1016/j.ygeno.2018.03.016.

Arkia, S., Siahkalroudi, S.Y. and Kheradpir, N. 2019. Chironomidae (Insecta: Diptera) biodiversity at generic level in Lar River, Tehran Province with introducing two new genera for Iranian fauna. Journal of Wildlife and Biodiversity **3(1)**: 31-39. doi:10.22120/jwb.2018.93291.1031.

Augustinos, A.A., Tsiamis, G., Caceres, C., Abd-Alla, A.M.M. and Bourtzis, K. 2019. Taxonomy, diet, and developmental stage contribute to the structuring of gut-associated bacterial communities in tephritid pest species. Frontiers in Microbiology **10**: 13. doi:10.3389/fmicb.2019.02004.

Barahona-Segovia, R.M. and Barcelo, M. 2019. A new flower fly species of *Cepa* Thompson & Vockeroth (Diptera: Syrphidae from the Valdivian evergreen forest hotspot, Chile. Zootaxa **4612(3)**: 431-439. doi:10.11646/zootaxa.4612.3.9.

Baranov, V., Kvifte, G.M., Muller, P. and Bernal, X.E. 2019. A new species of fossil *Corethrella* (Diptera, Corethrellidae) from mid-Cretaceous Burmese amber. Cretaceous Research **101**: 84-91. doi:10.1016/j.cretres.2019.05.002.

Baranov, V.A., Schadel, M. and Haug, J.T. 2019. Fly palaeo-evo-devo: immature stages of bibionomorphan dipterans in Baltic and Bitterfeld amber. Peerj **7**: 49. doi:10.7717/peerj.7843.

Barbier, E., Graciolli, G. and Bernard, E. 2019. Structure and composition of Nycteribiidae and Streblidae flies on bats along an environmental gradient in northeastern Brazil. Canadian Journal of Zoology **97(5)**: 409-418.

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Canadian National Collection of Insects
Agriculture & Agri-Food Canada
K.W. Neatby Building, C.E.F.
Ottawa, Ontario, CANADA, K1A 0C6

Tel.: (613) 759-1795
FAX: (613) 759-1927
Email: james.ohara@canada.ca

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