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SCOPE

Fly Times accepts submissions on all aspects of dipterology, providing a forum to report on original research, ongoing projects, Diptera survey activities and collecting trips, interesting observations about flies, new and improved methods, to discuss the Diptera holdings in various institutions, to make specimen requests, to advertise opportunities for dipterists, to report on or announce meetings or events relevant to the community, to announce new publications and websites, to examine the historical aspects of dipterology and Diptera literature, to honor our recently deceased colleagues, and anything else fly-related that you can think of. And of course with all the images you wish to provide.

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INSTRUCTIONS TO AUTHORS

Although not a peer-reviewed journal, all submissions are carefully considered by the editor before acceptance. We encourage submissions from dipterists worldwide on a wide variety of topics that will be of general interest to other dipterists, as an attractive medium for dipterists of all types to showcase their activities.

The requirements for submission are simple. Please send me a single-spaced text file (doc, rtf, odt preferred) along with separate image files (jpg, tif, png preferred).

Following are some specific dos and don'ts, bearing in mind that consistency among manuscripts is important:

1) *Do not* embed images into the text file (but *do* indicate in the text file approximately where each image should be placed).
2) *Do* submit image files of a reasonable size (no more than about 2MB per image file, or larger for full plates).
3) *Do not* use embedded styles (e.g., the various heading styles, small caps, paragraph spacing, etc.). *Do* limit styles to italics, bold, and (if you must) underline, and single-spaced.
4) *Do not* use different fonts, different font-sizes, or different colored fonts as headings. *Do* use Times New Roman, 11.5 point, black.

The approximate deadlines for submission are mid May and mid November, although this is flexible up to the time of publication (which will generally be mid June for the spring issue and mid December for the fall issue). For larger manuscripts your submissions may be considered for inclusion in the *Fly Times Supplement* series. Note, submission of a manuscript to *Fly Times* or *Fly Times Supplement* grants the Dipterists Society the non exclusive right to reproduce these contributions in whole or part

Please submit manuscripts to the editor-in-chief at:
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Our Mission is to advance the scientific study, understanding and appreciation of the insect order Diptera, or true flies. To accomplish this, we aim to foster communication, cooperation, and collaboration among dipterists, and to promote the dissemination and exchange of scientific and popular knowledge concerning dipterology.

As an **international society**, there are no boundaries, and our core activities are geared towards all dipterists, not a subset. We aim to provide a common stage for all people interested in flies, a place where our community can closely interact. Among our core activities, we produce Society publications such as this one (as well as the *Fly Times Supplement* and *Myia*), facilitate or organize Society and other Diptera-related meetings and events, provide grants and awards in support of dipterological activities and achievements, perform outreach activities and provide educational opportunities and resources, and maintain an organizational website, an online Directory of World Dipterists, a dipterists mailing list server, and social media presence. In these efforts, we as a group can make our society as successful as we want!

A note about Society membership – To thrive as an organization and to provide all the resources we can for the dipterological community, we need your support through becoming a member (https://dipterists.org/membership.html) or making donations (https://dipterists.org/support.html). Please see our website to understand our vision for our society!

From the Editor – Welcome to the latest issue of *Fly Times*! As usual, I am very impressed with the variety of excellent submissions, and I hope they are enjoyable to the readers. Please consider writing an article or two (or 10 like Larry Hribar this issue!) for the next issue, which is slated for spring of 2026. And for larger works, please consider the *Fly Times Supplement* series, found at https://dipterists.org/fly_times_supplement.html.

Thank you to Zachary Dankowicz for another excellent cover photo! Moving forward, I encourage the photographers out there to submit images for the cover – keep dimensions in mind – they will be produced at 8-1/2 X 11 inches (*Fly Times* page size). Photos not used for the cover can still be included in the Diptera Are Amazing section. For now, I'll be changing up the covers issue to issue, so please feel free to send your design ideas to sgaimari@gmail.com (cc editor@dipterists.org).

Cover photo – Male *Diostracus prasinus* Loew (Dolichopodidae) from Kephart Prong Trail in the Great Smoky Mountains National Park, North Carolina, USA. This species has fascinatingly pubescent palpi which shimmer in the light and are used for courting interactions. Photograph by Zachary Dankowicz.

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CONTENTS

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NEWS AND RESEARCH

Guidelines for the use of pan traps to collect flying insects, in particular Diptera (Dolichopodidae)

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Summary

In contrast to sweep nets and Malaise traps, pan traps are much less frequently used to collect Diptera. This is quite surprising as they are not only complementary to the other techniques, but for some families like Dolichopodidae, they are by far the most productive collecting method. Moreover, multiple other fly families are also captured readily with pan traps, sometimes in numbers. In this paper the field work with pan traps is described, including the selection of sampling sites, the installation of the trap units and their servicing. Pan traps are always installed at soil surface level, certainly to collect Dolichopodidae, as the highest diversity is encountered there. Traps are arranged in units comprising one yellow, one white and one blue trap. In areas rich in different habitats, more sampling sites are selected and investigated with 5 pan traps units, while in less diverse areas, less sampling sites are selected but sampled with 10 units. Information about the most suitable fixative fluids is given. Two check lists are also provided, one for the installation of the traps and one for the servicing. Finally, some successes and failures of past pan trap sampling are shortly discussed.

Introduction

Though there is a wide array of specialized devices to collect particular fly species or even entire families, overall the most widely applied techniques for this order remain sweep nets and Malaise traps. Both have their benefits and restrictions, but in particular Malaise traps have been the standard method to collect Diptera in many large scale and multitaxon surveys (e.g., Borkent et al. 2017; Brown et al. 2018; Touroult et al. 2018, 2023; de Souza Amorim et al. 2022). For some unknown reasons, pan traps on the other hand have much fewer supporters and have only recently been added as an integral part of the collecting methods set-up of international expeditions (e.g., Touroult et al. 2018, 2023; Leponce et al. 2024). Nevertheless, for a number of Diptera families this method nicely complements Malaise trap sampling (e.g., Pollet & Grootaert 1987), and also provides relevant information on the ecology and biology of the collected species, certainly in combination with other collecting methods. In addition, not only can different features of this method be adjusted according to the research question (size, colour, installation height), but for some families it appears by far the most productive technique, in terms of both species and certainly specimens. One of these families is Dolichopodidae, but multiple other families are also often found in large numbers in these traps. So it remains bizarre why not more dipterists apply it and if lack of experience with this method is the reason, then I hope that this note might provide them with handy tips and tricks.

In what follows, I'll describe the way I operate in the field (mostly Palaearctic realm and Neotropics) when I carry out a pan trap sampling campaign with focus on Dolichopodidae.

Basic research question
My main aim is mostly to assess the **species diversity** (number of species and abundances) and to gather information on the **ecology** and **habitat preference** of the (often undescribed) species. For that reason, multiple sampling sites are selected and pan traps of three different colours are employed. Next to pan traps, dolichopodids are also collected by sweep nets and sometimes Malaise traps, but pan traps remain the main sampling device.

The pan traps
Since 2009 I have been using a type that has been manufactured by Amscan in Germany (Figure 1) as a disposable plate for BBQs (I never used them for this purpose, by the way). I got in touch with these traps (of a smaller size) around 2001 thanks to Dr Scott Brooks (CNC, Ottawa, Canada) and purchased my first series in 2009 via contacts in Illinois before they became available in Belgium and The Netherlands. About 5 years ago, Amscan ceased the production of these plates as a result of new European rules on sustainability, but fortunately, the Muséum national d'Histoire naturelle (Paris, France) was so clever to buy the entire stock. For that reason, these traps will be available for any future MNHN expeditions.

Figure 1. Pan traps (Amscan) used by author during expeditions in the Palaearctic and Neotropics since 2009.

The trap itself is very light weight, has an inner diameter of 15 cm with upper rims of 1 cm wide, and a depth of 4 cm. Next to its light weight and sufficiently large capture surface, another major advantage of this type is its stackability. And as I tend to use them during expeditions in the tropics, these aspects are of paramount importance (one hundred of these traps do not take too much space and are not too heavy). On the other hand, the traps are rather fragile, and last for at most two seasons when not operated in direct sunlight (which turns them brittle). There are definitely sturdier types that last longer, but cannot be piled up so easily, are heavier or do not come in the necessary colours. But, e.g., cottage cheese or ice cream containers also work well, if you are content with white traps.

This type of trap is **not UV reflectant** which means that pollinators are not very attracted to it. But like in pollinator studies, I always use three colours: yellow, white and blue. The main reason is that each colour attracts other species: while arboreal species are mainly attracted to blue traps and most other species to yellow traps, white traps produce a mix of both arboreal and non-arboreal species (Pollet, unpubl. data). In other words, white traps provide a better idea about the dolichopodid diversity than yellow ones (which tend to produce the highest numbers, often of the same species). It is always worthwhile to make **2-3 tiny holes** just below the upper rim of the trap, to allow surplus liquid (caused by heavy rainfall) to flow away while leaving the yield in the trap.

Since the early 1990'ies (while using another type of trap), I have been installing the traps **at soil surface level**. As a matter of fact, two methodological studies (Pollet & Grootaert 1987, 1991) revealed that the majority of dolichopodid species mainly occurred on low foliage or were soil-dwelling.

Selection and number of sampling sites

In case of Dolichopodidae preferably **wet or moist sites** are investigated as they hold the highest diversity. Especially interesting are seeps and beds and banks of streams (Figure 2), which can be extremely productive. On the other hand, dry habitats should not be forgotten, as their faunas include mostly entirely different species than wet sites.

Fig. 2A. Seep in dry forest

Fig. 2B. River bank

Fig. 2C. Seep on rocky outcrop

Fig. 2D. Stream bed in pozzine landscape

Figure 2. Selection of suitable sites for sampling with pan traps for Dolichopodidae (Our Planet Reviewed Corsica survey, Alta Rocca 2019)

In order to achieve the highest species diversity at a particular site with pan traps, as many (micro)habitat types as possible are investigated. There is, of course, the constraint of logistics and post-processing of the samples that must be taken into account. On this basis, it has been decided that per study site, at least one and at most 8 sampling sites are selected for pan trap sampling.

In order to decide exactly how many sampling sites will be in place, the best approach is to start by exploring the study area soon upon arrival. If habitat diversity is low, then at most 4 sampling sites are investigated, each with 10 pan trap units (for more information, see further). If habitat diversity is high, it is better to select more sampling sites (up till a max. of 8) but each with only 5 pan trap units.

In summary, per study site the total number of pan traps employed ranges from **30** (one sampling site, with 10 pan trap units) to **120** (from 4 sampling sites with each 10 pan traps units to 8 sampling sites with each 5 pan trap units).

Pan trap installation
In each sampling site, thus 5 (or 10) pan trap units are installed. One **pan trap unit** consists of one blue, one white and one yellow pan trap (see Figs 3, 5B). Traps of the same unit are placed closely together (Figure 3), at less than 25 cm from each other, to avoid a possible bias caused by differences in microhabitat. Two successive pan trap units are installed at a distance of 3-5 m from each other.


Fig. 3A. Trap sample set at seep site


Fig. 3B. Trap sample sets along seep

Figure 3. Operational pan trap units (Our Planet Reviewed Corsica survey, Alta Rocca 2019).

Each trap is **slightly dug** into the soil (to decrease evaporation but also to increase access for soil-dwelling invertebrate species) and fixed with **2 metal pins**. On rocky substrates, pebbles or small rocks are used instead to stabilize and fix the traps. I manufacture these metal pins myself by cutting them from galvanized wire (Ø 1.6 – 1.8mm is fine), straighten them and tucking one end in a 90° angle. If these are stored dry after the field campaign, they last for several years.

The traps are filled to ¾ with a fixative liquid with a composition that largely depends on the duration of the sampling. In all cases, it does contain a sufficient amount of liquid colourless and odorless **detergent** (about 5-10 good squeezes per 10 liters of fixative liquid) in order to lower the surface tension (crucial for the capture of flying insects)!

These are the fixative liquid compositions that work:

- 1 – at most 2 days: ordinary water (+ detergent)
- 3–4 days: salty water (+ detergent) – make sure you add enough salt to the water
- from 5 days onwards: propylene glycol or – if allowed/available – a mild formaline solution (+ detergent)

The **concentration** of the formalin solution (= 40% formaldehyde) used depends entirely on the duration of the collecting and the weather conditions:

- < 7 days: 2.5% might do (= dilute the formalin solution by 40 times)
- 7 days: 5% (= dilute the formalin by 20 times)
- ≥ 7 days (best not longer than 14 days): 10% (= dilute the formalin by 10 times)

In the **rainy season**, it is best to service the traps more regularly than during dry periods. Also, after heavy rainfall, it is recommended to check the status of the traps as soon as possible, especially if they are installed close to streams or in sites that might get inundated. On the other hand, in very warm and dry periods, it is often good to refill the traps on a regular basis to prevent them from drying up largely or entirely.

If traps are serviced but remain operational afterwards, then the fixative liquid is best **recycled**. This can be done in two ways: (i) pouring the yield through a sieve (see Figure 4) while collecting the liquid in a fresh trap that replaces the one that is emptied or (ii) collecting the yields from all traps in a sampling site first, while catching the liquid from all of them and then refill the traps with the recycled fixative liquid (this requires an empty gallon container). It is always recommendable to have some fresh fixative fluid held aside in case the liquid has been severely diluted (by rain), decreased in volume (from drying out) or changed in colour (due to fallen leaves).

Collection of the yields

Per sampling site, yields of 5 traps of the same colour are **pooled** into one pan trap sample. As such, a sampling site with 5 pan trap units (= 15 pan traps) produces 3 samples per servicing, one with 10 pan traps units provides 6 (two for each colour).

For servicing the traps, you'll need at least **a sieve** to capture the collected insects (Figure 4), **a small painter's brush** to gently manipulate this insect mass, and a **collecting device** (jar, whirlpak) to store the yield. Do not forget to add a (preferably preprinted) **label** to the sample. If formalin is used, it is recommended to carry out the procedure with disposable gloves.

Fig. 4A. Pouring yield of pan trap into sieve.

Fig. 4B. Positioning yield in one half of sieve to facilitate the transfer into storage receptacle.

Fig. 4C. Transfer of yield into storage receptacle with painter's brush.

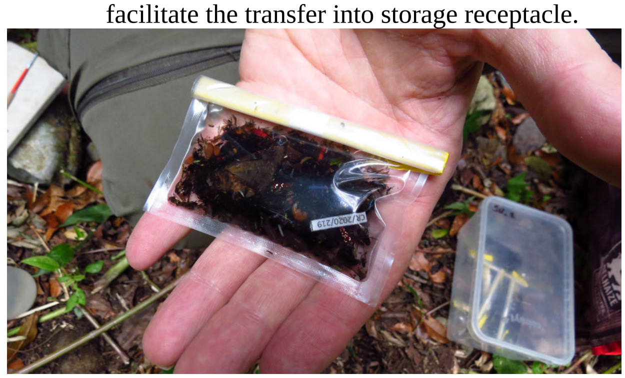

Fig. 4D. Pooled sample of 5 yellow pan traps in whirlpak.

Figure 4. Procedure for servicing pan traps (San Gerardo de Dota, Costa Rica, 2020)

The sample collecting procedure is as follows: move the pins away, lift the operational pan trap (i.e. the trap with the yield) and pour its content into the sieve (Figure 4A). Repeat this action for the following 4 traps of the same colour (yes, it implies a bit back and forth walking). Subsequently submerge the sieve into liquid (e.g., in stream, or an pan trap) so that the entire yield accumulates to one side (Figure 4B) and then transfer the yield gently with the small painter's brush into the collecting receptacle (Figure 4C). For storage, either use a collecting jar with a sufficiently large opening, or a whirlpak (207 ml bags are perfect) (Figure 4D). Add 75% alcohol solution to the sample.

Finally add a **preprinted label** to the sample and you're ready (finally ...). You can choose your own label format. I mostly use the following format: [country code] / [year] / [location code] / [site code] / [number and colour of pan traps] / [date(b)-date(e)] / [initials of collector]. For example, CR/2024/MV/01/5YPT/10-15.iii.2020/MP. Alternatively, the following format is sometimes used as well (Figure 4D): [country code] / [year] / [serial number] / [initials of collector] though it is more tricky (the collector needs to know exactly what sample corresponds with the preprinted sample code).

It is truly important to collect as much **information** as possible on the sampling sites, including photographs of the habitat, the installed pan traps and latitude – longitude – elevation coordinates.

Field check lists
Below, two check lists are provided for field work encompassing pan traps, one for the installation and one for the servicing of the traps. Screening them prior to field work should avoid inconvenient situations in the field (e.g., forgetting the sieve for servicing would be rather disastrous). They include the **necessary equipment** for the investigation of one minor sampling site (**multiply by 4** in case of 4 sampling sites, *except for the equipment in italics*):

Installation phase (per sampling site)

- 5 blue pan traps
- 5 yellow pan traps
- 5 white pan traps
- 30 metal pins
- one gallon of 5 liters of fixative liquid (do not forget to add the detergent beforehand!)
- *scissors (to cut the vegetation)*
- *camera (to make pictures of habitat and pan trap settings)*
- *apparatus with GPS (to record the exact latitude, longitude and elevation); I usually record only the central pan trap unit, not the others.*

Servicing phase (per sampling site)

- *sieve*
- *small (painter's) brush*
- 2 polymer gloves (in case you used formalin solution and want to work entirely safe)
- 3 preprinted labels (1 for the pooled blue, yellow and white trap yield)
- 3 collecting jars or whirlpaks (to store the samples)
- about 0.1-0.2 liter of alcohol 75% (to add to the samples)
- some spare pan traps of each colour (only useful if the pan trap campaign is continued after servicing; they are used to replace possibly damaged, destroyed or vanished ones, but can also be very handy to recycle the fixative liquid (see above).

- *funnel and empty gallon container (in case you want to recycle the fixative liquid for another sampling period or you intend to collect the liquids at the removal of the traps)*
- *camera (e.g., to make pictures of the traps and their yields)*
- *bag, box or container to store the samples and collecting gear.*

Field observations, successes and failures

In suitable habitats for Dolichopodidae, specimens are trapped in the foam on top of the trap seconds after the trap is filled with the fixative fluid, sometimes in numbers and especially in the yellow traps. This has been observed on many occasions. However, the traps do not remain that attractive during the entire sampling period, but it's not clear why. On one occasion (Figure 5A), one single yellow pan trap yielded 136 specimens of 9 species in only 45 minutes.

Fig. 5A. Yellow pan trap 45 minutes after installation (Vallei van het Merksken, the Netherlands, 13/6/2015)

Fig. 5B. Pan trap unit after 14 days of operation (Bos t'Ename, Belgium, 10/7/2015)

Fig. 5C. Yellow pan trap after 4 days of operation (Serra-di-Scopamène, Corsica – France, 30/6/2019)

Fig. 5D. Regularly flooded pan traps in palm swamp (Mitaraka, French Guyana, 6/3/2015)

Figure 5. Successes and failures with pan traps in the field.

Two of the most successful (read: productive) pan trap surveys I conducted in the past three decades remain those of Bos t'Ename (Belgium, 2015) and Alta Rocca (France, Corsica, 2019). During the first one, 90 pan traps were operational in 7 sampling sites in mainly moderately humid to humid deciduous forest between early May and early October 2015. They assembled 77,058 specimens of 116 species (Figure 5B). During the Alta Rocca expedition, the 2019 edition of the La Planète Revisitée (Our Planet Reviewed) program, 17 sampling sites at four locations were investigated using 255 pan traps in higher elevation biotopes ranging from dry oak forest, beech forest, fire forest and alpine meadows (pozzines) between 23 and 30 June 2019. This survey produced 13,659 specimens of 49 species. In some sites the traps were so attractive that after four days, flies (different families) piled up in the traps above the liquid surface, especially in the pozzine habitat (Figure 5C). In both cases, the weather conditions (long hot periods without any or much rain) in combination with specific site features (humid, often including small streams) seem to have been the most important success factors. Many more sites in both the Palaearctic and Neotropics were investigated with pan traps during the past two decades, but samples of several of these surveys still remain unprocessed.

The pan trap campaign in Mitaraka (southwestern French Guiana) remains the biggest disappointment in terms of return on investment: between 25 February and 10 March 2015, no less than 280 pan traps collected only 1,179 specimens of 49 species. The main reason for these low numbers was clearly the nearly daily heavy rainfall which flushed most of the yields and sometimes also flooded the traps entirely (Pollet et al. 2018; Figure 5D). The same observation was made during the most recent Our Planet Reviewed expedition on the isles south of Guadeloupe (LPRIG 2024), where downpours ruined most pan trap efforts on Marie-Galante (Justin Runyon, pers. comm.).

Whirlpaks are excellent as storage receptacles for this kind of sampling. They are light weight, do not take a lot of space and when wrapped well (i.e., not containing a lot of air), prevent damage to the included invertebrates. So they are perfect in case the samples must be transported long distance. If these samples are in turn stored in closed containers or ziplock bags, they remain in a good state for several months (up till half a year) but not for years. So if you aim at long term storage, samples are better transferred to other containers that cannot dry out.

Conclusive remarks

Under optimal conditions, i.e., suitable habitats, sufficiently high temperatures and no heavy rainfall, pan traps are indispensable in surveys aiming at establishing the diversity of Dolichopodidae in different realms. Due to the characterictics of their populations, in general, species richness in these traps in higher in the Neotropics, while abundances are higher in the Palaearctic. Certainly in case of inventories that last only a few days, pan traps are way more productive than Malaise traps. Next to Dolichopodidae, multiple other fly families are captured readily in these traps, as so many Diptera researchers can testify (who received samples from me over the years). Which non-dolichopodid taxa are most attracted to blue, white and yellow should be revealed by completed identification lists returned by experts who received pan trap samples. It is still my firm intention to carry out a comprehensive analysis on these data in order to make recommendations on the use of pan traps for Diptera in general.

Acknowlegments

Our Planet Reviewed – Corsica 2019-2021 survey was organized by the Muséum national d'Histoire naturelle (MNHN) in collaboration with and funded by the Collectivité de Corse (CdC) and the Office français de la Biodiversité (OFB) (previously known as the Agence française de la Biodiversité – AFD). The following logistic partners assisted with field work: the communes of Alta Rocca (Serra di Scopamène, Zonza and Zicavo) and Tartagine (Olmi-Capella and Mausoléo), the Office de l'Environnement de la Corse (OCIC et CBNC), the Direction Régionale de l'Environnement de l'Aménagement, the Direction du Logement (DREAL) and the Office National des Forêts (ONF). Special thanks are due to the mayor of the village of Serra di Scopamène, Mr Jean-Paul Roccaserra, the 'Communauté de Communes de l'Alta Roca' and its Écogardes have provided the necessary infrastructure and invaluable support during the expedition. I am grateful to the leaders of the expedition, Dr Julien Touroult and Dr Olivier Pascal, for their professional guidance, and the entire researcher team for the great company. Many thanks are due to the King Leopold III Fund for Nature Exploration and Conservation (Brussels, Belgium) for its financial support of the "Dolichopodid survey of lowland and montane rain forests in Costa Rica (Diptera: Dolichopodidae)" expedition in Costa Rica (7-28 March 2020). The Agency for Nature and Forest (ANB, Brussels, Belgium) launched the "Inventarisatie van de biodiversiteit in het Bos t' Ename (ANB_OV_2015_01)" survey (Belgium) while Mr Guido Tack kindly guided this investigation. Finally, none of the above mentioned pan trap field work would have been possible without the assistance and advice of my wife, Anja De Braekeleer, to whom I am truly indebted.

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Membership in the Dipterists Society

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The Dipterists Society was incorporated as the North American Dipterists Society on 27 November 2019. During the ramp up of the Society, there were not yet any "members". Instead I was working on all of the tasks and paperwork to register as a US 501(c)(3) nonprofit, among other things, to become a fully recognized organization. Since much of this required funds, I became the first real (i.e., paying) member on 8 May 2020, depositing $5000 into the newly opened Society bank account. Once our nonprofit status was given, annual membership was opened up with two categories – Founding Members ($150) and Individual Members ($40), noting there were reduced-fee memberships for students, K-12 teachers, and members from countries with low and middle income economies. The Founding Member level was intended to help provide that critical extra support in the early phase of the Society, and has been very successful!

The membership webpage went live right at the end of 2020, really with the intention that membership would be open on 1 January 2021. But a couple of keen-eyed dipterists who saw the webpage right when it went live registered on 31 December 2020! These were Peter Cranston and James Kennedy, both of whom joined as Founding Members. Our current membership is 173, which includes all members, even those not up-to-date with payment. The trajectory of our membership is seen in Figure 1.

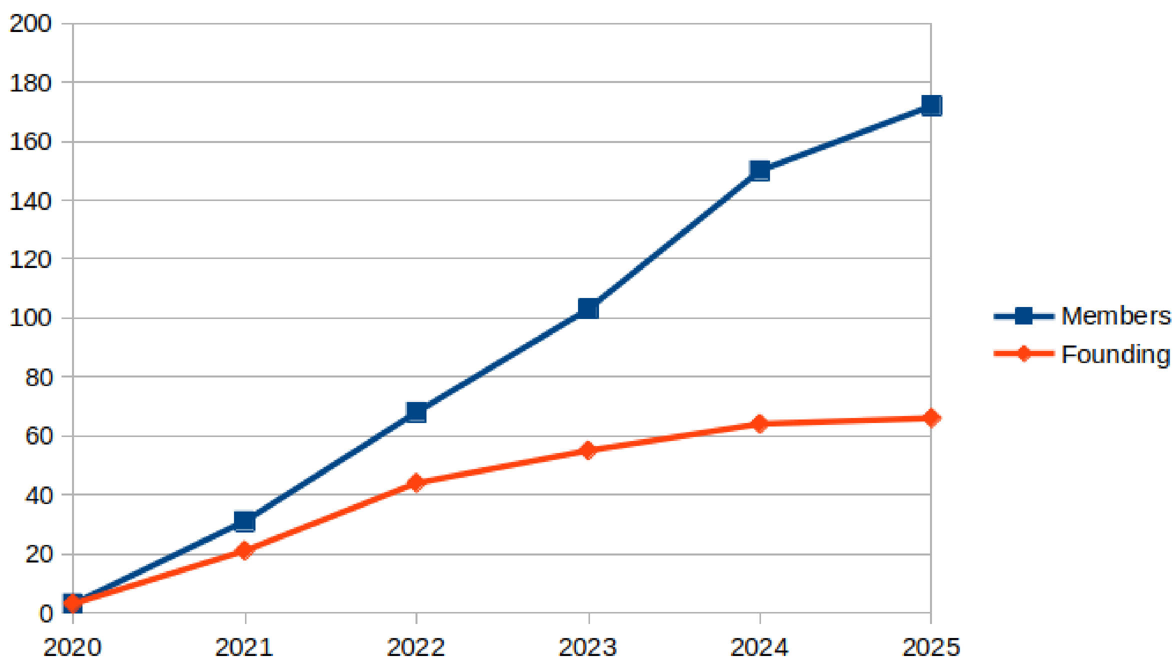

Figure 1. Trajectory of the growth in total membership, and Founding Memberships starting with the first three in 2020.

Although growth has been slow to date (there are a lot more than 173 dipterists out there!), it has been steady year-to-year. **I want to thank all of you who have joined**, and we look forward to growing our membership even more in 2026! It would be great to see a big spike in the Figure 1 graph this coming year! It is worth noting that we have had many non-North American members since the start (we have members from 27 countries!), but I think the low numbers in the first few years have, at least in part, been due to our society name implying a regional emphasis for North American dipterists – something never intended – and which we changed this past year to affirm our commitment to the entire dipterological community. Another issue, again at least in part, may have been that all our resources have been freely available to the entire community. That is, no pay walls, Society publications are open access, the Dipterists Directory and dipterists mailing list are open, etc. – all resources have been for everyone. As a community of dipterists, we can make this society exactly what we want and need, so long as we are in it together and all become part of the effort!

Important announcements regarding membership.

1) Founding Member category coming to a close on 31 June 2026.

The Founding Membership program has been running now for nearly 5 years, an appropriate time period for such a purpose. To date, we have a total of 66 Founding Members (Table 1), more than 1/3 of our total membership, whose early support for the Society has been critical towards our success!

The Dipterists Society heartily thanks each and every Founding Member for their help through these early days! There is still time to join the ranks of Founders, for the next six months or so!

Table 1. Our 66 Founding Members to date.
* = those who joined as Founding Members in the first year.
** = those who continued to pay at the full Founding Member rate over multiple years.

Matthew Bertone	Emily Hartop	C. Riley Nelson
Vladimir Blagoderov	Martin Hauser*, **	Allen Norrbom*
Art Borkent	Barbara Hayford**	James O'Hara
Christopher Borkent*	James Hogue	Thomas Pape**
Will Bouchard	Lawrence Hribar	Adrian Pont*, **
Brian Brown	Michael Irwin**	Erick Rodriguez
Stephen Bullington**	Morgan Jackson**	Paul Rude**
Chris Cohen	Nikolas Johnston	Bjoern Rulik*, **
Paul Cooney	Rasmus Keis Neerbek**	Justin Runyon
Peter Cranston*, **	James Kennedy*, **	Casey Rush*
Jeffrey Cumming	Robert Kimsey**	Ken Schneider**
Gregory Dahlem	Ashley Kirk-Spriggs	Gary Steck*, **
Torsten Dikow*	Giar-Ann Kung	John Stireman
Kathleen Donham*, **	Edward Lisowski	James Wallman**
Michael Engel*, **	Stephen Marshall	Heather Ward
Neal Evenhuis**	Jorge Luis Mederos López	Lauren Weidner
Raymond Gagné**	Ximo Mengual	Andrew Whittington
Stephen Gaimari*, **	John Midgley	Terry Whitworth**
Jon Gelhaus*	Julia Mlynarek*	Brittany Wingert
Jessica Gillung	Kevin Moulton**	Norman Woodley
David Grimaldi	Leonard Munstermann	David Yeates*
Krystal Hans	William Murphy	Andrew Young

Founding Membership is “pay it once and you are always a Founding Member” (i.e., it is fine to pay at the regular member rate after that first year), but it is noteworthy that over 1/3 of our Founding Members have continued to pay at the original rate ($150) over multiple years, and this is in addition to the many who have also added significant donations over the years! This generosity from our members is greatly appreciated! As I always say in my acknowledgment letter to new members, “Our society relies completely on memberships and donations, so every member and every supporter is important!” *As we make this change, all Founding Members remain Founding Members!* The external indicator for Founding Members, besides being recorded in the annals of the *Fly Times*, the Dipterists Directory, and in the minutes of our board meetings, is in their membership numbers starting with FM (individual members start with IM).

As we are at about the 5-year mark, to retain and emphasize the real meaning of the program to recognize for all time those individuals who offered their early extra support for the Society, the Board of Directors has decided to formally cease new memberships to this category, effective 31 June 2026.

2) Sustaining Member, a new membership category opening on 1 July 2026.
As we recognize and appreciate those members who want to contribute more to the Society, we will be opening a new category, the Sustaining Member. Similar to the Founding Member program, the Sustaining Member rate will start at $150, which will be formally recognized by the Society, and will have special membership numbers, i.e., starting with SM (note, Founding Members can also be Sustaining Members, although they always remain Founding Members, with the their membership number starting FM). However, one difference is that to retain Sustaining Membership from year-to-year, the contribution must be at that level – hence “Sustaining”! Details of our new Sustaining Member category, including tiers and incentives, will be given in the next issue of *Fly Times*!

3) Only members of the Dipterists Society are eligible to apply for our Grants Program.
The program has been very popular up to this point, having handled 75 applications, and awarded 36 research and travel grants so far to dipterists representing 17 countries. In total, we have awarded more than $30,000 in grants since our first grant competition in 2022 (which awarded a total of $1000). Moving forward, only Dipterists Society members in good standing (dues up to date) are eligible to apply for Society grants. (See the call for grant applications later in this issue of *Fly Times*!)

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The larval habitats of the Muscidae (Diptera)

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In 1964 the late Willi Hennig concluded his monumental revision of the Palaearctic Muscidae with a table of larval habitats (*Die Fliegen der palaearktischen Region*, 64b: 1083–1085). Starting in the same year, I used Hennig's framework to include muscid larval habitats from all biogeographic regions of the world and continued updating it over the following decades. It now seems timely to publish the results which, it is believed, will be of interest not just to muscid specialists but also more widely in dipterology, to larval taxonomists, systematists and phylogeneticists. As this list was compiled piecemeal over the decades, sometimes from published literature and new publications as they appeared and sometimes from Museum and other collections I studied, no bibliography was prepared and so this list must of necessity appear without references.

What is clear from this compilation is the sheer variety of substrates exploited by muscid larvae. Whether the larvae are phytophages, coprophages, saprophages or predators, they have been able to exploit almost every available ecological niche with success. They thrive from the seashore to savannah, from commercial livestock to agricultural landscapes, from rain forests to montane forests, from Arctic and Antarctic tundra to the nival zone of mountains, from sea level to even the highest mountains.

Larval habitats of muscid larvae

1 In sap runs or rot holes in trees.

Graphomya kovaci Pont
Graphomya maculata Scopoli
Helina evecta Harris
Helina pertusa Meigen
Mydaea ancilla Meigen
Phaonia amicula Villeneuve
Phaonia canescens Stein
Phaonia cincta Zetterstedt
Phaonia czernyi Hennig
Phaonia exoleta Meigen
Phaonia fugax Tiensuu
Phaonia gobertii Mik (including *Cossus* borings)
Phaonia gracilis Stein (including *Cossus* borings)
Phaonia laeta Fallén (including *Cossus* oak)
Phaonia mystica Meigen
Phaonia ninae Sorokina
Phaonia palpata Stein
Phaonia pratensis Robineau-Desvoidy
Phaonia rufiventris Scopoli
Phaonia subventa Harris
Phaonia trimaculata Bouché

Phaonia valida Harris
Potamia littoralis Robineau-Desvoidy
Potamia scabra Giglio-Tos
Potamia setifemur Stein
Thricops diaphanus Wiedemann

2 In rotten wood or beneath moss or bark on dead or dying trees.

Coenosia agromyzina Fallén
Coenosia emiliae Lukasheva
Coenosia means Meigen
Coenosia mollicula Fallén
Coenosia testacea Robineau-Desvoidy
Dichaetomyia parimpar Pont
Hebecnema umbratica Meigen
Helina abdominalis Zetterstedt
Helina consimilis Fallén
Helina depuncta Fallén
Helina evecta Harris
Helina impuncta Fallén
Helina pertusa Meigen
Helina reversio Harris
Helina subvittata Séguy
Helina vicina Czerny
Hydrotaea sp. near *ringdahli* Stein
Lispocephala biseta Grimshaw
Lispocephala bispina Malloch
Lispocephala difficilis Hardy
Lispocephala hamifera Hardy
Lispocephala hirtifemur Malloch
Lispocephala ingens Grimshaw
Lispocephala latitarsis Hardy
Lispocephala montgomeryi Hardy
Lispocephala paloloae Malloch
Lispocephala pauciseta Hardy
Lispocephala planifemorata Hardy
Lispocephala villosifemora Hardy
Muscina stabulans Fallén
Mydaea ancilla Meigen
Mydaea humeralis Robineau-Desvoidy
Neodexiopsis basalis Stein
Phaonia amabilis Meigen
Phaonia amicula Villeneuve
Phaonia angelicae Scopoli
Phaonia atkinsoni Emden
Phaonia atrocyanea Ringdahl
Phaonia babarabica Sorokina
Phaonia canescens Stein

Phaonia cincta Zetterstedt
Phaonia consobrina Zetterstedt
Phaonia czernyi Hennig
Phaonia errans Meigen
Phaonia erronea Schnabl
Phaonia exoleta Meigen
Phaonia fugax Tiensuu
Phaonia gobertii Mik
Phaonia gracilis Stein
Phaonia grandaeva Zetterstedt
Phaonia harti Malloch
Phaonia impura Zinoviev
Phaonia ishizuchiensis Shinonaga & Kano
Phaonia kowarzii Schnabl
Phaonia laeta Fallén
Phaonia mystica Meigen
Phaonia pallida Fabricius
Phaonia palpata Stein
Phaonia picealis Huckett
Phaonia pratensis Robineau-Desvoidy
Phaonia profugax Pandellé
Phaonia pudoa Hall
Phaonia rufiventris Scopoli
Phaonia serva Meigen
Phaonia subfuscinervis Zetterstedt
Phaonia subventa Harris
Phaonia taigensis Zinoviev
Phaonia tiefii Schnabl
Phaonia trimaculata Bouché
Phaonia valida Harris
Phaonia wahlbergi Ringdahl
Potamia littoralis Robineau-Desvoidy
Potamia scabra Giglio-Tos
Potamia setifemur Stein
Spilogona contractifrons Zetterstedt
Thricops diaphanus Wiedemann
Thricops ?*nigrifrons* Robineau-Desvoidy
Thricops semicinereus Wiedemann

3 In fungi on trees or on the ground.

Arthurella nudiseta Albuquerque	Agaricaceae
Gymnodia delecta Wulp	*Pleurotus ostreatus*
Helina pertusa Meigen	*Inonotus hispidus*
Helina reversio Harris	“puffball”
Hydrotaea armipes Fallén	*Armillaria* sp.
	Boletus chrysenteron
Hydrotaea capensis Wiedemann	*Termitomyces* sp.

Hydrotaea dentipes Fabricius	
Lispocephala ingens Grimshaw	
Muscina angustifrons Loew	“fungus”
Muscina dorsilinea Wulp	*Boletus* sp.
Muscina levida Fallén	*Agaricus abruptibulbus*
	Agaricus arvensis
	Agaricus bernardii
	Agaricus bitorquis
	Agaricus bresadolanus
	Agaricus campestris
	Agaricus maskae
	Agaricus silvicolus
	Agaricus xanthodermus
	Amanita caesarea
	Amanita rubescens
	Amanita vaginata
	Amanitopsis virginata
	Armillaria mellea
	Boletus aestivalis
	Boletus americanus
	Boletus badius
	Boletus edulis
	Boletus griseus
	Boletus luridus
	Boletus queletii
	Boletus regius
	Boletus reticulatus
	Boletus rhodoxanthus
	Boletus sp.
	Calocybe gambosa
	Coprinus fuscescens
	Hypholoma fasciculare
	Inocybe jurana
	Lactarius acerrimus
	Lactarius serifluus
	Leccinum carpini
	Leccinum griseum
	Lepiota acutesquamosa
	Leucopaxillus tricolor
	Macrolepiota procera
	Macrolepiota rhacodes
	Marasmius oreades
	Morchella esculenta
	Pluteus petasatus
	Polyporus squamosus
	Psalliota campestris
	Psathyrella candolleana
	Ptychoverpa bohemica

	Russula amoenicolor
	Russula basifurcata
	Russula cyanoxantha
	Russula depallens
	Russula foetens
	Russula furcata
	Russula grisea
	Russula melliolens
	Russula ochroleuca
	Russula rosacea
	Russula rosea
	Russula vesca
	Russula virescens
	Russula sp.
	Scleroderma citrinum
	Suillus piperatus
	Xerocomus rubellus
	Xerocomus subtomentosus
Muscina minor Porchinskiy	
Muscina pascuorum Meigen	*Amanita citrina*
Muscina prolapsa Harris	
Muscina stabulans Fallén	*Agaricus aurantiacus*
	Agaricus bernardii
	Agaricus bitorquis
	Amanita aspera
	Amanita sp.
	Boletus edulis
	Boletus luridus
	Boletus nigrescens
	Boletus sp.
	Calocybe gambosa
	Inocybe sp.
	Lactarius sp.
	Leccinium griseum
	Leccinum sp.
	Lepista cristata
	Pleurotus cornucopiae
	Pleurotus sp.
	Polyporus squamosus
	Polyporus umbellatus
	Polyporus varius
	Psalliota arvensis
	Russula emetica
	Russula rosea
	Russula sp.
	Suillus granulatus
Mydaea affinis Meade	*Amanita rubescens*
	Boletus bovinus

	Boletus radicans
	Boletus sp.
	Gomphidius glutinosus
	Lactarius resimus
	Leccinum scabrum
	Leccinum versipelle-group
	Piptoporus betulinus
	Suillus granulatus
	Suillus grevillei
	Suillus luteus
	Suillus variegatus
Mydaea ancilla Meigen	*Laetiporus sulphureus*
Mydaea corni Scopoli	*Armillaria mellea*
	Lactarius deterrimus
	Russula luteotacta
	Russula nigricans
Mydaea detrita Zetterstedt (*electa* auctt.)	*Boletus erythropus*
	Boletus queleti
	Boletus variegatus
	Chroogomphus rutilus
	Lactarius deliciosus-group
	Lactarius deterrimus
	Lactarius torminosus
	Leccinum scabrum
	Leucopaxillus tricolor
	Macrolepiota procera
	Meripilus giganteus
	Phallus impudicus
	Russula densifolia
	Russula foetens
	Russula integra
	Russula luteotacta
	Russula nigricans
	Suillus collinitus
	Suillus granulatus
	Suillus luteus
	Suillus variegatus
Mydaea flavicornis Coquillett	
Mydaea flavitarsis Lobanov	*Pleurotus ostreatus*
	Pleurotus sp.
	Polyporus sp.
Mydaea humeralis Robineau-Desvoidy	*Agaricus augustus*
	Amanita caesarea f. *alba*
	Amanita citrina
	Amanita fulva
	Amanita pantherina
	Amanita rubescens
	Amanita vaginata

Amanita umbrina
Amanita sp.
Armillaria mellea
Armillaria tabescens
Boletus edulis
Boletus erythropus
Boletus impolitus
Boletus luridus
Boletus pinicola
Chamaemyces fracidus
Clitocybe gibba
Collybia fusipes
Collybia sp.
Cortinarius bulliardii
Cortinarius glaucopus var. *olivaceus*
Cortinarius mucifluoides
Cortinarius mucosus
Cortinarius nemorensis
Cortinarius sciophyllus
Cortinarius trivialis
Eutoloma rhodopolius
Eutoloma sinuatus
Fomes hispidus
Hebeloma crustuliniforme
Hygrophorus dichrous
Hygrophorus eburneus
Hypholoma fasciculare
Inocybe aeruginascens
Inocybe agardhii
Inocybe fastigiata
Inocybe jurana
Lactarius acerrimus
Lactarius azonites
Lactarius circellatus
Lactarius chrysorrheus
Lactarius deliciosus
Lactarius forminosus
Lactarius insulsus
Lactarius rufus
Lactarius semisanguifluus
Lactarius subdulcis
Lactarius torminosus
Lactarius vellereus
Lactarius vietus
Lactarius sp.
Leccinum scabrum-group
Lentinus cyathiformis
Lepiota cristata

Leucopaxillus paradoxus
Lyophyllum loricatum
Marasmius oreades
Megacollybia platyphylla
Melanoleuca grammopodia
Morchella sp.
Omphalotus olearius
Oudemansiella platyphylla
Paxillus involutus
Pleurotus cornucopiae
Pluteus cervinus
Psathyrella hydrophila
Psathyrella sp.
Pseudopyrochroa sp.
Rozites caperata
Russula aeruginea
Russula alutacea
Russula atropurpurea
Russula aurata
Russula basifurcata
Russula curtipes
Russula cyanoxantha
Russula decolorans
Russula delica
Russula densifolia
Russula foetens
Russula fragilis
Russula furcata
Russula grisea f. *typica*
Russula grisea var. *pinicola*
Russula integra
Russula lutea f. *luteorosella*
Russula luteotacta
Russula maculata
Russula nigricans
Russula olivacea
Russula ochroleuca
Russula pectinata
Russula pectinatoides
Russula romellii f. *rubeola*
Russula rosacea
Russula rosea
Russula sanguinea
Russula subcompacta
Russula vesca
Russula veternosa
Russula vinosa
Russula vinosopurpurea

	Russula virescens
	Russula xerampelina
	Russula sp.
	Suillus collinitus
	Suillus granulatus
	Suillus grevillei
	Suillus luteus
	Suillus piperatus
	Suillus variegatus
	Tricholoma equestre
	Tricholoma populinum
	Tricholoma scalpturatum
	Tricholoma terreum
	Xerocomus chrysenteron
	Xerocomus rubellus
	Xerocomus subtomentosus
	Xerocomus sp.
Mydaea maculiventris Zetterstedt	*Polyporus squamosus*
Mydaea montana Lobanov	
Mydaea ?*neobscura* Snyder	*Pleurotus ostreatus*
Mydaea nubila Stein	*Pleurotus pulmonarius*
Mydaea orthonevra Macquart	*Amanita citrina*
	Amanita vaginata-group
	Boletus piperatus
	Lactarius trivialis
	Suillus granulatus
	Suillus grevillei
	Suillus luteus
Mydaea setifemur Ringdahl	*Amanita muscaria*
	Amanita regalis
	Amanita rubescens
	Collybia sp.
	Lactarius deliciosus-group
	Lactarius helvus
	Lactarius necator
	Lactarius torminosus
	Lactarius trivialis
	Paxillus involutus
	Pluteus cervinus
	Russula aeruginea
	Russula decolorans
	Russula lundelli
	Russula vesca
	Suillus granulatus
Mydaea urbana Meigen	*Fomes hispidus*
	Polyporus squamosus
Neodexiopsis floridensis Malloch	*Pleurotus ostreatus*
Neomuscina capalta Snyder	

Neomuscina pictipennis Bigot	*Fomes* sp.
Phaonia bitincta Rondani	*Ramaria* sp.
Phaonia boleticola Rondani	*Boletus luridus*
Phaonia bysia Walker	*Hygrophorus* sp.
Phaonia canescens Stein	
Phaonia gobertii Mik	*Polyporus hispidus*
Phaonia latipalpis Schnabl	
Phaonia pallida Fabricius	*Amanita rubescens*
	Clitocybe inversa
	Clitocybe nebularis
	Xerula radicata
Phaonia rufiventris Scopoli	*Boletus edulis*
	Cantharellus cibarius
	Cortinarius privignoides
	Cortinarius sciophyllus
	Cortinarius torvus
	Craterellus cornucopioides
	Lactarius semisanguifluus
	Lactarius vellereus
	Lyophyllum decastes
	Merulius tremellosus
	Polystictus sp.
	Russula cyanoxantha
	Russula delica
	Suillus luteus
	Tricholoma ustale
	Tricholomopsis rutilans
Phaonia subventa Harris	*Agrocybe cylindrica*
	Armillaria gallica
	Armillaria mellea
	Armillaria sp.
	Boletus edulis
	Clitocybe nebularis
	Gymnopilus junonius
	Hypholoma fasciculare
	Lactarius quietus
	Paxillus involutus
	Phallus impudicus
	Pleurotus cornucopiae
	Polyporus betulinus
	Ramaria formosa
	Russula delica
	Russula sp.
	Stereum rugosum
	Tricholoma pessundatum
Phaonia trimaculata Bouché	*Inonotus cuticularis*
Potamia littoralis Robineau-Desvoidy	*Phallus impudicus*
	Lactarius sp.

Prohardyia fungorum Pont
Prohardyia intermedia Pont
Prohardyia pollinosa Malloch
Stomoxys calcitrans Linnaeus
Thricops diaphanus Wiedemann

Amanita muscaria
Armillaria mellea
Boletus edulis
Boletus luridus
Cortinarius glaucopus
Cortinarius hinnuleus
Cortinarius trivialis
Hebeloma crustuliniforme
Lactarius sanguifluus
Lactarius trivialis
Pholiota squarrosa
Russula cyanoxantha
Suillus luteus
Tricholoma saponaceum
Xerocomus chrysenteron

4 In plants.

4a In rotting plant material, leaf-litter or compost. Also in living plants already attacked by other invertebrates.

Atherigona distincta Malloch
Atherigona hendersoni Malloch
Atherigona integrifemur Emden
Atherigona longipalpis Malloch
Atherigona maculigera Stein
Atherigona orientalis Schiner
Atherigona pallidipalpis Malloch
Atherigona poecilopoda Bezzi
Atherigona seticauda Malloch
Atherigona tau Pont
Atherigona transversa Deeming
Atherigona yorki Deeming
Auria niveipalpis Stein
Bithoracochaeta sociabilis Blanchard
Gymnodia delecta Wulp
Buccophaonia subcostalis Emden
Coenosia agromyzina Fallén
Coenosia attenuata Stein
Coenosia dubiosa Hennig
Coenosia rufipalpis Meigen
Coenosia sexmaculata Meigen
Coenosia strigipes Stein
Coenosia testacea Robineau-Desvoidy

Coenosia tigrina Fabricius
Drymeia tetra Meigen
Eudasyphora cyanicolor Zetterstedt
Graphomya maculata Scopoli
Hebecnema umbratica Meigen
Hebecnema vespertina Fallén
Helina australasiae Malloch
Helina clara Meigen
Helina confinis Fallén
Helina coniformis Stein
Helina impuncta Fallén
Helina lucida Stein
Helina pubiseta Zetterstedt
Helina regina Malloch
Helina serrulata Thomson
Hydrotaea armipes Fallén
Hydrotaea capensis Wiedemann
Hydrotaea chalcogaster Wiedemann
Hydrotaea dentipes Fabricius
Hydrotaea ignava Harris
Hydrotaea irritans Fallén
Hydrotaea meteorica Linnaeus
Limnophora exigua Wiedeman
Lispocephala biseta Grimshaw
Lispocephala bispina Malloch
Lispocephala brachydexioides Hardy
Lispocephala flaccida Hardy
Lispocephala hirtifemur Malloch
Lispocephala lanaiensis Hardy
Lispocephala latitarsis Hardy
Lispocephala longipes Grimshaw
Lispocephala mauiensis Hardy
Lispocephala pallidibasis Malloch
Lispocephala zonata Hardy
Macrorchis meditata Fallén
Musca autumnalis De Geer
Musca domestica Linnaeus
Musca hervei Villeneuve
Musca sorbens Wiedemann
Muscina angustifrons Loew
Muscina levida Harris
Muscina pascuorum Meigen
Muscina prolapsa Harris
Muscina stabulans Fallén
Myospila argentata Walker
Myospila flavicans Malloch
Myospila lenticeps Thomson
Myospila prosternalis Emden

Phaonia angelicae Scopoli
Phaonia annulipes Stein
Phaonia atriceps Loew
Phaonia canescens Stein
Phaonia consobrina Zetterstedt
Phaonia errans Meigen
Phaonia exoleta Meigen
Phaonia femorata Stein
Phaonia gallicola Albuquerque
Phaonia gobertii Mik
Phaonia jaroschewskii Schnabl
Phaonia limbinervis Stein
Phaonia parviceps Malloch
Phaonia rhodesi Malloch
Phaonia rufiventris Scopoli
Phaonia subventa Harris
Phaonia trimaculata Bouché
Phaonia tuguriorum Scopoli
Phaonia valida Harris
Phaonia varians Bigot
Phaonia vittithorax Stein
Phaonina corbetti Malloch
Potamia littoralis Robineau-Desvoidy
Pseudohelina castanea Curran
Pseudohelina nigritarsis Jaennicke
Pseudohelina rufina Stein
Pyrellia rapax Harris
Pyrellina distincta Walker
Pyrellina ?*inventrix* Walker
Pyrellina rhodesi Malloch
Scenetes cardini Malloch
Stomoxys calcitrans Linnaeus
Stomoxys niger Macquart
Stomoxys sitiens Rondani
Synthesiomyia nudiseta Wulp
Thricops genarum Zetterstedt

4b In living plant tissue (phytophagous).

In *Nepenthes* pitchers	*Phaonia mogii* Shinonaga & Kurahashi
	Phaonia nepenthincola Stein
In *Nipa* inflorescence	*Phaonina corbetti* Malloch
Galls on *Pteridium aquilinum*	*Phaonia gallicola* Albuquerque
Campomanesia guaviroba	*Dolichophaonia cananeiensis* Gomes & Carvalho
In Poaceae	*Atherigona* species

Known hosts for *Atherigona* species

Andropogon gayanus var. *bisquamulatus* (Hochst.) Hackel	sp. 2 near *ancora* Deeming
Arundinella metzii Hochst. ex Miq.	*naqvii* Steyskal
Avena sterilis ssp. *ludoviciana* (Durien) Gill and Magne	*naqvii* Steyskal
Bothriochlora pertusa (L.) A.Camus	*bidens* Hennig
Brachiaria brizantha (Hochst. ex A.Rich.) Stapf	*soccata* Rondani
Brachiaria distachya (L.) Stapf	*oryzae* Malloch
	pulla Wiedemann
	punctata Karl
	reddyi Pont
	reversura Villeneuve
	soccata Rondani
Brachiaria eruciformis (Smith) Griseb. (syn: *Panicum isachne* Roth ex Roemer and Schultes)	*punctata* Karl
	sp.
Brachiaria mutica (Forsskal) Stapf	*punctata* Karl
Brachiaria ramosa (L.) Stapf	*bella* Frey
	falcata Thomson
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
Brachiaria reptans (L.) C.Gardner and C.E.Hubb.	*eriochloae* Malloch
	falcata Thomson
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
	soccata Rondani
	sp.VIII (Davies and Reddy, 1981)
	sp.
Brachiaria xantholeuca (Schinz) Stapf	*albistyla* Deeming
Chloris barbata Sw.	*falcata* Thomson
Chloris gayana Kunth	*hyalinipennis* Emden
Cymbopogon caesius (Nees ex Hook. and Arn.) Stapf	*oryzae* Malloch
	soccata Rondani
Cymbopogon citratus (DC.) Stapf	?*soccata* Rondani
Cynodon dactylon (L.) Pers.	*approximata* Malloch
	eriochloae Malloch
	falcata Thomson
	humeralis Wiedemann
	laeta Wiedemann
	laevigata Loew

	oryzae Malloch
	reversura Villeneuve
	soccata Rondani
	zeta Pont
	sp.VI (Davies and Reddy, 1981)
Cynodon nlemfuensis	*conigera* Emden
	tetrastigma Paterson
Cynodon sp.	*reversura* Villeneuve
Dactyloctenium aegyptium (L.) Willd.	*atripalpis* Malloch
	soccata Rondani
Danthonia decumbens (L.)	*varia* Meigen
Desmostachya bipinnata (L.) Stapf (syn: *Eragrostis cynosuroides* (Retz.) P.Beauv.)	*soccata* Rondani
Dichanthium annulatum (Forsskal) Stapf	*falcata* Thomson
	punctata Karl
Dichanthium caricosum (L.) A.Camus	sp.
Digitaria ciliaris (Retz.) Koeler (syn: *adscendens* (HBK) Henr.)	*falcata* Thomson
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
	soccata Rondani
	sp.
Digitaria decumbens Stent	*oryzae* Malloch
Digitaria longiflora (Retz.) Pers.	*bella* Frey
	oryzae Malloch
Digitaria sanguinalis (L.) Scop.	*naqvii* Steyskal
	soccata Rondani
Digitaria scalarum (Schweinf.) Chiov.	*soccata* Rondani
Echinochloa colona (L.) Link.	*approximata* Malloch
	atripalpis Malloch
	eriochloae Malloch
	falcata Thomson
	orientalis Schiner
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
	reversura Villeneuve
	simplex Thomson
	soccata Rondani
Echinochloa crusgalli (L.) P.Beauv.	*eriochloae* Malloch
	falcata Thomson
	pulla Wiedemann
	soccata Rondani
Echinochloa frumentacea Link.	*falcata* Thomson
	?*pulla* Wiedemann
Echinochloa stagnina (Retz.) P.Beauv.	*falcata* Thomson

Echinochloa utilis Ohwi and Yabuno	*falcata* Thomson
Eleusine coracana (L.) Gaertner	*gilvifolia* Emden
	miliaceae Malloch
	naqvii Steyskal
	reversura Villeneuve
	soccata Rondani
Eleusine indica (L.) Gaertner	*gilvifolia* Emden
	soccata Rondani
	sp.
Eleusine sp.	*soccata* Rondani
Eragrostis cilianensis (All.) Vign. ex Janchen	*punctata* Karl
Eragrostis interrupta P.Beauv.	sp.
Eragrostis japonica (Thunb.) Trin.	*atripalpis* Malloch
	falcata Thomson
	orientalis Schiner
	oryzae Malloch
	soccata Rondani
Eragrostis tef (Zucc) Trotter	*hyalinipennis* Emden
	longifolia Emden
	ugandae Emden
Eriochloa anulata ...	*soccata* Rondani
Eriochloa procera (Retz.) C.E.Hubb.	*eriochloae* Malloch
	falcata Thomson
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
	reversura Villeneuve
	simplex Thomson
	soccata Rondani
Eriochloa sp.	?*pulla* Wiedemann
	?*simplex* Thomson
AFRICA *Gossypium herbaceum* [not a grass]	*bedfordi* Emden
Hemarthria compressa (L.f.) R.Br.	?*oryzae* Malloch
Heteropogon contortus (L.) P.Beauv. ex Roemer and Schultes	*oryzae* Malloch
Hordeum vulgare Linn.	*naqvii* Steyskal
Hyparrhenia cyanescens (Stapf) Stapf	*bedfordi* Emden
	mitrata Séguy
	yorki Deeming
Hyperthelia dissoluta (Stend.) W.D.Clayton	sp. 2 near *ancora* Deeming
Imperata cylindrica (L.) Raersch	*hennigi* Pont
Isachne globosa (Thunb.) Kuntze	*delta* Pont
?*Isachne* sp.	?*miliaceae* Malloch
	?*simplex* Thomson
Ischaemum afrum (Gmelin) Dandy (syn: *pilosum* (Willd.) Wight)	*falcata* Thomson
Miscanthus sinensis Andersson	*boninensis* Snyder

Oryza barthii A.Chev.	*bedfordi* Emden
Oryza minuta Presl	sp.
Oryza sativa L.	*mitrata* Séguy
	?*orientalis* Schiner
	oryzae Malloch
	iota Pont
Panicum antidotale Retz. (syn: *miliare* Lam.)	*falcata* Thomson
	miliaceae Malloch
	pulla Wiedemann
	soccata Rondani
Panicum maximum Jacq.	*matilei* Deeming
	soccata Rondani
Panicum miliaceum L.	*miliaceae* Malloch
	pulla Wiedemann
	?*punctata* Karl
	soccata Rondani
	sp. (Australia)
Panicum psilopodium Trin.	*atripalpis* Malloch
	bidens Hennig
	eriochloae Malloch
	falcata Thomson
	oryzae Malloch
	pulla Wiedemann
	reversura Villeneuve
Panicum repens L.	*eriochloae* Malloch
	falcata Thomson
	?*oryzae* Malloch
	pulla Wiedemann
	?*punctata* Karl
	reversura Villeneuve
	soccata Rondani
Panicum sumatrense Roth ex Roem. & Schult.	*pulla* Wiedemann
Paspalidum flavidum (Retz.) A.Camus	*punctata* Karl
Paspalum scrobiculatum L. (syn: *orbiculare*)	*lineata* Adams
	oryzae Malloch
	pulla Wiedemann
	simplex Thomson
	soccata Rondani
Paspalum sp.	?*oryzae* Malloch
	soccata Rondani
?*Paspalum* sp.	*bedfordi* Emden
Pennisetum glaucum (L.) R.Br. (syn: *americanum* (L.) Leeke) (syn: *typhoides* (Burm.) Stapf and C.E.Hubb.)	*approximata* Malloch

	naqvii Steyskal
	oryzae Malloch
	ponti Deeming
	punctata Karl
	soccata Rondani
	yorki Deeming
Pennisetum pedicellatum Trin.	*mitrata* Séguy
	steeleae Emden
Pennisetum purpureum Schum.	*bedfordi* Emden
	mitrata Séguy
	steeleae Emden
	yorki Deeming
	sp. nov. near *soccata* Rondani
Pennisetum sp.	*approximata* Malloch
	?*soccata* Rondani
Poa abyssinica Jacq.	*hyalinipennis* Emden
Rhytachne rottboellioides Desv.	*longifolia* Emden
	sp. 2 near *ancora* Deeming
Rottboellia cochinchinensis (Lour.) W.D.Clayton (syn: *exaltata*, nom. illegit.)	*marginifolia* Emden
	soccata Rondani
Saccharum officinarum L.	*acutipennis* Villeneuve
	boninensis Snyder
	ramu Pont
	sp. (Australia)
Sehima nervosum (Rottler) Stapf	*reversura* Villeneuve
Setaria faberii Herrm.	*biseta* Karl
Setaria geniculata	sp.
Setaria intermedia Roemer and Schultes	*atripalpis* Malloch
	oryzae Malloch
	soccata Rondani
Setaria italica (L.) P.Beauv.	*approximata* Malloch
	atripalpis Malloch
	biseta Karl
	pulla Wiedemann
	punctata Karl
	sp.
Setaria pallidefusca	*hyalinipennis* Emden
Setaria plicata (Lam.) T.Cooke	*kappa* Pont
Setaria pumila (Poiret) Roemer and Schultes (syn: *glauca* (Linn.) P.Beauv.) (syn: *pallidefusca* (Schum.) Stapf and C.E.Hubb.)	*atripalpis* Malloch
	biseta Karl
	falcata Thomson
	hyalinipennis Emden
	lineata Adams

	marginifolia Emden
	occidentalis Deeming
	oryzae Malloch
	pulla Wiedemann
	punctata Karl
	secrecauda Séguy
	soccata Rondani
Setaria verticillata (L.) P.Beauv.	*soccata* Rondani
Setaria viridis (L.) P.Beauv.	*biseta* Karl
Setaria sp.	?*miliaceae* Malloch
Sorghum arundinaceum (Desv.) Stapf	
(syn: *verticilliflorum*)	
(syn: *virgatum*)	*soccata* Rondani
Sorghum bicolor (L.) Moench	
(syn: *almum* Parodi)	
(syn: *dochna* (Forsskal) Snowden)	
(syn: *saccharatum* Moench)	
(syn: *fastigiatum* (Sw.) Kuntze)	
(syn: *macrospermum* Garber)	
(syn: *vulgare* Pers.)	*approximata* Malloch
	atripalpis Malloch
	bedfordi Emden
	bidens Hennig
	campestris Deeming
	eriochloae Malloch
	falcata Thomson
	gilvifolia Emden
	hyalinipennis Emden
	laevigata Loew
	lineata Adams
	naqvii Steyskal
	occidentalis Deeming
	orientalis Schiner
	oryzae Malloch
	ponti Deeming
	pulla Wiedemann
	punctata Karl
	reversura Villeneuve
	secrecauda Séguy
	simplex Thomson
	soccata Rondani
	steeleae Emden
	tomentigera Emden
	ugandae Emden
	yorki Deeming
Sorghum x *drummondii* (Nees ex Steudel) Millsp. and Chase	
(syn: *sudanense*)	*soccata* Rondani

Sorghum halepense (L.) Pers.	*oryzae* Malloch
	soccata Rondani
Sorghum propinquum (Kunth) Hitchc.	*soccata* Rondani
Sorghum purpureosericeum (A.Rich.) Aschers.	*soccata* Rondani
Sorghum sp.	*hyalinipennis* Emden
Sporobolus pyramidalis P.Beauv.	*bedfordi* Emden
Thelepogon elegans Roemer and Schultes	*soccata* Rondani
Themeda quadrivalvis (L.) Kuntze	*eriochloae* Malloch
	falcata Thomson
	oryzae Malloch
Trifolium alexandrinum [berseem, AFRICA]	*bedfordi* Emden
	humeralis Wiedemann (= *ferruginea* Emden)
Triticum aestivum L.	*bedfordi* Emden
	falcata Thomson
	naqvii Steyskal
	oryzae Malloch
	?*punctata* Karl
	?*simplex* Thomson
	soccata Rondani
	tritici Pont & Deeming
Zea mays L.	*acutipennis* Villeneuve
	bidens Hennig
	falcata Thomson
	naqvii Steyskal
	orientalis Schiner
	oryzae Malloch
	punctata Karl
	reversura Villeneuve
	soccata Rondani
	tau Pont
	yorki Deeming

5 In dry mosses and in soil.

5a Beneath moss on soil or rocks.

Achanthiptera rohrelliformis Robineau-Desvoidy
Helina annosa Zetterstedt
Helina consimilis Fallén
Helina evecta Harris
Helina reversio Harris
Helina vicina Czerny
Phaonia angelicae Scopoli
Phaonia jaroschewskii Schnabl
Phaonia valida Harris
Phaonia villana Robineau-Desvoidy
Phaonia zugmayeriae Schnabl

Pseudocoenosia abnormis Stein
Spilogona brunneisquama Zetterstedt
Spilogona contractifrons Zetterstedt
Spilogona marginifera Hennig
Thricops nigrifrons Robineau-Desvoidy
Thricops rostratus Meade
Thricops semicinereus Wiedemann

5b In sandy and humus soil (including woodland soil).
Some of these evidently just spend the puparial stage in the soil

Coenosia agromyzina Fallén
Coenosia bonita Huckett
Coenosia lacteipennis Zetterstedt (sand)
Coenosia minutalis Zetterstedt (sand)
Coenosia tigrina Fabricius
Drymeia cinerea Meigen
Drymeia segnis Holmgren
Helina addita Walker
Helina caerulescens Stein
Helina calceata Rondani
Helina depuncta Fallén
Helina evecta Harris
Helina impuncta Fallén
Helina pertusa Meigen
Helina protuberans Zetterstedt (sand)
Helina pubiseta Zetterstedt
Helina punctata Stein
Helina rufitibia Stein
Hydrotaea armipes Fallén
Hydrotaea dentipes Fabricius
Hydrotaea diabolus Harris
Hydrotaea irritans Fallén
Muscina prolapsa Harris
Mydaea detrita Zetterstedt
Phaonia californiensis Malloch
Phaonia consobrina Zetterstedt
Phaonia errans Meigen
Phaonia halterata Stein
Phaonia incana Wiedemann
Phaonia pallida Fabricius
Phaonia palpata Stein
Phaonia parviceps Malloch
Phaonia serva Meigen
Phaonia subventa Harris
Phaonia trimaculata Bouché
Phaonia tuguriorum Scopoli
Phaonia valida Harris

Phaonia zugmayeriae Schnabl
Thricops cunctans Meigen
Thricops nigrifrons Robineau-Desvoidy
Thricops rostratus Meade
Thricops semicinereus Wiedemann
Thricops simplex Wiedemann
Thricops ?*sudeticus* Schnabl
Villeneuvia aestuum Villeneuve (strandline)

6 In mud and water and amongst aquatic mosses.

6a In mud by rivers and coasts.

Lispe nigrimana Malloch
Lispe tentaculata De Geer

6b In muddy water.

Graphomya maculata Scopoli
Lispe pygmaea Fallén
Lispe superciliosa Loew

6c In water (amongst mosses, vascular plants, algae) or in vegetable debris alongside water-margins (lake shores, ditch sides, etc).

Agenamyia colombiana Carvalho, Wolff & Fogaça
Agenamyia exotica Carvalho & Couri
Agenamyia maculata Carvalho, Wolff & Fogaça
Agenamyia timida Carvalho, Wolff & Fogaça
Atherigona culicivora Kovac, Pont & Deeming
Coenosia lacteipennis Zetterstedt (seaweed)
Coenosia minutalis Zetterstedt
Graphomya kovaci Pont
Graphomya maculata Scopoli
Graphomya minuta Arntfield
Hydrotaea armipes Fallén
Hydrotaea dentipes Fabricius
Hydrotaea ignava Harris
Limnophora approximatinervis Stein
Limnophora discreta Stein
Limnophora exuta Kowarz
Limnophora innocua Malloch
Limnophora maculosa Meigen
Limnophora nigripes Robineau-Desvoidy
Limnophora olympiae Lyneborg
Limnophora pulchriceps Loew
Limnophora riparia Fallén
Limnophora tigrina Am Stein

Lispe caesia Meigen
Lispe consanguinea Loew
Lispe loewi Ringdahl
Lispe metatarsalis Thomson
Lispe sericipalpis Stein
Lispe tentaculata De Geer
Lispe uliginosa Fallén
Lispocephala alma Meigen
Lispocephala brachialis Rondani
Lispocephala erythrocera Robineau-Desvoidy
Lispocephala fusca Malloch
Lispocephala kaalae Williams
?*Lispocephala parydra* Hardy
Lispocephala philydra Hardy
Lispocephala vitripennis Ringdahl
Lispoides aequifrons Stein
Phaonia mogii Shinonaga & Kurahashi
Phaonia nepenthincola Stein
Schoenomyza litorella Fallén
Spilogona atricans Pandellé
Spilogona contractifrons Zetterstedt
Spilogona falleni Pont
Spilogona marina Collin
Spilogona setigera Stein
Spilogona torreyae Johannsen
Spilogona varsaviensis Schnabl
Spilogona veterrima Zetterstedt
Stomoxys calcitrans Linnaeus
Villeneuvia aestuum Villeneuve
Xenomyia oxycera Emden

7 In nests, burrows, roosts.

7a In birds' nests.

Helina australasiae Malloch
Helina balsaci Séguy
Helina pertusa Meigen
Helina pulchella Ringdahl
Helina sexmaculata Preyssler
Helina vicina Czerny
Hydrotaea armipes Fallén
Hydrotaea basdeni Collin
Hydrotaea capensis Wiedemann
Hydrotaea dentipes Fabricius
Hydrotaea ignava Harris
Hydrotaea kumagera Shinonaga & Iwasa
Hydrotaea nidicola Malloch

Hydrotaea ringdahli Stein
Musca albina Wiedemann
Musca domestica Linnaeus
Muscina levida Harris
Muscina stabulans Fallén
Mydaea urbana Meigen
Myospsila meditabunda Fabricius
Passeromyia steini Pont
Philornis aitkeni Dodge
Philornis rufoscutellaris Couri
Philornis spp. [see under myiasis]
Potamia littoralis Robineau-Desvoidy
Stomoxys calcitrans Linnaeus

7b In mammal burrows, bat roosts, etc.

Helina depuncta Fallén	mole
Helina pertusa Meigen	squirrel
Helina sexmaculata Preyssler	
Hydrotaea armipes Fallén	vole
Hydrotaea basdeni Collin	bats
Hydrotaea capensis Wiedeman	rodents, *Tachyoryctes*
Hydrotaea cyrtoneurina Zetterstedt	badger
Hydrotaea ringdahli Stein	squirrel
Lispe elegantissima Stackelberg	toad
Mydaea rufinervis Pokorny	marmot
Phaonia pallida Fabricius	
Phaonia rufiventris Scopoli	mouse
Potamia littoralis Robineau-Desvoidy	
Stomoxys calcitrans Linnaeus	

7c In nests of social Hymenoptera.

Achanthiptera rohrelliformis Robineau -Desvoidy	*Vespa germanica*, *V. vulgaris*
Drymeia sp.	bumble bees
Helina adelpha Schiner	*Bombus*
Helina reversio Meigen	hornets
Hydrotaea floccosa Macquart	bumble bees
Hydrotaea ignava Harris	bumble bees
Hydrotaea ringdahli Stein	*Lasius fuliginosus*
Lispocephala brevispina Malloch	*Hylocrabro*
Musca domestica Linnaeus	honey bees
Muscina levida Harris	
Muscina prolapsa Harris	bumble bees, *Vespa vulgaris*
Muscina stabulans Fallén	wasps, bees
Phaonia rufiventris Scopoli	wasps
Phaonia subventa Harris	wasps

Philornis vespidicola Dodge	*Paracharitopus frontalis* (Vespidae); fortuitous?
Potamia littoralis Robineau-Desvoidy	bumble bees, *Vespa crabro, Vespa maculifrons*

8 In carrion.

8a In vertebrate carrion.

Atherigona longipalpis Malloch
Atherigona orientalis Schiner
Helina allotalla Meigen
Helina coerulescens Stein
Helina pulchella Ringdahl
Helina regina Malloch
Hydrotaea acuta Stein
Hydrotaea aenescens Wiedemann
Hydrotaea armipes Fallén
Hydrotaea capensis Wiedemann
Hydrotaea chalcogaster Wiedemann
Hydrotaea dentipes Fabricius
Hydrotaea ignava Harris
Hydrotaea meteorica Linnaeus
Hydrotaea pilipes Stein
Hydrotaea rostrata Robineau-Desvoidy
Hydrotaea similis Meade
Hydrotaea solitaria Albuquerque
Hydrotaea spinigera Stein
Morellia nilotica Loew
Morellia prolectata Walker
Morellia sp.
Musca albina Wiedemann
Musca domestica Linnaeus
Musca sorbens Wiedemann
Musca terraereginae Johnston & Bancroft
Musca vetustissima Walker
Musca xanthomelas Wiedemann
Muscina angustifrons Loew
Muscina levida Harris
Muscina pascuorum Meigen
Muscina prolapsa Harris
Muscina stabulans Fallén
Neomyia nudissima Loew
Passeromyia longicornis Macquart
Phaonia subventa Harris
Potamia scabra Giglio-Tos
Stomoxys calcitrans Linnaeus
Synthesiomyia nudiseta Wulp

8b In invertebrate carrion.

Including dead and dying insect larvae and adults, orthopteroid egg-pods, from which it has been concluded that some of these species are parasitoids.

Aethiopomyia steini Curran
Alluaudinella bivittata Macquart
Alluaudinella flavicornis Macquart
Atherigona orientalis Schiner
Atherigona pallidipes Malloch
Charadrella malacophaga Lopes
Dichaetomyia pallitarsis Stein
Helina calyptrata Malloch
Helina fuscoapicata Malloch
Helina impuncta Fallén
Helina lucida Stein
Helina nemoralis Stein
Helina sexmaculata Preyssler
Hydrotaea aenescens Wiedemann
Hydrotaea armipes Fallén
Hydrotaea capensis Wiedemann
Hydrotaea chalcogaster Wiedemann
Hydrotaea ignava Harris
Hydrotaea solitaria Albuquerque
Musca autumnalis De Geer
Musca domestica Linnaeus
Muscina levida Harris
Muscina pascuorum Meigen
Muscina prolapsa Harris
Muscina stabulans Fallén
Myospila lenticeps Thomson
Myospila meditabunda Fabricius
Ochromusca trifaria Bigot
Phaonia angelicae Scopoli
Phaonia errans Meigen
Phaonia giacomeli Carvalho
Phaonia interfrontalis Emden
Phaonia meigeni Pont
Phaonia rufiventris Scopoli
Phaonia serva Meigen
Phaonia subventa Harris
Phaonia tipulivora Malloch
Phaonia trimaculata Bouché
Phaonia tuguriorum Scopoli
Phaonia valida Harris
Phaonia varians Bigot
Synthesiomyia nudiseta Wulp

9 In dung.

KEY

a	small rodents	h	horse	q	camel
aa	emu	h_2	ass	r	marmot
b	rabbit	h_3	mule	s	bat guano
bb	bison	i	cow	ss	tortoise
c	dog	j	chicken manure	t	cat
d	pig	k	cow/horse manure	u	lechwe
e	human	l	elephant	v	birds
f	sheep	m	black/white rhinoceros	w	yak
ff	goat	n	buffalo, water buffalo	x	bear
g	deer	o	mink	y	privies, cesspits, urinals
gg	moose	p	kangaroo, wallaby	z	sewage, sewage beds

Afromydaea versatilis Curran	i
Atherigona aberrans Malloch	h
Atherigona orientalis Schiner	c, e, i, s
Azelia ?*aequa* Stein	i
Azelia cilipes Haliday	h, i, x
Azelia monodactyla Loew	h, i
Azelia nebulosa Robineau-Desvoidy	d, h, i, k
Azelia parva Rondani	
Azelia zetterstedtii Rondani	i
Cyrtoneurina parescita Couri	i
Coenosia flavicoxa Stein	i
Coenosia lata Walker	i
Coenosia octosignata Rondani	r
Coenosia testacea Robineau-Desvoidy	i
Cyrtoneuropsis pararescita Couri	i
Dasyphora albofasciata Macquart	i
Dasyphora asiatica Zimin	d, i, k
Dasyphora gussakovskii Zimin	e, i, k, w
Dasyphora himalayensis Pont	
Dasyphora pratorum Meigen	i
Dasyphora stackelbergiana Sychevskaya	i, w, y
Dichaetomyia distanti Malloch	i
Dichaetomyia nubiana Bigot	
Dichaetomyia quadrata Wiedemann	i
Dimorphia cognata Robineau-Desvoidy	i, n
Dimorphia tristis Wiedemann	
Drymeia gymnophthalma Hennig	k
Drymeia vicana Harris	h, i, k
Eudasyphora cyanella Meigen	i
Eudasyphora cyanicolor Zetterstedt	f, i
Eudasyphora tateyamensis Shinonaga	x
Eudasyphora zimini Hennig	i

Graphomya maculata Scopoli	d, h, i, j, k, y
Gymnodia ascendens Stein	i, k
Gymnodia debilis Williston	i, ss
Gymnodia delecta Wulp	c, e, i
Gymnodia distincta Stein	
Gymnodia eremophila Brauer & Bergenstamm	i, k
Gymnodia ezensis Shinonaga & Kano	h
Gymnodia flavescens Stein	i
Gymnodia flexa Wiedemannn	i
Gymnodia gentilis Robineau-Desvoidy	
Gymnodia humilis Zetterstedt	h
Gymnodia lasiopa Emden	i
Gymnodia nigrogrisea Karl	i
Gymnodia normata Bigot	i
Gymnodia obliterata Malloch	aa, i
Gymnodia quadristigma Thomson	i
Gymnodia ruficornis Malloch	i, n
Gymnodia subtilis Stein	h
Gymnodia tohokuensis Shinonaga & Kano	h
Gymnodia tonitrui Wiedemann	i, k
Gymnodia versicolor Stein	i
Haematobia exigua de Meijere	i, n
Haematobia irritans Linnaeus	i, k
Haematobia minuta Bezzi	i
Haematobia titillans Bezzi	i, k
Haematobosca alcis Snow	gg
Haematobosca latifrons Malloch	
Haematobosca sanguinolenta Austen	i, k
Haematobosca stimulans Meigen	g, h, i
Hebecnema fumosa Meigen	i, k
Hebecnema nigra Robineau-Desvoidy	i
Hebecnema nigricolor Fallén	i
Hebecnema umbratica Meigen	i
Hebecnema uniseta Hennig	
Hebecnema vespertina Fallén	i
Helina celsa Harris	h, i
Helina consimilis Fallén	i
Helina deleta Stein	i, x
Helina depuncta Fallén	i
Helina ?*hypopleuralis* Malloch	i
Helina impuncta Fallén	i
Helina ?*laxifrons* Zetterstedt	b, i
Helina lucida Stein	i, j
Helina obscurata Meigen	i
Helina quadrum Fabricius	i
Helina ?*regina* Malloch	i
Helina reversio Harris	i

Helina spp. 1-4 (Ferrar)	
Heliographa excellens Stein	i
Hennigiola setulifera Stein	
Huckettomyia watanabei Pont & Shinonaga	x
Hydrotaea aenescens Wiedemann	d, j, k
Hydrotaea albipuncta Zetterstedt	i
Hydrotaea armipes Fallén	j, k
Hydrotaea australis Malloch	i
Hydrotaea borussica Stein	i
Hydrotaea capensis Wiedemann	d, e, h_3, i, j, k, y, z
Hydrotaea chalcogaster Wiedemann	d, e, j, k
Hydrotaea dentipes Fabricius	b, d, e, h, i, j, k, o, y, z
Hydrotaea diabolus Harris	d, i, k
Hydrotaea floccosa Macquart	d, e, h, i, k, y
Hydrotaea houghi Malloch	e
Hydrotaea ignava Harris	d, e, h, i, j, k, o, y
Hydrotaea irritans Fallén	i, k
Hydrotaea meridionalis Porchinskiy	i
Hydrotaea meteorica Linnaeus	i
Hydrotaea militaris Meigen	i
Hydrotaea obscurifrons Sabrosky	c
~~*Hydrotaea pandellei* Stein~~	
Hydrotaea parva Meade	h
Hydrotaea pellucens Porchinskiy	i
Hydrotaea penicillata Rondani	i
Hydrotaea pilipes Stein	d
Hydrotaea similis Meade	d
Hydrotaea rostrata Robineau-Desvoidy	d, j
Hydrotaea solitaria Albuquerque	e
Hydrotaea spinigera Stein	e, j, k, y
Hydrotaea tuberculata Rondani	i
Hydrotaea velutina Robineau-Desvoidy	i, k
Limnophora mesolissa Bezzi	i
Limnophora quaterna Loew	i
Limnophora simulans Stein	i
Lispe orientalis Wiedemann	
Lispe tentaculata De Geer	“dung”
Mesembrina meridiana Linnaeus	h, i
Mesembrina mystacea Linnaeus	i
Mesembrina resplendens Wahlberg	x
Mesembrina solitaria Knab	bb, i, gg
Metopomyia atropunctipes Malloch	
Morellia abdominalis Stein	i
Morellia aenescens Robineau-Desvoidy	h, i, k, r, w
Morellia asetosa Baranoff	h, i, j, k, v
Morellia hortensia Wiedemann	d, i, n
Morellia hortorum Fallén	d, e, i, k, q
Morellia micans Macquart	e, i

Morellia nilotica Loew	d, i
Morellia podagrica Loew	gg
Morellia prolectata Walker	h, i
Morellia simplex Loew	d, i, k
Morellia violacea Robineau-Desvoidy	e
Morellia zimini Sychevskaya	i
Musca aethiops Stein	i
Musca amita Hennig	h, i, k, q
Musca autumnalis De Geer	bb, d, f, i, k
Musca bakeri Patton	i, n
Musca bezzii Patton& Cragg	h, i, k
Musca biseta Hough	e, i
Musca conducens Walker	h, i, k
Musca confiscata Speiser	i, n
Musca convexifrons Thomson	?h, i, n
Musca craggi Patton	i
Musca crassirostris Stein	i, n
Musca domestica Linnaeus	b, c, d, e, f, ff, h, h_3, i, j, k, p, q, v, y, z
Musca fergusoni Johnston & Bancroft	i, p, v
Musca formosana Malloch	i
Musca hervei Villeneuve	i, k
Musca illingworthi Patton	
Musca inferior Stein	i, n
Musca interruptella Pont	i
Musca larvipara Porchinskiy	d, e, h_2, i, k
Musca lucidula Loew	i
Musca lusoria Wiedemann	e, i
Musca munroi Patton	?e
Musca osiris Wiedemann	f, i, q
Musca pattoni Austen	k
Musca planiceps Wiedemann	i
Musca seniorwhitei Patton	n
Musca sorbens Wiedemann	b, c, d, e, ff, h, i, k, n, t, ?v, y
Musca tempestiva Fallén	e, f, h, i, k, q
Musca terraereginae Johnston & Bancroft	e, h, i
Musca ventrosa Wiedemann	e, i, k, n
Musca vetustissima Walker	c, d, e, f, h, i, p
Musca villeneuvii Patton	l
Musca vitripennis Meigen	d, h_2, i, k
Musca xanthomelaena Wiedemann	e, i
Muscina angustifrons Loew	j
Muscina levida Harris	a, d, i, j, k, y
Muscina prolapsa Harris	i
Muscina stabulans Fallén	c, d, e, h. i, j, k, o, y
Mydaea ancilla Meigen	c, y
Mydaea corni Scopoli	i
Mydaea humeralis Robineau-Desvoidy	
Mydaea obscurella Malloch	d

Mydaea urbana Meigen	e, i
Myospila bina Wiedemann	i
Myospila cuthbertsoni Snyder	i
Myospila japonica Shinonaga & Iwasa	i
Myospila laevis Stein	e
Myospila lauta Stein	e
Myospila meditabunda Fabricius	c, d, e, f, h, i, k, q, y
Neivamyia lutzi Pinto & Fonseca	
Neomyia albigena Stein	i
Neomyia australis Macquart	i
Neomyia coeruleifrons Macquart	i
Neomyia cornicina Fabricius	d, h, i, k, r, w
Neomyia gavisa Walker	h
Neomyia indica Robineau-Desvoidy	i, j, k
Neomyia laevifrons Loew	h, i, k
Neomyia lauta Wiedemann	i, n
Neomyia nudissima Loew	i
Neomyia scatophaga Malloch	i
Neomyia splendida Adams	i
Neomyia timorensis Robineau-Desvoidy	e, i, k
Neomyia viola Bigot	j
Neomyia viridescens Robineau-Desvoidy	d, h, i, k
Neurotrixa felsina Walker	i
Phaonia modesta Sorokina	e
Philornia aitkeni Dodge	v
Philornia rufoscutellaris Couri	v
Polietes domitor Harris	h, i, k
Polietes hirticrus Meade	bb, i
Polietes lardarius Fabricius	c, i
Polietes meridionalis Peris & Llorente	i
Polietes nigrolimbatus Bonsdorff	i
Polietes orichalceoides Huckett	i
Polietes steini Ringdahl	?h
Potamia littoralis Robineau-Desvoidy	b, c, d, e, h, j, k, s, v, y
Potamia scabra Giglio-Tos	i
Prohardyia carinata Stein	i
Prohardyia macalpinei Pont	
Psilochaeta chalybea Wiedemann	s
Pygophora torrida Wiedemann	j
Pyrellia minuta Zimin	e
Pyrellia rapax Harris	i
Pyrellia scintillans Bigot	
Pyrellia tasmaniae Macquart	h, i, k
Pyrellia vivida Robineau-Desvoidy	h, i, k
Rhinomusca dutoiti Zumpt	m
Spilogona pacifica Meigen	
Stomoxys bilineatus Grünberg	i
Stomoxys calcitrans Linnaeus	b, d, e, f, ff, h, i, j, k, l, y

Stomoxys indicus Picard	
Stomoxys niger Macquart	i
Stomoxys pallidus Roubaud	
Stomoxys sitiens Rondani	
Stygeromyia maculosa Austen	e, h, i, k
Synthesiomyia nudiseta Wulp	
Thricops semicinereus Wiedemann	i
Xestomyia pamirensis Hennig	i, r, w

10 In living animal tissue, causing myiasis.

10a In humans

?*Hydrotaea meteorica* Linnaeus
Musca crassirostris Stein
Musca domestica Linnaeus
Muscina levida Harris
Muscina prolapsa Harris
Muscina stabulans Fallén
Stomoxys calcitrans Linnaeus
Synthesiomyia nudiseta Wulp

10b In other mammals

Calliphoroides antennatis Hutton	Sheep
Hydrotaea rostrata Robineau-Desvoidy	Secondary/tertiary, sheep
Hydrotaea spinigera Stein	Tertiary, sheep
Musca domestica Linnaeus	
Musca terraereginae Johnston & Bancroft	Tertiary, sheep
Muscina prolapsa Harris	Sheep
Muscina stabulans Fallén	Tertiary, sheep
Stomoxys calcitrans Linnaeus	
Synthesiomyia nudiseta Wulp	Sheep

10c In birds

Hydrotaea dentipes Fabricius
Hydrotaea similis Meade
Muscina levida Harris
Muscina stabulans Fallén
Passeromyia heterochaeta Villeneuve
Passeromyia indecora Walker
Philornis aguapey Pucheta, Kopuchian, Di Giacomo, Bulgarella & Patitucci
Philornis angustifrons Loew
Philornis bellus Couri
Philornis deceptivus Dodge & Aitken
Philornis diminutus Couri
Philornis downsi Dodge & Aitken

Philornis falsificus Dodge & Aitken
Philornis frontalis Couri
Philornis gagnei Couri
Philornis glaucinis Dodge & Aitken
Philornis insularis Couri
Philornis masoni Couri
Philornis medianus Couri
Philornis mimicola Dodge
Philornis molestus Meinert
Philornis nielseni Dodge
Philornis niger Dodge & Aitken
Philornis obscurinervis Couri
Philornis pici Macquart
Philornis porteri Dodge
Philornis querulus Dodge & Aitken
Philornis sanguinis Dodge & Aitken
Philornis seguyi Garcia
Philornis spermophilae Townsend
Philornis torquans Nielsen
Philornis trinitensis Dodge & Aitken
Philornis vulgaris Couri

11 In domestic garbage.

Atherigona orientalis Schiner
Coenosia testacea Robineau-Desvoidy
Eudasyphora cyanicolor Zetterstedt
Graphomya maculata Scopoli
Graphomya rufitibia Stein
Hebecnema nigra Robineau-Desvoidy
Hebecnema vespertina Fallén
Helina evecta Harris
Helina lucida Stein
Hydrotaea aenescens Wiedemann
Hydrotaea chalcogaster Wiedemannn
Hydrotaea dentipes Fabricius
Hydrotaea diabolus Harris
Hydrotaea floccosa Macquart
Hydrotaea ignava Harris
Musca autumnalis De Geer
Musca domestica Linnaeus
Musca sorbens Wiedemann
Musca terraereginae Johnston & Bancroft
Musca vetustissima Walker
Musca ventrosa Wiedemann
Muscina angustifrons Loew
Muscina levida Harris
Muscina prolapsa Harris

Muscina stabulans Fallén
Myospila meditabunda Fabricius
Potamia littoralis Robineau-Desvoidy
Stomoxys calcitrans Linnaeus

12 In sewage sludge

Lispe superciliosa Loew

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Hemipenthes albus Ávalos-Hernández, 2009 (Diptera: Bombyliidae), new to the United States

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Abstract: *Hemipenthes albus* Ávalos-Hernández, 2009, described from and known only from Mexico, is reported from multiple sites and years in southeastern Arizona (USA). Male terminalia were dissected and an example is illustrated to confirm identification. Two adults are illustrated.

Resumen: *Hemipenthes albus* Ávalos-Hernández, 2009, descrito y conocido únicamente en México, es reportado en múltiples sitios y años en el sureste de Arizona (EE. UU.). Se diseccionaron las cápsulas genitales masculinas, y se ilustra un ejemplo para confirmar la identificación. Dos adultos están ilustrados.

Introduction

Species of *Hemipenthes* Lowe, 1869 inhabiting the Northern Hemisphere have been well-known for more than a century (Hull 1973). More recently, Ávalos-Hernández (2009) discussed the 23 North American taxa in his thorough revision of the genus. In doing so, he made several taxonomic alterations: he moved *H. yaqui* (Painter), from *Hemipenthes* to *Chrysanthrax* Osten Sacken, described two new species from Mexico: *H. translucens* Ávalos-Hernández and *H. albus* Ávalos-Hernández, and removed *H. morio* (Linnaeus) from inclusion in the Nearctic fauna. A decade later, Evenhuis (2020) placed *H. curta* (Loew) and *H. celeris* (Wiedemann) in the newly-created genus *Ins* Evenhuis, reducing the number of North American *Hemipenthes* to 21 species.

One of the authors of the present article (Danforth), resides in Bisbee, Cochise Co., Arizona. Based upon the key and wing illustrations in Ávalos-Hernández (2009), he believed for several years that *H. albus* was present in his yard (Fig. 1). The location of his house near 1,700 m elevation, and the thick oak forest surrounding it, supported the possible occurrence of a northern Mexican, montane species. Photographs of live specimens were compared with Ávalos-Hernández's descriptions and wing images. A series of specimens was collected, and others were photographed in the Huachuca Mountains, the Mule Mountains at Bisbee, on the Mt. Hopkins Road in the Santa Rita Mountains

Figure 1. *Hemipenthes albus* - West Blvd, Bisbee, Cochise Co., AZ, 27 Aug 2011 (D. Danforth photo).

north of Patagonia, Santa Cruz Co., and at several middle and higher elevation sites scattered across southeastern Arizona (Fig. 2).

To confirm the Arizona occurrences, we needed to consider the species described by Ávalos-Hernández and a potential species known from further east. The first of Ávalos-Hernández's two new species, *H. translucens*, is known from the Mexican state of Morelos. The location in Morelos is approx. 1,200 km from the U.S. border, thus its occurrence in the southern USA seemed unlikely. The second species, *H. albus*, was verified from three states in northern Mexico: Zacatecas, Durango, and Chihuahua. The Chihuahua locations are farthest north and approx. 500 km from the southern border of Arizona. Additionally, known Mexican locations of *H. albus* are relatively montane with uplands supporting pine-oak woodland and related habitats. The presence of *H. albus* in southern Arizona, therefore, appeared to be a possibility.

Figure 2. *Hemipenthes albus* - Lower Ash Canyon, Hereford, Cochise Co., AZ, 26 Sept. 2011 (R.A. Behrstock photo).

The widely distributed eastern U.S. species, *H. webberi* (Johnson) has not been reported in Arizona. Its wing infuscation is similar to *H. albus* (Kitts, et al., 2020). Because many plants and animals exhibit a crescent-shaped distribution, occurring in the northern and eastern USA and Mexico, and then extending northward into the American Southwest (e.g. Eastern bluebird, *Sialia sialis* and the butterfly Red-spotted purple, *Limenitis arthemis*), we considered the possibility that the unknown *Hemipenthes* was a disjunct segregate of the widespread eastern *H. webberi* rather than a northern extension of the Mexican *H. albus.* To eliminate that possibility, an examination was conducted of genitalia of the unknown species.

Materials and Methods

To confirm identification, the distal-most portion of the abdomen of four possible *H. albus* were submitted to Socrates Letana for dissection. One of the four specimens was a female and unsuitable for our purposes. The other three were dissected and the terminalia (genital capsule) were photographed from several aspects to verify the presence or absence of unique characteristics. Under a stereomicroscope, the terminalia were cut out using a fine needle. They were macerated in 10% potassium hydroxide solution for 30 minutes, then washed in 70% and 95% ethanol respectively for 25-30 minutes. Finally, they were examined and stored in glycerin. Observation and photographs were made with a Keyence VHX Digital Microscope.

The following data, ranging from 11 Aug 2010–10 Sep 2024, are for all *H. albus* collected or photographed in southeastern Arizona. Those submitted for genitalic examination are boldfaced:

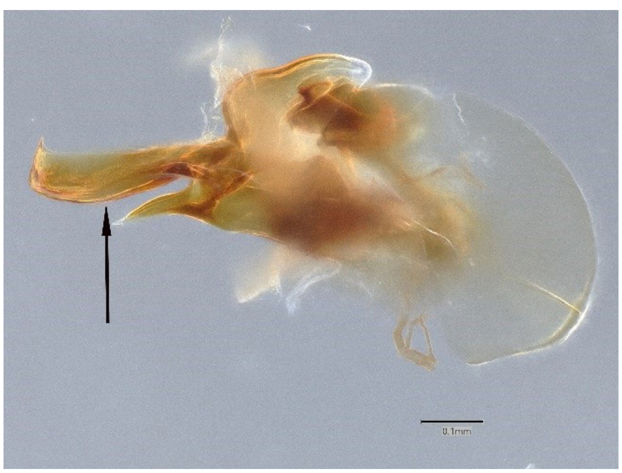
> 16 miles N of I-10 at Benson, Cochise Co., 29 Sep 2021, 32.1595 x -110.3019 (Danforth & Bailowitz); 1 mile S of I-10 on Sibyl Rd, Cochise Co., 30 Aug 2021, 31.9811 x - 110.1803 (Danforth & Bailowitz); **milepost 13, Mt. Hopkins Rd, Santa**

Rita Mtns, Santa Cruz Co., 27 Sep 2023, 31.6733 x -110.8802 (R. Bailowitz); Hwy 186, 2 miles E of Dos Cabezas, Cochise Co., 10 Sep 2024, 32.1601 x - 109.5695 (R. Bailowitz); Hereford, Ash Canyon, Huachuca Mtns, Cochise Co., 25 & 26 Sep 2011, 31.3802 x -110.2279 (R. Behrstock) (Behrstock, 2011); Whitetail Canyon Rd. at Paradise Rd., Chiricahua Mtns, Cochise Co., 8 Aug 2022, 32.0036 x -109.1914 (D. Danforth); south of Miller Canyon, Huachuca Mtns, Cochise Co., 24 Sep 2023, 31.4180 x -110.2451 (S. Tracey); **N Bisbee, Mule Mtns, Cochise Co. 26 Aug 2021, 31.4549 x -109.9408, 2 males (D. Danforth)**; Middle Garden Canyon picnic area, Fort Huachuca, Huachuca Mtns, Cochise Co., 16 Sep 2010, 31.4780 x -110.3435 (R. Behrstock); Miller Canyon, Huachuca Mtns, Cochise Co., 11 Aug 2010, 31.4238 x -110.2637 (C. Melton) (Melton, 2010).

Results

Features cited by Ávalos-Hernández characterizing *H. albus* as belonging to *Hemipenthes* include: flagellomere with a subconical base tapering to a styliform filament for two-thirds of its length, short proboscis, basal third of the wing pigmented brown (with a stair-stepped margin to the infuscation), and relative lengths of several mid-wing veins. Separation of *H. albus* from other members of *Hemipenthes* is based upon th lack of a single microscopic character of the male genital capsule: "The genitalia of *Hemipenthes albus* are unlike any other found within the genus, different by a broad epiphallus in lateral view and absence of a medial extension" (Ávalos-Hernández, 2009, p. 9).

All three Arizona male specimens dissected for species confirmation lacked the downward median extension of the epiphallus and were identified as *H. albus* (Fig. 3). An image of the genitalia of *Hemipenthes bigradata* (Loew), possessing the typical downward median extension is included for comparison (Fig. 4).



Figures 3 (left) and 4 (right). Male terminalia, lateral view: 3. *Hemipenthes albus* Ávalos-Hernández 2009, lateral view. Cochise Co., Arizona. Broad epiphallus lacking downward extension is indicated. (Scale bar equals 0.1 mm.) (S. Letana photo). 4. *Hemipenthes bigradata* (Loew) lateral view (from Ávalos-Hernández 2009, p. 10). Epiphallus with downward medial extension is indicated. (Scale bar equals 600 µm.) Reproduced with permission.

Based on these data, the range of *H. albus* in the USA is nearly confined to Cochise Co. (with one Santa Cruz Co. record) in the southeastern corner of Arizona. The known flight period extends from mid-August through the end of September. With our limited sample size, little can be said about the biology of *H. albus* in the USA. Most individuals were netted or photographed while on the ground, rocks, or low vegetation, in upper Chihuahuan Desert and Madrean Woodland at elevations of approx. 1,010–2,160 m. An individual photographed 25 September 2011 in the Ash Canyon garden of one author (Behrstock), nectared on Scarlet spiderling (*Boerhavia coccinea*: Nyctaginaceae). At the same site the following day, a female was photographed depositing an egg or filling her sand chamber (perivaginal pouch) prior to oviposition.

With the addition of *H. albus*, 14 of the 21 known species of Nearctic *Hemipenthes* are recorded in Arizona.

Acknowledgments

We greatly appreciate the assistance of Socrates Letana, Bohart Museum of Entomology, University of California, Davis, for dissections and photographs of the terminalia of *H. albus*. Omar Ávalos-Hernández, Museum of Zoology (Entomology), National Autonomous University of Mexico, graciously allowed us to reproduce his image of the male terminalia of *H. bigradata*. Thanks to Neal L. Evenhuis, Bishop Museum, Honolulu, Hawai‘i for reviewing a late draft of the manuscript.

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Teratological wings in *Chaetominettia corollae* (Lauxaniidae)

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Teratology among insects seems to be rarely encountered, or at least rarely reported. In Lauxaniidae, an example is a single specimen of *Minettia obscura* (Loew) having one of its antennae bearing a duplicated first flagellomere arising from a broadened scape, with both first flagellomeres bearing their own arista (Steyskal 1968).

The current contribution discusses a wing aberration in a specimen of the lauxaniid species *Chaetominettia corollae* (Fabricius). Before discussing the aberrant wing, it is worth noting that lauxaniids (and many Diptera families) have a diversity of "odd", taxon-specific, wing characteristics – not abnormalities for a given taxon (i.e., they are the norm for those taxa), but odd relative to what may be considered "typical". Following are a few vein-related and structural examples of such wing characters for lauxaniids:

- Loss of anal lobe: *Katalauxania*, *Neogeomyza*, *Noonamyia*
- Presence of auxiliary stump veins (incomplete crossveins): *Dyticomyia*, *Procrita*, *Proteaphila*, some *Siphonophysa*
- Presence of supplementary crossveins: *Xeniconeura*, *Xenopterella*
- Crossvein r-m nearly in line with crossvein dm-m, forming a tight zig-zag line: *Ritaemyia*
- Vein M_1 strongly curved towards tip of R_{4+5}: *Griphoneura*, *Griphominettia*
- Vein R_{2+3} strongly arching towards and closely associated with costal vein: *Depressa*, *Lauxanostegana*, *Lyperomyia*, *Steganolauxania*, *Steganopsis*, *Teratolauxania*

Besides these, an interesting character in some Celyphidae, another lauxanioid family, is the presence or absence of wing crossvein bm-m (see Gaimari 2017). Absence of this crossvein is the state for the genus *Acelyphus.* Although absence/presence had been used to differentiate two putative genera (*Chamaecelyphus* and *Spaniocelyphus*), Gaimari (2017) found that even within series of specimens of the same species in these genera, the presence or absence of this crossvein varied. So although this would not be considered an aberration in the same sense as what will be discussed below, it is interesting to note how some wing characters that are usually discrete can be truly variable.

Among teratologies in dipteran wings, it seems few examples have been published. Among them are:

- major duplication of the venation of the left wing in a specimen of the tabanid *Haematopota abacis* Philip (Philip, 1965)
- a series of spurious crossveins between veins R_{2+3} and R_{4+5} in *Helcomyza mediterranea* (Loew) (Munari 2011)
- loss of vein M_4 in the mycetophilid *Leia ventralis* Say (Oliveira & Amorim 2021)
- various wing teratologies in the hippoboscid genus *Ornithoica* (Bequaert 1954, Maa 1966)
- a bizarre series of spurious crossveins and longitudinal veins in *Platypalpus articulatus* Macquart (Edwards 1914).

So let's introduce our main character – *Chaetominettia corollae*, a widespread and common species throughout Central and South America. Wayne Mathis collected a series of these (housed in the USNM) on 4 February 1984 in a spot 1km south of Tingo María, the capital of Leoncio Prado Province in the Huánuco Region of central Peru. Figure 1 shows a typical individual.

Figure 1. Typical *Chaetominettia corollae* (Fabricius), female, with labels (inset) and wing (bottom).

One individual of the series displayed asymmetrical teratology. Both wings displayed a similar, but slightly different, abnormality related to the r-m crossvein. On the right wing (Figure 2, top), there is a second fully formed r-m crossvein slightly basal to the normal r-m crossvein, complete with the typical wing darkening through the crossvein over both vein R_{4+5} and M_1. To decipher which is the normal r-m crossvein, see Figure 1. On the left wing (Figure 2, bottom), in the same spot as the anomalous second r-m crossvein of the right wing, there is a tiny auxiliary stump vein (really just a bump), along with a darkened spot, but only along vein R_{4+5}.

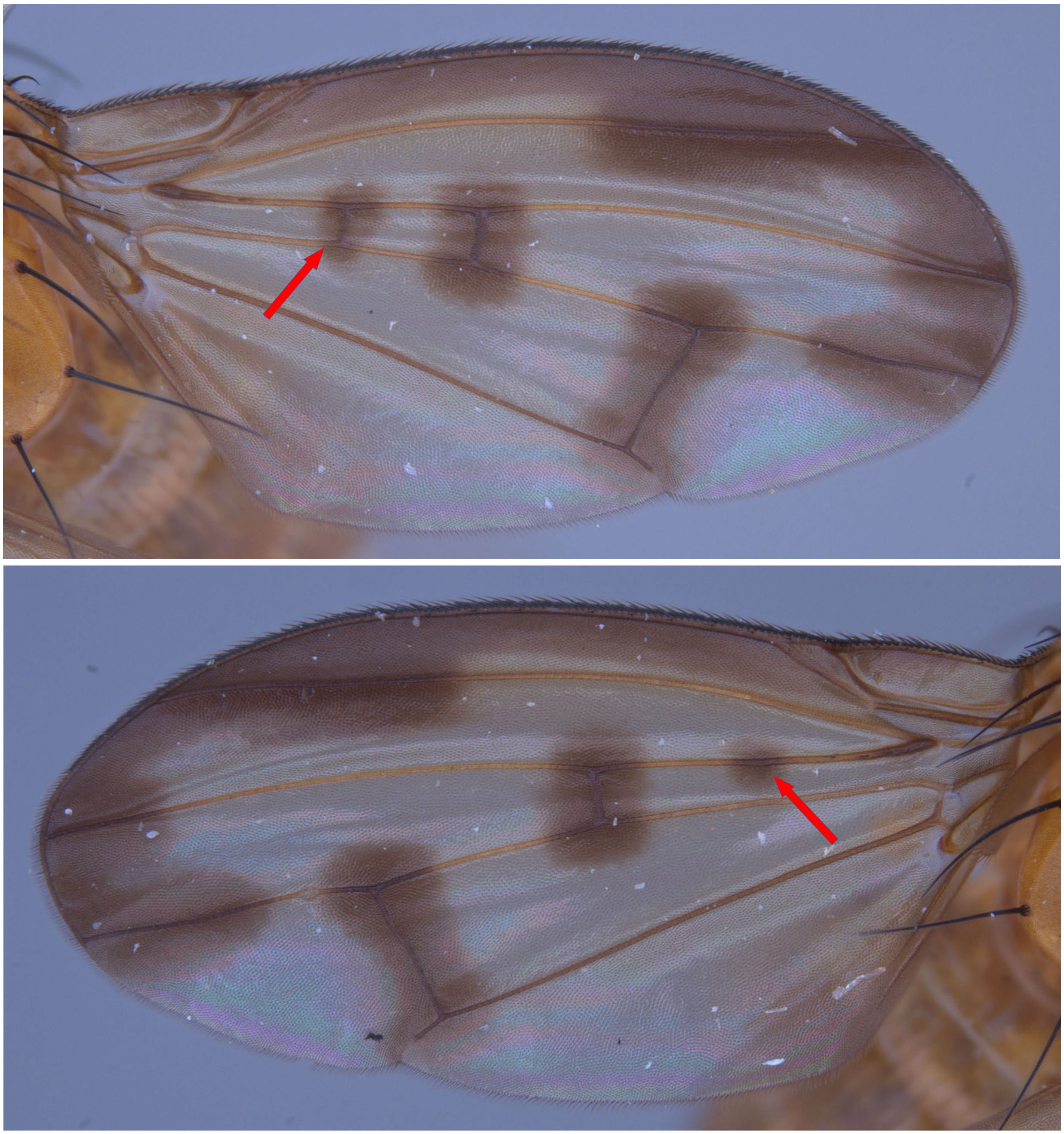

Figure 2. Teratological *Chaetominettia corollae* (Fabricius), right wing (top) and left wing (bottom). Red arrows point to the teratologies on each wing.

To record the entire teratological specimen, the habitus and labels are given in Figure 3.

Figure 3. Teratological *Chaetominettia corollae* (Fabricius), male, with labels (inset).

My final comment on this interesting anomaly is that I would bet many of us encounter such oddities from time to time. But surprisingly very few examples seem to be found in the literature for flies. There seem to be many examples within Hymenoptera and Coleoptera, so I would encourage that such teratologies in Diptera be recorded!

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Systema Dipterorum
The BioSystematic Database of World Diptera

Systema Dipterorum Version 6.5 update
(posted online 29 November 2025)

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[NB: Our full article in the June issue of *Fly Times* inadvertently got clipped. We here present an updated version of it as well as updated credits to those who have assisted us.]

The work on *Systema Dipterorum* continues, and the backlog of taxonomic papers that need to be combed carefully for relevant data is less intimidating than ever before. We are pleased to be able to report the following current numbers (as of 29 November 2025):

Extant valid species-group names: 172,178
Valid genus-group names: 12,751
References databased: 42,271

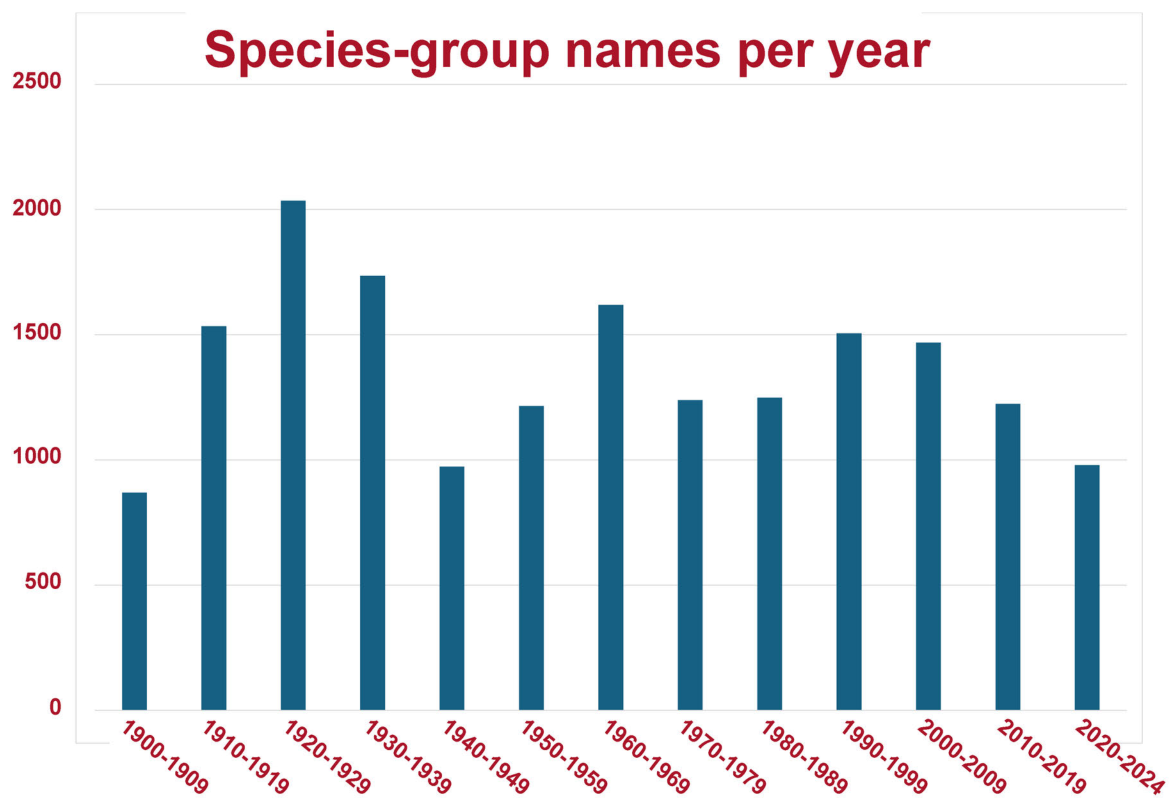

Fig. 1. Number of species-group names of Diptera proposed per year broken up into decades (1900-date).

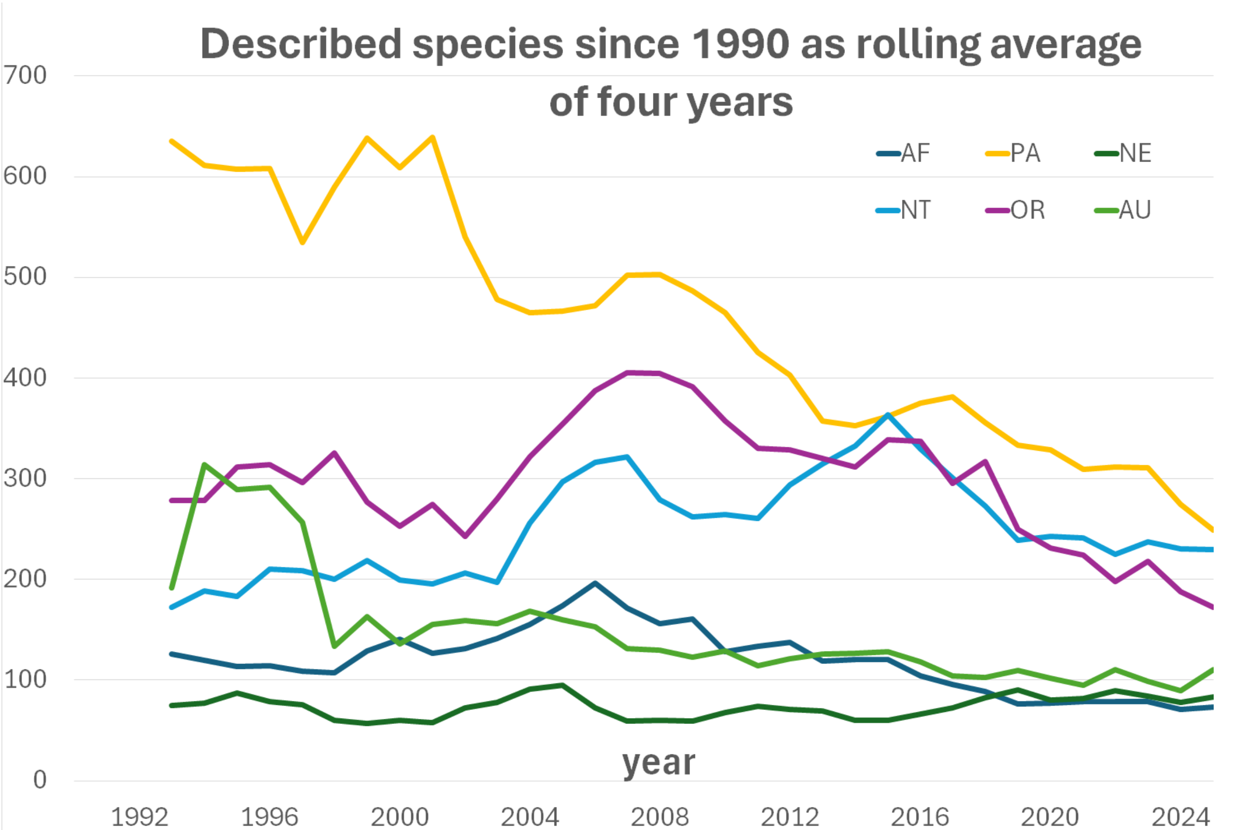

Fig. 2. Number of species-group names of Diptera proposed per year shown as a rolling average since 1990 and separated into the six biogeographical regions. AF = Afrotropical, PA = Palaearctic, NE = Nearctic, NT = Neotropical, OR = Oriental, AU = Australasian–Oceanian.

A total of 170,000+ named species of Diptera certainly is impressive, but looking at the annual taxonomic output since year 1900 shows a worrying decline since the turn of the millennium, with the average number of species proposed per year dropping to a level not seen since WWII (Figs 1, 2). Breaking the data up based on biogeographical region shows that a large part of the decline falls on the Palaearctic Region. With half of the present decade ahead of us, there is still time to change this gloomy trend, and Pierfilippo Cerretti *et al*. (2025) recently published an opinion piece in *Systematic Entomology* (https://resjournals.onlinelibrary.wiley.com/doi/abs/10.1111/syen.70019) discussing the situation and suggesting a way forward.

Meanwhile our work on refining *Systema Dipterorum* continues, and we are happy to bring shout-outs and thanks to those who have helped with corrections, literature, and various other assistance since the notice in *Fly Times* in December 2024 (not in any particular order):

Jean-Sébastien Girard, Steve Gaimari, Vlad Blagoderov, Marc Pollet, Jim O'Hara, Art Borkent, Carlos Lamas, Ximo Mengual, Francisco Welter-Schultes, Adrian C. Pont, Lance E. Jones, Martin Ebejer, Niladri Hazra, Libor Mazanek, Martin Hauser, Luanna Barros, Doug Yanega, Gary Steck, Richard L. Pyle, Barry P. Warrington, Stuart Longhorn, Yury Roskov, Eberhard Zielke, Inocêncio de Sousa Gorayeb, Olga Sivell, Lisa Fisler, Chris Grinter, Paul Rude, Pjotr Oosterbroek, Rüdiger Wagner, Shawn Brescia, Daniel Whitmore, Cecilie Svenningsen, Chris Angell, Alexey Prozorov, Dan Bickel, Charlie Marsh, Eileen Buss, Jeroen van Steenis, Bastiaan Wakkie, Wonseop Lim, Lucio Bonato, Russel Cox, Atanu Naskar, Jens-Hermann Stuke, Peter Chandler, Allen L. Norrbom, Arthur Felipe, Mousumi Chowdhury, Nikita Vikhrev, Mihaly Földvari, Zachary Dankowicz, Jan Ševčík, Stephen M. Smith, Steve Marshall, David Nicholson, Olaf Bánki, Wren Falcon, Arthur Frost, Elisabeth Stur, Hiroshi Shima, Arion Tulio Aranda, Dinesh Sharma, Guangchun Liu, Lius Tito de Morais, Charley Eiseman, Peter Kolesik and Santiago Jaume-Schinkel, Socrates Letana, Brad Sinclair, Paul Langlois, Matthias Jaschhof, Netta Dorchin, Aleida Ascenzi, Barbara Hayford, Shannon Henderson, Carlo Monari, Jere Kahanpää, Daubian Santos, Jing Du, Lucas Sousa-Paula, Tony Rees, Peter Adler, Gil Felipe Gonçalves Miranda, Andrey Przhiboro.

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English common name usage over time in Syrphidae, Asilidae, Chloropidae, and Phoridae

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I thought it would be entertaining to graph the usage over time of the competing English common names used for various groups of flies. This was not done in order to take a side to which term “should” be used, but is just posted here as an illustrative source of information. Google Ngrams (https://books.google.com/ngrams/) does this, and it is relatively simple to plug in two or more common names and let Google Ngrams tell you what the usage is (based on percentage occurrence in the books they have scanned [it does not include Google Scholar]). The graphs below are for Asilidae (robber flies vs assassin flies) (Figure 1); Chloropidae (frit flies vs grass flies) (Figure 2); Phoridae (humpback flies vs scuttle flies vs coffin flies) (Figure 3); and Syrphidae (flower flies vs hover flies) (Figure 4). There are undoubtedly more terms that can be compared, but I thought this might whet the appetite for others to do further analyses.

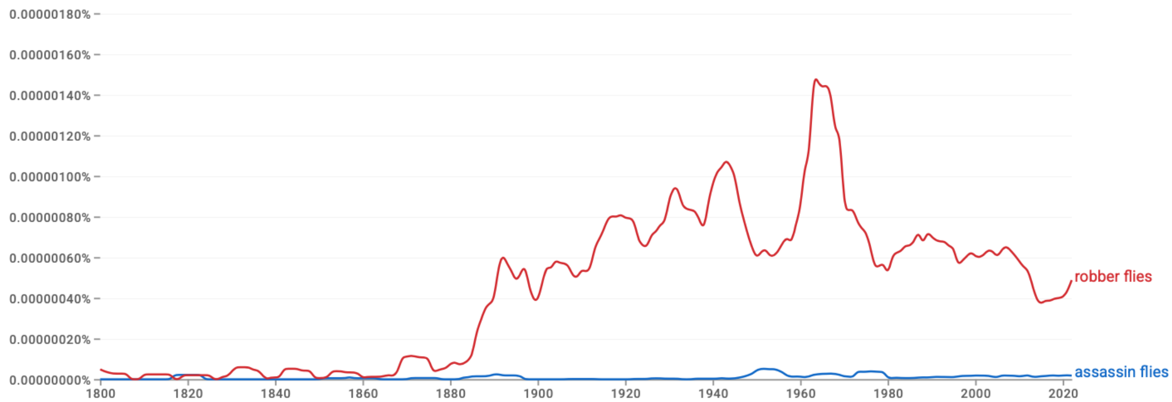

Figure 1. Asilidae common names.

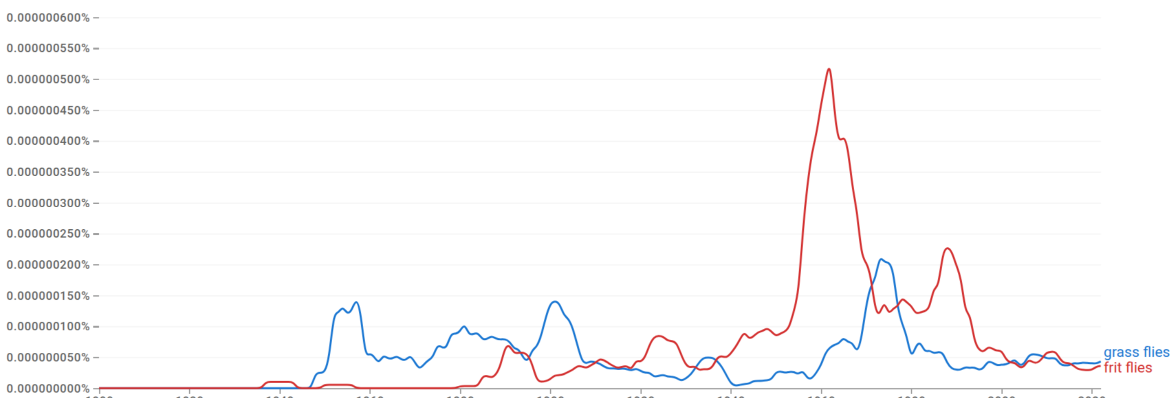

Figure 2. Chloropidae common names.

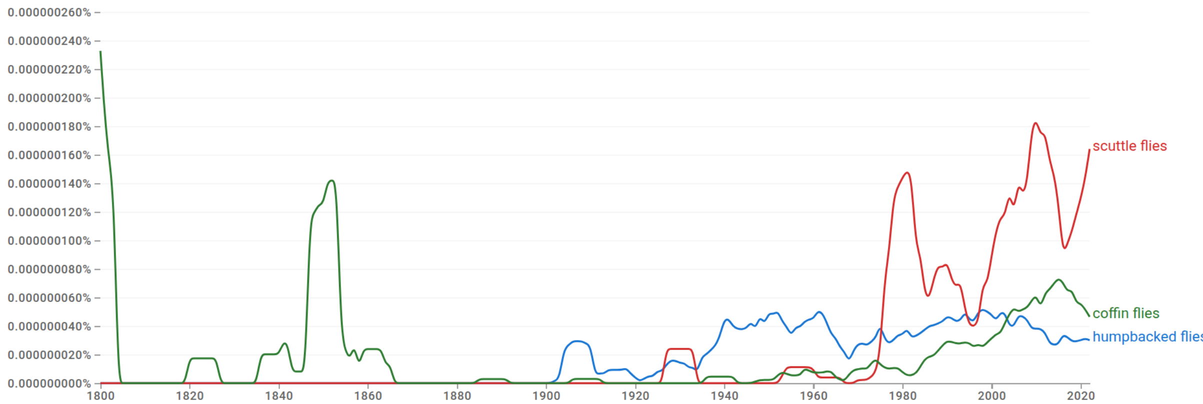

Figure 3. Phoridae common names.

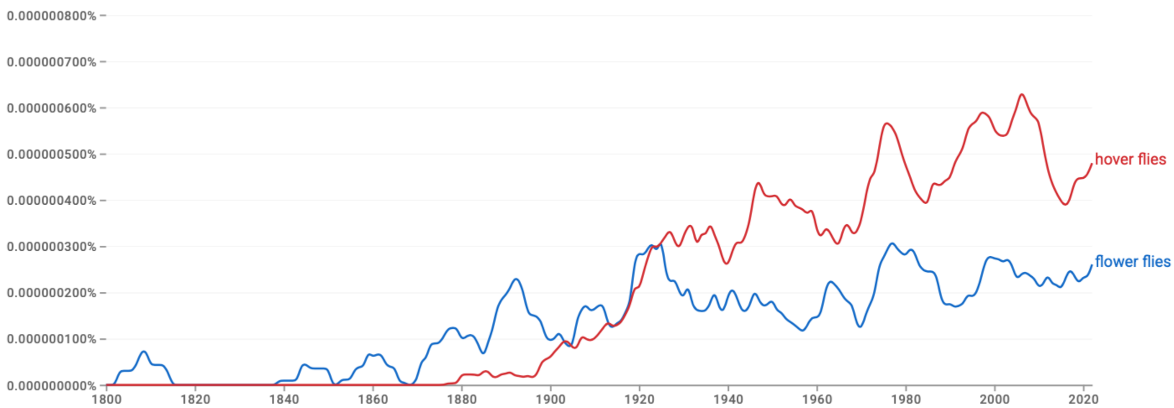

Figure 4. Syrphidae common names.

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Glycerin in an alcohol vial averts loss of a specimen

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Arthropod specimens need to be preserved properly; otherwise, there is no reason to maintain a collection that will soon deteriorate. Preservation is necessary for documentation of occurrence, and to make specimens available for further study. Further study may be morphological or molecular work, each of which may require a different method of preservation. Alcohols have been used as preservatives at least since the 1600s and possibly earlier (Simmons 2014). Glycerin has long been used to preserve small parts of the insect body such as genitalia (Gurney et al. 1964). Early on, entomologists realized that preserving specimens in alcohols alone leads to deterioration of color; using glycerin alone can cause specimens to fall apart (Packard 1873, Kellogg & Slosson 1889). The two alcohols that are most easily obtainable are ethanol and isopropanol; they can be purchased from chemical supply companies, in hardware stores, or in drug stores. The best for DNA extraction seems to be 95% ethanol, with the specimens immersed longer than 24 hours. Isopropanol may result in more discoloration over time and isopropanol removes lipids from specimens over time (King & Porter 2004). Ethanol in a concentration of less than 70% or a volumetric ratio of less than 2:1 (alcohol:specimen) leads to deterioration of the specimen (Webber 2006). Improper storage can allow the alcohol to evaporate, causing the specimen to dry out and become very brittle and shriveled (Banks 1909). Insufficient ethanol concentration can allow fungi and insects to infest and destroy specimens, especially larger taxa such as fish (Pocklington 2015). Banks (1905) mentions that addition of a small amount of glycerin to alcohol may allow the specimen to be salvaged if it dried out. Long ago, in my first entomology course, the professor, the late C.W. Rutschky of the Pennsylvania State University, told the class that putting a drop or two of glycerin into a vial with an alcohol-preserved specimen would give us a chance to save the specimen when (not if) the alcohol evaporated from the vial. Recently I had an opportunity to see this for myself.

I have been collecting Anisopodidae when I find them. I put them into glass vials with alcohol (ethanol or isopropanol) and two or three drops of a 50/50 glycerin/water solution. While looking through my small collection I found a specimen that was lying in a small puddle of glycerin in the bottom of its vial. All of the alcohol had evaporated (Fig. 1). The glycerin itself appeared to have become very viscous and I was worried that the wings of the fly might be damaged. I refilled the vial with ethanol and let it sit while I watched the sequence of events within the vial. It took a while but slowly the alcohol diluted the glycerin and specimen floated to the top of the alcohol in the vial (Fig. 2). The wings were undamaged (Fig. 3). I refilled the vial to its capacity with alcohol and put a piece of Parafilm® over the mouth of the vial before replacing the lid. Thank you, Dr. Rutschky, forty-five years later.

Figure 1. Specimen dried out and adhering to bottom of vial.


Figure 2 (left). Specimen floating freely in new alcohol.
Figure 3 (right). Specimen with (mostly) undamaged wings.

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A look at the presumptive eggs of *Holoplagia guamensis* (Johannsen) (Diptera: Scatopsidae)

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On 28 October 2024, I collected a female specimen of *Holoplagia guamensis* (Johannsen) from the feathers of a Common Yellowthroat, *Geothlypis trichas* (L.) (Passeriformes: Parulidae). During the process of clearing the specimen for a slide mount, I found five eggs in the clearing solution. There being no other specimens in the solution I assumed that the eggs were those of the fly. I mounted each egg on a slide and took photographs through the dissecting microscope and the compound microscope. The eggs were less than a quarter-millimeter long and oval-shaped (Figure 1). They appear yellow because I mounted them in balsam.

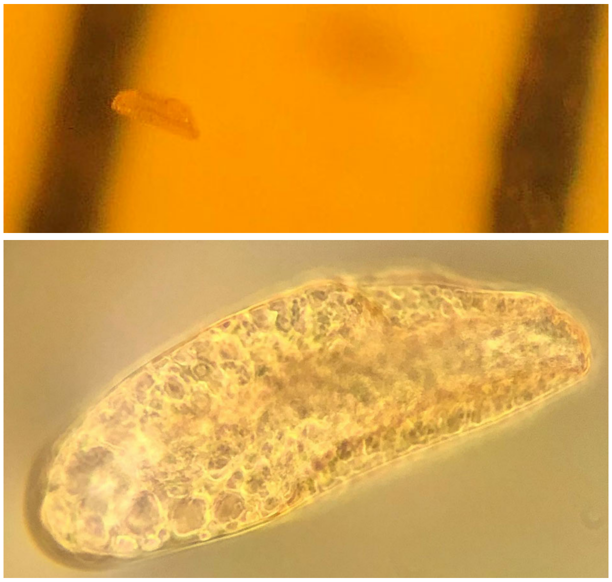

Figure 1. Two putative eggs of *Holoplagia guamensis*. (top) Length; (bottom) Closer view.

The only description of scatopsid eggs that I found was that provided by Cook (1981), who described the eggs of "*Coboldia*" as "ovoid and white". That I found only five eggs is interesting. Meade and Cook (1961) described the oviposition behavior of *Coboldia fuscipes* (as *Scatopse fuscipes*). [Cook (1974) transferred *fuscipes* from *Scatopse* to *Coboldia*.] Meade and Cook (1961) reported that *C. fuscipes* laid eggs in a mass consisting of between 135 and 320 eggs per mass. Choi et al. (2000) also studied the oviposition of *C. fuscipes* and they reported a mean of 282 eggs per female. Mating behavior and oviposition are temperature dependent, as is larval survival (Zhang et al. 2009).

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More mites on midges (Ceratopogonidae)

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On 3 January 2025, I found two midges, a female and a male, of an unidentified Forcipomyiinae, in bycatch from routine mosquito surveillance. Each had a single mite larva attached (Fig. 1). The mites and midges were cleared in a solution of phenol in ethanol and mounted onto microscope slides. The midges are similar in size and coloration to the one that I previously reported from a ceratopogonid midge (Hribar 2024).

Collection data:
USA, Florida, Monroe Co., City of Marathon, Flamingo Island, 26 December 2024, A. Loftus.

Figure 1. Ceratopogonidae with attached mites.

Reference

Hribar, L.J. 2024. Mites (Trombidioidea) taken from Diptera (Ceratopogonidae, Drosophilidae). *Fly Times*, 73: 52-53.

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Water mite larva on a crane fly

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In July of 2025, I was visiting relatives in the Borough of South Heights, Beaver County, Pennsylvania. I took along my personal light traps and set them before dusk on 11 July, retrieving them the next day. I examined the catch after my return to Marathon, Florida. Among the contents of the trap bag was a detached crane fly abdomen. An orange mite was visible attached to the dorsum (Figure 1).

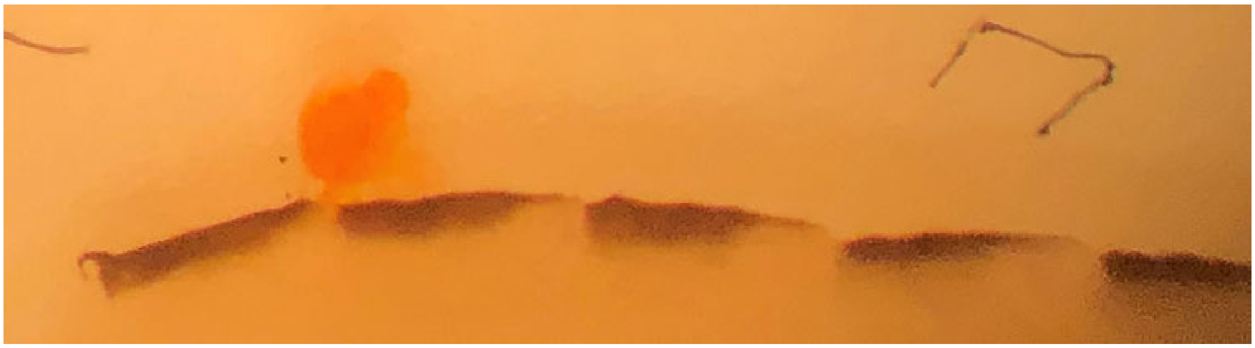

Figure 1. Water mite in situ.

I placed the entire abdomen into a clearing solution, intending to remove the mite after it had softened in the clearing agent. After clearing the abdomen, I removed the mite and I mounted both the abdomen and the mite on a microscope slide under the same cover slip. The abdomen was full of eggs (Figure 2).

Figure 2. Crane fly abdomen full of eggs.

The mite is a typical water mite larva, recognizable by the large plates on its venter (Figure 3). Across the street in Hopewell Township is a small spring and a small, unnamed stream that runs to a larger stream, in the past called Mixter's Run or Mixture's Run, and thence to the Ohio River. Springs are important habitats for water mites (Goldschmidt 2016). Water mite larvae are known to parasitize limoniid and tipulid crane flies (Smith 1991, Hirabayashi & Fukunaga 2007). There is little doubt in my mind that the water mite larva was dispersing to new habitat via the fly. The egg-laden female was in search of a suitable habitat in which to oviposit. I did not attempt to identify the mite. Keys are available (Cook 1974, Smit 2020) but the sheer number of genera (485 in 55 families) and the number of described species (≈ 7,500; Smit 2020) makes for a daunting task for a non-specialist.


Figure 3. Water mite larva from crane fly.

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A mite taken from *Chymomyza amoena* (Drosophilidae)

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On the evening of 12 July 2025, I set a light trap in a patch of woods in Hopewell Township, Beaver County, Pennsylvania. I poured about a cupful of old red wine onto the ground below the trap and then I connected the battery. I retrieved the trap the following morning. Among the few insects that I collected was a small fly with banded wings. Upon closer examination, I noticed a mite attached to the dorsum of the abdomen (Figure 1). I consulted the Bug Guide web page and iNaturalist and determined that the fly was a drosophilid fly, *Chymomyza amoena* (Loew) (Figure 2). The mite appears to be one of the Trombidioidea (Figure 3), a superfamily of mites that contains species that are parasitic on Diptera during the mite's larval stage (Zhang 1998).

Figure 1. *Chymomyza amoena* with mite *in situ*.

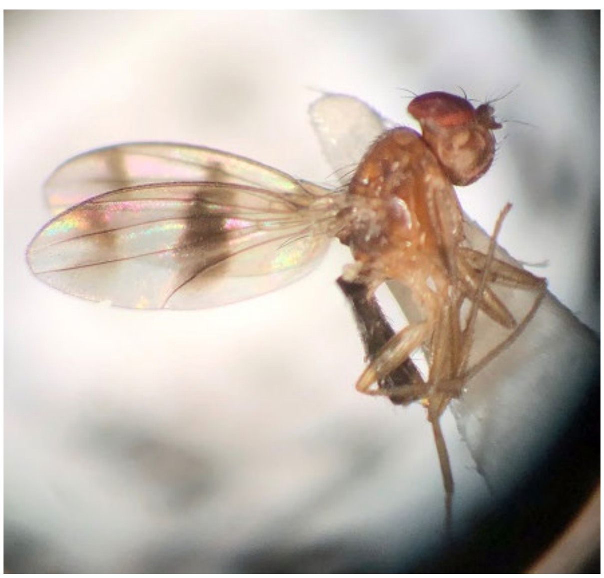

Figure 2 (left). Point-mounted *Chymomyza amoena*.
Figure 3 (right). Cleared, slide-mounted mite larva taken from *Chymomyza amoena*.

Chymomyza amoena (Loew) occurs mostly in the eastern USA but can also be found extending into the western part of the country and even into Mexico and Canada; it has been introduced into Europe, (GBIF Secretariat 2023). Wheeler (1952) provides a key for identification. According to Sturtevant (1921), this fly has been reared from walnut husks (*Julgans nigra*), butternut husks (*Julgans cinerea*), and acorns (*Quercus* spp.), in addition to fruits of apple (*Malus domestica*) and banana

(*Musa* sp.). Band (1988) cites a number of papers corroborating the use of nut husks and acorns as larval habitats, and reports rearing *C. amoena* from apple, crabapple (*Malus coronaria*), and plum (*Prunus* spp.). Upon its introduction into Europe, *C. amoena* exploited a vacant niche and began infesting fruits and nuts (Band et al. 2005). *Chymomyza amoena* is a secondary invader of damaged acorns; it exploits the acorn after curculionid larvae have breached it (Dorsey et al. 1962, Band 1991). The collection site is within the Oak-Hickory Forest where red oak (*Q. rubra*) and white oak (*Q. alba*) are mixed with tulip tree (*Liriodendron tulipifera*), red maple (*Acer rubrum*), and hickories (*Carya* spp.) (Commonwealth of Pennsylvania 2025).

I looked up the definition of "amoena" and found that it is the feminine of "amoenus" that translates as, "lovely, delightful, agreeable, pleasant, beautiful" (neuter = amoenum).

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Erythraeidae mites (Trombidiformes) on *Aedes taeniorhynchus* (Culicidae)

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On 20 August 2025, CC retrieved an American Biophysics Company (ABC) trap deployed the previous evening for routine mosquito surveillance. Among the insects in the collection bag was a female *Aedes taeniorhynchus* Wiedemann that had a small orange mite attached to the anterior dorsal part of the abdomen (Figure 1). LH removed the mite from the fly, cleared it in a solution of phenol in ethanol, and mounted it onto a microscope slide in CMC-10 medium; LH mounted the mosquito on a paper point.

Figure 1. Mite attached to abdomen of *Aedes taeniorhynchus* female.

Collection data:

USA, Florida, Monroe Co., Cudjoe Key, Blimp Road, 20 August 2025, C. Cerminara. ABC Trap.

The mite appears to be in the family Erythraeidae based on the mouthparts and the swollen tarsi on the forelegs (Figure 2).

Figure 2. Prosoma of mite including mouthparts.

On 20 August 2025, AW retrieved another ABC trap deployed the previous evening for routine mosquito surveillance. Another female *Ae. taeniorhynchus* Wiedemann had a small red mite attached to the cervix between the head and thorax (Figure 3). LH removed the second mite from the mosquito, cleared it in a solution of phenol in ethanol, and mounted it onto a microscope slide in CMC-10 medium; the mosquito was mounted on a paper point.

Collection data:

USA, Florida, Monroe Co., Big Pine Key, Big Pine Street, 22 August 2025, A. Weeks. ABC Trap.

The second mite also appears to be in the family Erythraeidae, based on the mouthparts and forelegs (Fig. 4).


Figure 3 (left). Mite attached to cervix of *Ae. taeniorhynchus* female.
Figure 4 (right). Mouthparts and forelegs of mite.

Both of these mite collections were from single mosquitoes amongst hundreds of other conspecific specimens. This is not surprising. Erythraeid mites are not often taken from mosquitoes (Mullen 1975, Williams and Proctor 2002, Milne et al. 2009, Kirkhoff et al. 2013). Williams and Proctor (2002) examined 19,280 mosquitoes in 16 species and found only a single erythraeid mite attached to a mosquito. Simmons and Hutchinson (2016) include a report of an Australian record by Kay (1979), but this seems to be an error; Kay (1979) refers to mites in the genus *Arrenurus* (Arrenuridae). Interestingly, none of the reports and neither of the reviews cited herein mention *Ae. taeniorhyncus* as a host for mites.

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Water mite larvae (Trombidiformes) on *Anopheles crucians* (Culicidae)

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On 3 September 2025, AW retrieved an American Biophysics Company (ABC) trap deployed the previous evening for routine mosquito surveillance. Among the insects in the collection bag was a female *Anopheles crucians* Wiedemann (sensu lato) that had two small red mites attached to the ventral part of the abdomen (Figure 1). LH removed the mites, cleared them in a solution of phenol in ethanol, and mounted them onto microscope slides in CMC-10 medium; LH mounted the mosquito on a paper point.

Figure 1. Mites attached to abdomen of *Anopheles crucians* female.

Collection data:

USA, Florida, Monroe Co., Big Pine Key, Big Pine Street, 3 September 2025, A. Weeks. ABC Trap.

The mites appear to be in the family Arrenuridae, probably a species in the genus *Arrenurus* Dugès, based on the mouthparts, the sclerites, and the setation of the forelegs (Zawal 2008) (Figure 2).

Figure 2. Water mite showing dorsal shield.

We did not attempt species-level identification of the mosquito because what was thought to be one species, *Anopheles crucians* Wiedemann, is in fact a complex of at least five species (Wilkerson et al. 2004, Harbach 2013) and it is unknown which of those species is the true *An. crucians* (sensu stricto) (Harbach et al. 2004).

Water mites in the genus *Arrenurus* (Trombidiformes: Arrenuridae) are commonly found parasitizing *An. crucians* (s.l.) (Lanciani & Boyt 1977). In fact, their seasonal abundance corresponds closely with the seasonality of their host mosquitoes, with the highest numbers reported in May and September (Piwowarek et al. 2024). Thibault (1910) seems to have been the first to report parasitic water mites on *An. crucians* (Mullen

1975). Larvae of *Arrenurus* spp. first attach to the pupa of the mosquito and then move to the eclosing adult (Lanciani 1979a) which they parasitize. The mite larvae sometimes attempt to attach to mosquito larvae. The *Anopheles* larvae can and do defend themselves against mite attack (Lanciani 1988a). Water mite larvae form a tube-like structure called a stylostome in the host tissues; the form of the stylostome is species-specific to the mite (Lanciani & Smith 1989).

Parasitism by water mites can be fatal to the mosquito (Lanciani 1979b, c). Female mosquitoes are parasitized more often than are males (Lanciani 1988b). The parasitic mites reduce survivorship of unengorged female mosquitoes and reduce the number of eggs produced by blood-fed mosquitoes. The proportion of unengorged females bearing mites is higher and the number of parasites carried by unengorged females is greater than for engorged females. Heavily parasitized females have a higher probability of dying before they feed on blood than do unparasitized or lightly parasitized females (Lanciani & Boyt 1977). Mite larvae also parasitize male mosquitoes. Male *An. crucians* die sooner than do females as the number of mites feeding on them increases (Lanciani 1987).

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***Liohippelates peruanus* (Becker) (Diptera: Chloropidae) taken in BG Sentinel® traps set for mosquito surveillance**

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During the spring and summer of 2025, small black Chloropidae started showing up in BG Sentinel® traps (BioGents, Regensberg, Germany) that were set for routine mosquito surveillance (Figure 1). The traps were baited with a proprietary BG bait and carbon dioxide in the form of dry ice. LJH collected specimens from two sites on Vaca Key, City of Marathon, Florida. Collection data are presented in Table 1.

Figure 1. *Liohippelates peruanus* collected in Marathon, Florida, USA.

Most specimens were provisionally identified as *Liohippelates peruanus* (Becker, 1912) with the aid of the identification key of Kumm (1936) and the illustrations provided by Bassett (1973) and Paganelli & Sabrosky (1993). GAF compared our specimens with voucher specimens deposited into the Smithsonian Institution's Museum of Natural History by C.W. Sabrosky. Thirty-five specimens were verified as *L. peruanus*. Two specimens were *L. pusio* (Loew). We are confident that our specimens are *L. peruanus* based on the coloration of the genae, legs, and abdomen, and the setation of the scutum. The taxonomy of the Chloropidae has changed since those papers were published and eye gnats, formerly in the genus *Hippelates*, now are placed in the genus *Liohippelates* (Sabrosky 1980).

Table 1. Chloropidae collected on Vaca Key, City of Marathon, Monroe County, Florida, USA.

Date	Locality	Species	Number
20 May 2025	41st Street	*L. peruanus*	9
1 July 2025	Porpoise Drive	*L. peruanus*	7
1 July 2025	41st Street	*L. peruanus*	5
24 July 2025	41st Street	*L. peruanus*	9
30 July 2025	41st Street	*L. peruanus*	5
30 July 2025	41st Street	*L. pusio*	2

Becker (1912) described *L. peruanus* based on specimens from Argentina, Paraguay, and Peru. The known distribution of this species is: Peru, Argentina, Paraguay, U.S.A. (Florida), Mexico, Guatemala, Nicaragua, Costa Rica, Bahamas, Cuba, Jamaica, Haiti, Dominican Republic, Puerto Rico, Virgin Islands, Guadeloupe, Trinidad, Tobago, Colombia, Venezuela, Ecuador, Bolivia, Brazil (Riccardi 2016). *Liohippelates peruanus* is known from Baker and Monroe counties in Florida (Florida State Collection of Arthropods, pers. comm.). It is likely that it occurs in other counties but has not yet been reported.

It is probable that the flies came from the immediate area around the traps because those areas are in or near grassy fields, larval habitat for Chloropidae (Klepzig et al. 2022). However, it is possible that they may have come from farther away. Dispersal seems not to have been documented for *L. peruanus*, but *L. pusio* can disperse upwind and downwind, between 100 and 1106 meters (Williams & Kuitert 1974).

Pogue & Xue (2023) collected unidentified *Liohippelates* gnats in BG Sentinel traps baited with a proprietary BG bait. Females may be collected from a variety of baits, including liver (Bigham 1941, Axtell & Edwards 1970), shrimp (Spielman 1962, Floore & Ruff 1982), eggs (Axtell & Edwards 1970, Legner 1971), fish (Taylor & Olinger 1958, Axtell & Edwards 1970), animal tissues undergoing initial decomposition (Kumm 1936, Séguy 1940), and carbon dioxide (DeFoliart & Morris 1967, Thompson 1967, Burg et al. 1991, Pucci et al. 2003, Mercer & Moore 2004). Female eye gnats are also easily collected from human or animal bait (Sabrosky 1935; Gerhardt & Axtell 1972, 1973). Males are usually found on flowers (Sabrosky 1935), however, we collected several males in the BG traps.

Whether the flies collected for this report were attracted to the BG bait, the carbon dioxide, or a combination of the two is unknown at this time. An experiment comparing BG traps provided with the different baits to an unbaited trap and a trap with both baits could be conducted to answer this question.

Eye gnats have been recognized as pests in Florida for a long time (Schwarz 1895). Not only are eye gnats pests, some species are associated with disease transmission. Sanders (1940) reported that eye gnats were associated with transmission of bovine mastitis. Historically, the presence of *L. pusio* was correlated with seasonal acute conjunctivitis in the southeastern US (Bigham 1941). In Trinidad, *L. peruanus* were found to be contaminated with *Streptococcus pyogenes*, along with *L. currani* (Aldrich) and *L. flavipes* (Loew) (Bassett 1970, 1973). In Brazil, *L. peruanus* was found to be contaminated with *Haemophilus aegyptius*, the causative agent of Brazilian Purpuric Fever (Tondella et al. 1994). Tondella et al. (1994) collected their flies from or near the eyes of children who had acute conjunctivitis. Legner (1971), however, stated that *L. peruanus* was among a group of species that had "lesser or no response to humans." Reilly et al. (2007) found an association between infection of children with *Chlamydia trachomatis* and the number of Chloropidae collected in the home.

Acknowledgment
Jung (Woogie) Kim and Robert Finn (USDA-APHIS-PPQ-PHP, National Identification Services) graciously provided photographs of a Sabrosky-determined specimen of *L. peruanus* deposited in the National Museum of Natural History, Washington, D.C.

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A new county record for *Culicoides* (*Oecacta*) *stellifer* (Coquillett) (Ceratopogonidae) in Pennsylvania

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In July 2025, while on a visit to see relatives, I hung an ABC light trap (Clarke, Rosedale, IL) from an apple tree in a residential yard in Beaver County, Pennsylvania. Among the few insects collected was a biting midge (Ceratopogonidae). The specimen was cleared and mounted in Canada balsam on a glass microscope slide. It was identified via use of available keys (Foote & Pratt 1954, Blosser et al. 2024). The specimen was determined to be a female *Culicoides* (*Oecacta*) *stellifer* (Coquillett).

Slide Data: PA, Beaver County / South Heights / Oak Avenue / 12 Jul 2025 / L. Hribar // *Culicoides* / (*Oecacta*) / *stellifer* / (Coquillett) / L. Hribar / N. Burkett-Cadena / det.

The specimen was deposited into the Carnegie Museum of Natural History, Pittsburgh, PA.

Culicoides stellifer is a very widespread midge in the United States; it is known from 46 of the 48 contiguous states (Fox 1955, Borkent and Grogan 2009). It is one of 25 species of *Culicoides* reported from Pennsylvania (Hribar and Grogan 2011). Interestingly, there is evidence that *C. stellifer* may contain at least one cryptic species (Shults et al. 2020). The subgeneric classification is also subject to change, as recent studies have revealed that *Oecacta* is paraphyletic (Augot et al. 2017).

The only localities in Pennsylvania where this midge has been reported are Phillipsburg (Centre County) and Fayetteville. This second published record is ambiguous because no county data are given by Foote and Pratt (1954). There is a Fayetteville (census-designated place) in Franklin County, and there is another Fayetteville (hamlet) in Allegheny County.

This new record is of interest not because of the locality but due to the habitat associations and host records for *C. stellifer*. Whereas Erram et al. (2019) reported collecting most of their *C. stellifer* specimens from puddle edges, Blanton and Wirth (1979) wrote that this species utilizes stream margins as a larval habitat. Less than 100 yards from where the trap hung is a small stream that runs directly from a spring into a larger stream, unofficially known as Mixter's Run. This second stream drains into the Ohio River near the Beaver – Allegheny County border. The stream that arises from the spring runs through a wooded area of approximately 0.14 square miles. That wooded area is habitat for White-tailed deer (*Odocoileus virginianus*). Deer frequently visit the apple trees in the aforementioned yard to feed on apples that have dropped to the ground. *Culicoides stellifer* readily feeds on deer (Gerhardt 1986, Smith et al. 1996, McGregor et al. 2019). At least seven arboviruses are associated with *C. stellifer* and some of them are pathogenic to ruminant animals (McGregor et al. 2022). Although Foote and Pratt (1954) wrote that there were no confirmed records of *C. stellifer* biting humans, Reeves et al. (2004) collected this species from human bait.

Acknowledgment
N. Burkett-Cadena, University of Florida, verified my identification of the midge.

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Miscellaneous fly collections in Florida and Pennsylvania

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Psilonyx annulatus (Say)

On the last Thursday of June 2025, I decided to clean out the freezer in one of the labs in the Marathon building. I took out stacks and stacks of Petri dishes and examined the contents for anything interesting. At one point, the contents were interesting to me, because I saved them, but that was a long time ago and usually I couldn't recall why I had saved them in the first place. I was pitching with pride until I ran across a dish of insects that I had collected a year previously, in July 2024, when I went to visit my mother in Pennsylvania. Amongst the insects contained therein I found this little guy (Fig. 1).

Figure 1. *Psilonyx annulatus* (Say).

This is a male *Psilonyx annulatus* (Say), either in the robber fly family (Asilidae) or closely related to the robber flies (Leptogastridae), depending on who you ask. I figured that was the end of it but as usual, it was not. I did a web search for this fly and found out that is has been intensively studied due to its predatory behavior. Its geographic distribution in the United States is east of the Mississippi River (except that is known from Arkansas) and it would be easier to make a list of states where it has not been found than a list of where it has been collected (GBIF). This group of flies is known as "pixies" and they can be mistaken for crane flies and they are very easily overlooked due to their size. This specimen is a little over an inch long and I've seen kite string that is thicker than his abdomen. It seems that one entomologist's trash can be his own treasure.

***Brachydeutera neotropica* Wirth**

Twenty years ago, in 2005, I reported finding the immature stages of a shore fly, *Brachydeutera neotropica* Wirth (Ephydridae), among a large quantity of larvae of the Southern House Mosquito, *Culex quinquefasciatus* (Say) collected from a sewage treatment plant on Vaca Key in the City of Marathon (Hribar 2005). I used to see the pupae of these flies occasionally in larval sample jars along with the *Cx*. *quinquefasciatus*. They can often be found in very polluted water along with the *Culex* spp. larvae. On 21 April 2025, I went looking for mosquito larvae for an experiment that I intend to run. I found an artificial pond that had not been tended to recently and within were thousands of *Cx. quinquefasciatus* larvae. I collected a couple of thousand of them and brought them to the Marathon lab to put them into emergence cages. During sorting of the mosquito larvae I found three dead larvae (Fig. 2, left) and two pupae (Fig. 2, right) of that same shore fly. I put the dead larvae into corrosive to clean them for slide mounts.

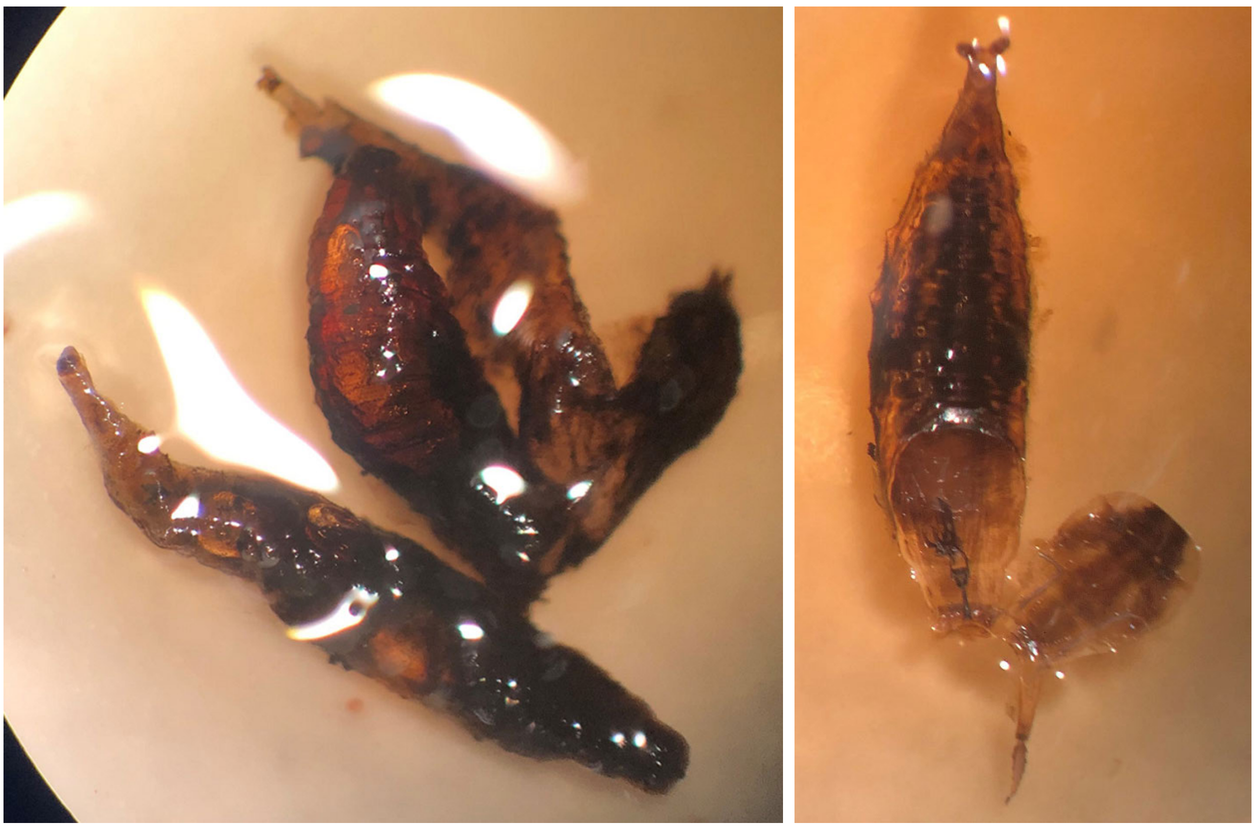

Figure 2. *Brachydeutera neotropica*. (left) Dead Larvae. (right) Puparium.

I was able to rear two adult flies from the pupae (Fig. 3). Even though the sewage situation on Vaca Key has been under control for a few years now, the flies are still around and looking for any crappy situation in which to lay their eggs.


Figure 3. Reared adult of *Brachydeutera neotropica*.

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Investing in intelligence and people: Anastasia Mosquito Control District's commitment to growth, opportunity, and excellence in the past 20 years

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Investing in intelligence by creation of employee continuing education fund & policy
In June 2005, after Dr. Xue became the District Director, he recognized that the district needed to establish and develop an employee continuing education program, which would be crucial to supporting employee success and attraction /retention of talent staff in the field (Xue, 2009, Xue et al. 2023). The Board of Commissioners of Anastasia Mosquito Control District (AMCD) supported his proposal about the creation and development of the employee continuing education funding and policy. The Employee Education Fund policy supports employees who wish to pursue their further education and high degree. The program is open to all eligible employees seeking associate, bachelor, or graduate degrees in fields aligned with operational needs that include biology, public health, data science, and administration. Tuition is fully covered, and employees are encouraged to integrate their academic learning into daily operations. Full-time employees are eligible after one year of employment in the district. Employees may use $3,000 per semester or $6,000 per year for class/course registration and tuition. Major courses are subject to approval by the Director and supervisors. After obtaining a higher degree, they are subject to employment-at-will status and are expected to retain their employment with AMCD for a minimum of one year. Several employees left the district for other jobs within one year after receiving his /her degree due to grant-funded programs or they paid the tuition back to the district.

The full-time biotechnician, biologist, Whitney Qualls, was provided with the 1st beneficiary and used the district education fund to complete her Ph.D. study through the University of Florida (Fig. 1). The 2nd employee, Education Specialist, Jodd Scott (Fig. 2), received her Ph.D. from the UF by using a collaboration grant fund, and the 3rd employee, Biologist, Christopher Bibbs (Fig. 3), received his Ph.D. from the UF through a collaboration grant fund. A former employee who was on an Education Doctor program in UF for about a year, then dropped the program and resigned from AMCD, but he paid the tuition back to the district. A former mosquito control technician dropped from a master's degree program from the University of North Florida and resigned from AMCD and paid the tuition back to the district. Another former employee in the BS program and dropped within a few months. Current employee, Kai Blore works on his Ph.D. and uses 2-year collaboration grant fund. The following AMCD employees received their advanced degrees, where, and what they are doing after they received the district education funds (Table 1).

The program enhanced operational efficiency through employee-led innovations in surveillance, data analysis, and workflow design, improved retention and morale**,** with many participants remaining long-term contributors and mentors, and stronger institutional knowledge, as graduates bring fresh insights and research-based practices into field and administrative protocols. This program supports our district's broader goals of building a replicable, evidence-based operational model, fostering dual-use technology integration for both civic and expeditionary vector control, and promoting staff development and leadership across all levels (Xue & Qualls 2022). It also aligns with Florida's emphasis on transparency and efficiency, reinforcing our commitment to public service and regulatory excellence.

Due to the employee continuing or advanced education for professional development, the district has gradually transformed its team structure with more highly educated employees. Currently, AMCD has six Ph. D.s, six M.S.s, and six B.S./A.A in the 42 full-time employees. The employee's continuing education has benefited AMCD's applied research, operation, and education programs (Xue et al. 2015). Average tenure of program participants vs. non-participants, retention rate increases post-degree, and promotions or role expansions following degree completion. Documented improvements tied to participant initiatives in surveillance protocol enhancements, data analysis innovations, training or onboarding improvements, and case studies of degree-related projects implemented in the district. Total tuition investment vs. estimated operational savings, reduction in recruitment/training costs due to internal advancement, efficiency gains or grant success linked to participant expertise. Survey results on morale, motivation, and professional growth, testimonials from participants and supervisors, impact on workplace culture and team collaboration (Xue et al, 2016). More employees received awards from state and national associations, authored or coauthored more than 100 publications, or gave about 150 presentations (Xue et al. 2014). The three employees who received their Ph.D. are active and become leading/influencing scientists in the field of biology and control of mosquitoes (Fig. 1-3). Other three employees who received their high degrees became managers and continue to serve the district (Fig. 4-6). They are Richard Weaver, Business Manager (Fig. 4), Dena Oliva, Operation Manager (fig. 5), and Scott Hanna, C.F.O. (Fig. 6). Measure metrics in Table 2 showed that the AMCD employee continue education program is successful and benefits graduates, partnerships or outreach initiated by participants, and may influence on other districts and programs.

Figure 1 (left). Dr. Whitney Qualls (2007-2012) is the 1st employee who received the district education fund to receive her Ph.D. in Entomology through the University of Florida. She was a biological technician and promoted to Biologist after her Ph.D. In October 2019 she was rehired as a scientific manager and promoted to assist director in October 2023.

Figure 2 (right). Dr. Jodi Scott (2012-2016) used partial district education funds, and a collaboration grant to receive her Ph.D. in Entomology through the University of Florida. She joined the U.S. Navy and currently, she serves to the Armed Forces Pest Management Board.

Investing people from seasonal and internship to build careers from the ground up

AMCD has built more than a workforce and the careers in the past 20 years. What began as an effort to promote reliable seasonal and intern employees into full-time positions has grown into a comprehensive career development program that fosters leadership from within. Today, many of AMCD's most experienced and respected staff including its current Operation Manager, Laboratory Manager, Assist Supervisor and Field Biologist, began their careers as interns or seasonal employees. For nearly two decades, AMCD has believed that great organizations are built from the inside out.

Every summer, the district welcomes interns and seasonal employees who assist in field operations, research, and public education during the peak mosquito season. These early experiences often become the foundation for lasting careers.

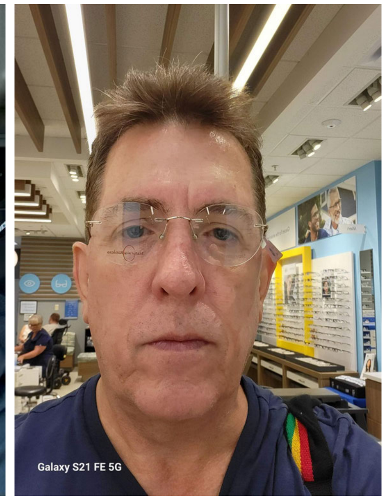

Figure 3 (upper left). Dr. Christoper Bibbs (2013-2019) used the district education fund, and a collaboration grant to receive his Ph.D. in Entomology through the University of Florida. Currently he is Laboratory Director, Salt Lake City Mosquito Abatement District.

Figure 4 (left). Mr. Richard Weaver used the district education fund and received his B.S. degree in Business Management from Flagler College. He was a VCMS Coordinator, promoted to Data Manager after his graduation, then Business Manager.

Figure 5 (upper middle). Dena Oliva was promoted from a seasonal employee to a full-time mosquito control technician. After she received her B.S. degree in Business Management from Flagler College, she was promoted as Assistant Supervisor, then Supervisor. She was promoted as Operation Manager after she used district education fund and received her MBA from Walden University.

Figure 6 (upper right). Mr. Scott Hanna used the district education fund and received his M.S. degree in Accounting from Liberty University. He was a senior accountant, then promoted to Chief Financial Office (CFO).

Recognizing the enthusiasm, curiosity, and dedication of its seasonal workers and intern students, AMCD began offering full-time opportunities to those who showed strong performance, reliability, and alignment with the district's mission when any position creation and vacancy. This approach has proven highly successful, and many former seasonal staff and intern students have advanced into technical, supervisory, and management positions. The result is a team deeply familiar with the organization's operations, culture, and community, and committed to AMCD's goal of protecting public health through effective mosquito control and education. Among the most inspiring examples of AMCD's "grow from within" philosophy is the story of the current Operation Manager. Beginning as a seasonal employee, this individual demonstrated exceptional dedication and initiative from the start. After working for one season, she was offered a full-time mosquito control technician position, where she continued to expand her knowledge and take on greater responsibility. As her education progressed, earning a Bachelor of Business, and eventually an MBA and professional advancement.

She moved from technician to assistant supervisor, then supervisor, and ultimately to Operation Manager. Each promotion reflected not only the personal performance and educational growth but also the evolving needs of the district as positions opened through retirements or restructuring. This journey exemplifies how AMCD's investment in people pays long-term dividends creating leaders who understand every level of the organization because they've experienced it firsthand.

Providing opportunities and internal promotions to retain district excellence and future growth
While the benefits are substantial, AMCD's leadership recognizes that internal promotion programs require thoughtful structure and ongoing support. Not every employee is immediately ready for leadership responsibilities. Transitioning from technical to supervisory roles requires new skills in communication, management, and strategic planning. To address these challenges, the district emphasizes training and mentoring at every stage. Employees are encouraged to pursue continued education, participate in workshops, and seek professional certifications. Supervisors and managers also provide coaching to help team members prepare for future opportunities. AMCD also remains open to external recruitment when fresh perspectives or specialized skills are needed. This balanced approach ensures that innovation and institutional experience grow hand in hand.

Nearly twenty years after its first seasonal/intern-to-full-time promotions, the Anastasia Mosquito Control District has created a proven model of workforce sustainability in public service. By providing clear pathways for advancement from student interns and summer seasonal employees, the district not only fills positions efficiently but also fosters loyalty, stability, and pride among its team members. The greatest strength has always been the people. This is often emphasized by leadership teams. When we invest in their growth, we invest in the long-term success of the community we serve. A total of 26 seasonal employees (Table 1) and 18 intern students (Table 2) were promoted to full-time employees.

AMCD's success story continues to evolve. Each year brings a new group of interns and seasonal employees, many of whom represent the next generation of mosquito control professionals. The district's commitment to mentorship, education, and opportunity ensures that these individuals have the same chance to grow and succeed as those before them. From intern to technician, from supervisor to manager, every step represents AMCD's enduring philosophy: invest in people, promote from within, and build a stronger future together.

Thank the current and former Board of Commissioners and AMCD's dedicated current and former staff and employees' hard work and fully collaboration and support to the mission and programs.

Table 1. Former and current AMCD employees have received the district's education funds to pursue their advanced degrees since 2007.

Year	Name	Degree	College/University	Organization/position
2007-2012	Whitney Qualls	Ph.D., Entomology	University of Florida	AMCD, Assist Director
2013-2016	Jodi Scott (grant fund)	Ph.D., Entomology	University of Florida	Lt. Col, Military
2015-2019	Christoper Bibbs (grant fund)	Ph.D., Entomology	University of Florida	Lab Manager, Salt Lake City MAD
2011-2013	Richard Weaver	B.S., Business & Administration	Flagler College	Business Manager, AMCD
2008-2010	Scott Hanna	M.S., Accountancy	Liberty University	CFO
2021-2026	Kai Blore (grant fund)	Ph.D. student, Entomology	University of Florida	Lab Manager, AMCD
2024-	Aye McKenny	M.B.A. student, Accounting	Flagler College	Accountant, AMCD
2019-2024	Morgan Duet	B.S., Business	Flagler College	Drone Pilot, AMCD
2022-2023	Dena Oliva	M.S., Business & Administration	Walden University	Operation Manager, AMCD
2024-2027	Oliva Sypes	M.S. student, Entomology	University of Florida	Biotech, AMCD
2024-	Edward Zeszutko	M.S. student, Entomology	University of Florida	Biotech, AMCD
2024-2026	Connor Kupe	M.S. student, Entomology	University of Florida	Biotech, AMCD
2024-2025	Heather Keating	B.S., Business & Administration	Flagler College	Administration Assist (resigned)
2010-2013	Kay Gaines	A.A., Business	St. Johns River State College	Operations manager, AMCD(retired)
2024-	Kody Fisher (not AMCD fund)	M.S. student, Epidemiology	University of South Florida	Biotech, AMCD
2024-	Lauren Van Rhee (not AMCD fund)	M.S. student, Forensics	University of Florida	Biotech, AMCD

Table 2. Measure metrics for employees continue education program at AMCD.

Metric	Description	Example
Total Participants	Cumulative number of employees enrolled	19 since 2007
Completion Rate	% of participants who completed their degree	79%
Degree Types	Breakdown by degree level	AA, BA, MS, Ph.D.
Retention Rate	% of participants still employed 3+ years post-degree	84%
Promotions	# of participants promoted post-degree	9
Operational Contributions	# of implemented projects tied to coursework	12 documented
Cost per Degree	Average tuition cost per participant	About $12,000 (some with different grants)
Return On Investment (ROI) Estimate	Operational savings or grant wins linked to participants	$500K in grant funding

Table 3. Employees were promoted from seasonal and intern student employees to full-time employees to build their careers in the past 20 years.

Year of Promotion	Seasonal Employee	Job Title	Intern Student	Job Title
2005	Steven Steele	Mosq Control Technician (MCT)		
2006	Tom Downey Michelle Davenport	MCT MCT		
2008	Dave King	MCT		
2013	Rick Stockley Ryan Grubbs	MCT-IT specialist MCT	Alice Fulcher	Biologist
2014	Barry Scott Mike Vaughn	MCT MCT	Jennifer Gibson Emily Thomoson	Biotech Biotech
2015	Jerry Iser John McClure Dena Autry/Oliva	MCT MCT MCT/Operation Mgr		
2016			Codi Anderson Joseph D'Amato	Biotech Biotech
2017	Morgan Duett	MCT-Dron Pilot	Steven Smoleroff Carly Mangue Jeremy Wholforth	Field Biol Biotech MC Tech
2019			Kai Blore Lea Bangonan Coutney Cunningham	Biotech/Lab Mgr Botech Biotech
2020			Olivia Sypes Madeline Steck	Biotech Biotech
2021	Dazmond Hackney	MCT	Taylor Ballantyne Aye McKenny	Ed Specialist Accountant
2022			Holly Usina Edward Zeszutko Heather Keating	MC Tech Biotech Adm Assist
2023	Nicole Blackwelder Kyle Graham	MCT MCT	Tomomi Hirokawa	Ed Specialist
2024	James Stockley Cameron Clark		Genhsy Monzon Lauran van Rhee Kody Fisher	DVEC Coordinator Biotech Biotech
2025	William Cotter Madison Morris Teresa Hairston Nydia Negron	Biotech MCT MCT Custodian	Uvina Allen	Biotech

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HISTORICAL DIPTEROLOGY

Rudolf Rozkošný, 1 September 1938 – 15 November 2025

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With deep sadness, I learned of the passing of Rudolf Rozkošný. We have not only lost one of the great dipterists of our time, but also an exceptional personality and a truly exemplary human being. Rudolf was an extraordinarily active, tireless dipterologist with profound expertise in Stratiomyidae and Sciomyzidae, and he contributed to many other families as well. His work in faunistics, larval descriptions, and biogeography further showcased the breadth of his knowledge. His achievements live on through his many papers and books, which now form an enduring legacy.

I would like to share a more personal perspective, because Rudolf was my mentor, my role model, and my friend. My interest in Diptera began early, and although I started with Syrphidae, I soon became increasingly fascinated by Stratiomyidae. That was how our correspondence began. Rudolf was always kind, patient, and genuinely supportive.

In the January 1997, Jens-Hermann Stuke and I decided to drive to the Czech Republic to visit several of our Diptera colleagues, and of course a visit to Brno to meet Rudolf was part of the plan. The meeting was interesting and wonderful. His openness and warm, welcoming manner instantly put us, two young students, at ease. I will never forget his white hair, giving him a professorial appearance, or his intelligent, focused eyes that always seemed to smile. He carried himself with the calm confidence of someone who knew what he had accomplished, self-assured without being arrogant or competitive.

During our conversation, I mentioned, in my youthful enthusiasm, a few errors I had found in his famous two-volume work on European Stratiomyidae. He listened carefully, smiled, and said, "Why don't you write a third volume, titled *Errors and Mistakes of Rozkošný*?" He said it in such a warm and humorous way that it felt like encouragement rather than criticism. We all laughed about it, and in the end, of course, there weren't nearly enough errors for even a single paper, let alone a book. From that moment on, our understanding of each other deepened, and we went on to collaborate on many projects and publications.

Reading his emails was always a joy: sharp observations, deep knowledge, and everything sprinkled with his fine sense of humor.

At one point, he was working on some specimens I had sent him and discovered a new species among material from Sri Lanka. I supported him by studying Erwin Lindner's material in Stuttgart, where I worked at this time, but I was genuinely surprised when he later sent me the manuscript listing me as a coauthor. When I told him I hadn't expected that, he simply replied that as a young entomologist, I would need publications to get a job. Not long after that, we worked on the checklist of Sri Lankan Stratiomyidae, and he insisted that I should be the first author because, as he told me, he already had a job, while I still needed a permanent position.

This generosity left a lasting impression on me, and I have tried to follow his example throughout my career. He worked, collaborated and published with many scientists from all around the world. He was always open to joint publication, and he knew there were enough species and discoveries for many more Dipterists, so there was no need to be territorial. Rudolf always chose cooperation over competition, support over criticism.

Perhaps that was his secret, always being so happy and content.

Whenever I study these fascinating flies, and find something I knew he would find interesting, I can see him smiling from a distance.

I realized that I have only this one photograph with Rudolf and myself in it. But at least it has also a very high number of Stratiomyidae workers in it. Taken at the Diptera Symposium in Curitiba (Brazil) in 2001. Lef to right: unknown, Martin Hauser, Rudolf Rozkošný, Wayne Mathis, Dianne Mathis, Jose Pujol-Luz, Ana Claudia Dias de Oliveira, Roberto de Xerez and Alexandre Ururahy-Rodrigues)

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A blast from the past!

Neal L. Evenhuis

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And here is a blast from the past, marking the 30th anniversary of the 1995 North American Dipterists Society Field Meeting in Costa Rica (at San Gerardo de Dota)! Sorry for the couple of missing names! And thanks very much to Chris Maier for filling in a couple where I wasn't sure!

Front row (L to R): Marion Kotrba, ??, Frank French, Greg Forbes, Hannah Nadal, Chen Young, Rob Cannings, Brian Brown, Marty Condon, Manuel Zumbado, Elke Buschbeck, Greg Dahlem
Second row (L to R): Neal Evenhuis, Donald Webb, Eric Fisher, Al Norrbom, Larry Quate, Thomas Pape, ??, Steve Marshall
Back row (L to R): David Caloren, Monty Wood, Riley Nelson, Mark Metz, Sturgis McKeever, Jeff Cumming, Dick Vockeroth, John Burger

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PHILAMYIANY

Diptera on stamps (10): Simuliidae

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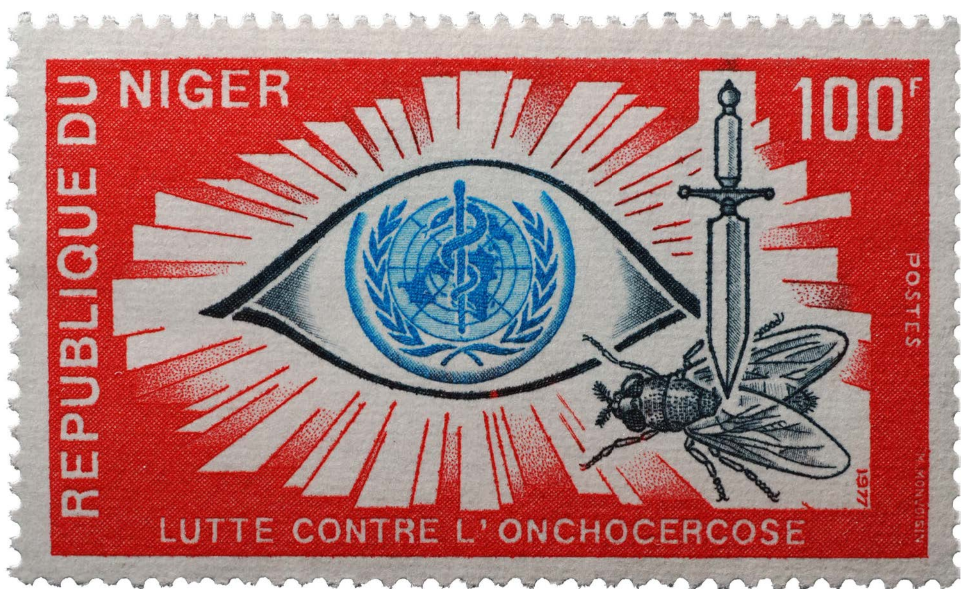
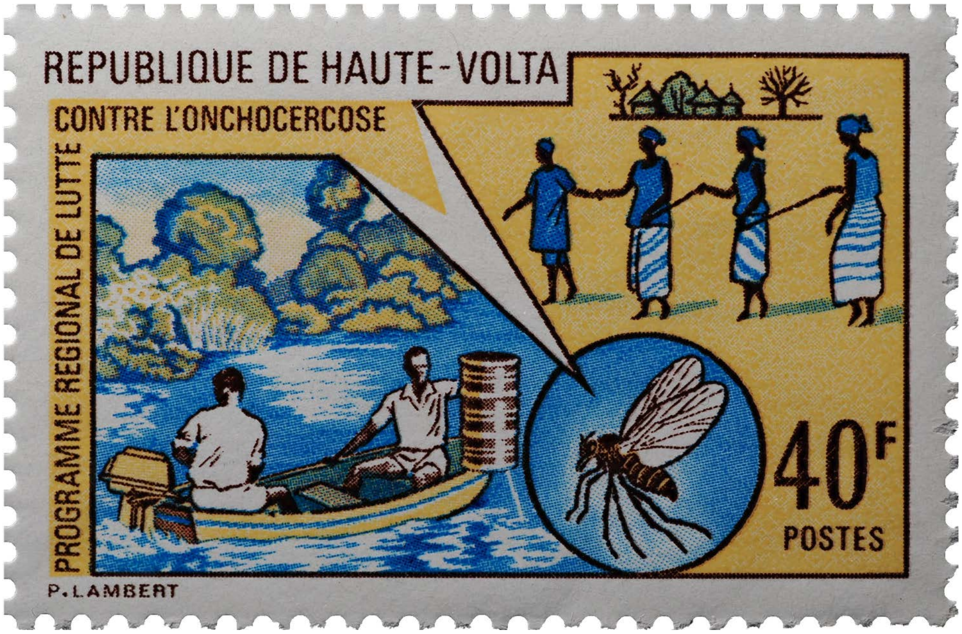
As bloodsuckers, Simuliidae are pests and transmit diseases. In particular, river blindness (onchocerciasis), which can lead to blindness and mainly occurs in tropical Africa, has Simuliidae as vectors. This is the reason why stamps from African countries (Burkina Faso, Ghana, Niger, Togo) indicate the danger of Simuliidae. Sandflies are a common sight among visitors to beaches in New Zealand or the West Indies, for example, and have certainly inspired the corresponding stamps.
Here are a few philatelic remarks. The overprint "O. M. S." on the Burkina Faso stamp stands for "Organization Mondiale de la Sante". The World Health Organization regularly used regional stamps with this overprint for its official mail. These stamps are regarded as independent postage stamps and therefore have their own number in the stamp catalogues. The absence of numbers, such as on the Nevis stamp, means that the issuers do not accept the stamp as an official postage stamp. This applies to stamps that were printed exclusively for collectors but could never be used to send letters. There are, of course, transitions when, for example, letters with canceled stamps exist without these actually having been transported from sender to recipient by a postal service. It can happen that stamps are given a number in one catalogue but not in another. An example of this is the stamp from New Zealand. The question of how many stamps show Simuliidae is therefore remarkably difficult to answer.

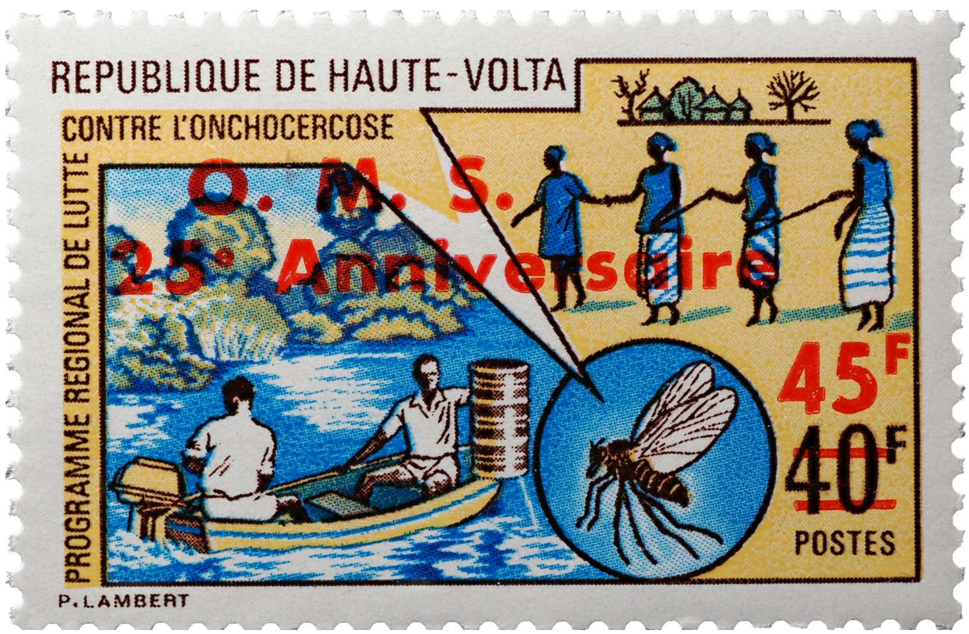
For each stamp I have provided the country and year of issue, title of stamp, title of stamp series (where available/relevant), face value, Michel number and stamp number (the latter both copied from https://colnect.com/).



***Simulium* spec. – Niger** 1977: Lutte contre l´onchocerose, 100 West African CFA franc. – Michel number: NE 578; stamp number: NE 398.



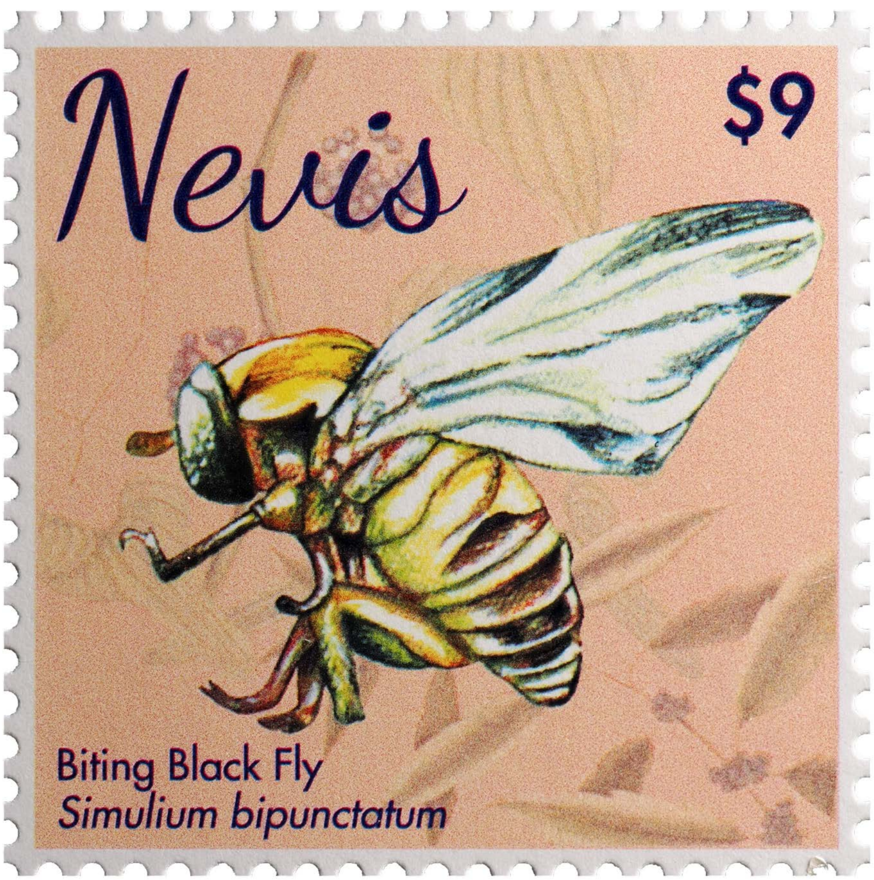
***Simulium* spec. – Burkina Faso [Upper Volta, 1958-1984]** 1971: Programme regional de Lutte contre L´onchocerose, 40 West African CFA franc. – Michel number: BF 355; stamp number: BF 260.



***Simulium* spec. – Burkina Faso [Upper Volta, 1958-1984]** 1973: Programme regional de Lutte contre L´onchocerose, 45 West African CFA franc. – Michel number: BF 295; stamp number: BF 286; with overprint „O. M. S. 25. Anniversaire, 45 F“.

***Simulium* spec. – Ghana** 1991: UNDP, 60 Ghanaian cedi. – Michel number: GH 1497; stamp number: GH 1292.

***Simulium* spec. – Ghana** 1976: Foresight prevents blindness, checking the effects of insecticide on blackfly larvae [World health day 7th.April, 1976], 1 Ghanaian new cedi. **–** Michel number: GH 661A; stamp number: GH 595.



***Simulium bipunctatum* Malloch, 1912 (= *ochraceum* Walker, 1861) – West Indies [Nevis]** 2013: Biting Black Fly, *Simulium bipunctatum* [Caribbean insects], 9 . **–** Michel number: -; stamp number: -.

***Simulium* spec. – Togo** 1976: Prevention de la cecite, 50 West African CFA franc. – Michel number: TG 1154A; stamp number: TG 930.

***Simulium* spec. – New Zealand** 2014: Pukekura´s Giant Sandfly [Legendary Landmarks], 80 New Zealand cent. – Michel number: NZ 3156; stamp number: -.

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MEETING NEWS

Dipterists Society – North American Field Meeting, June 2026

Will Bouchard[1] & Matt Petersen[2]

[1] Minnesota Pollution Control Agency St. Paul, Minnesota 55155, USA; bouc0048@umm.edu
[2] Department of Entomology, University of Minnesota, St. Paul, Minnesota 55108, USA; pet03207@umn.edu

We are pleased to announce that the 2026 Dipterist Society 19th North American Field Meeting will be held June 15-19, 2026 at the Itasca Biological Station and Laboratories (https://cbs.umn.edu/itasca) in northern Minnesota. The Itasca Biological Station is located at the headwaters of the Mississippi River and in Itasca State Park and was selected because it provides opportunities to sample in a variety of different habitats on the station campus and in the area. The facilities are also perfect to support this type of event. In previous years, meetings in the United States were held on either the East or West Coast and this will be the first meeting in the Upper Midwest so it will be an excellent opportunity to collect Diptera from a new region.

About the location: Itasca Biological station is on the eastern shore of Lake Itasca and consists of a 49-acre campus completely inside of Itasca State Park. It provides an excellent opportunity to sample fly diversity from a variety of interesting habitats, including dry old growth jack pine forest and extensive areas of groundwater-fed forested peatlands. The station is located near the convergence of four major biomes (Eastern Broadleaf, Mixed Forest, Prairie Parkland, Tallgrass Aspen), allowing for regional sampling that is not present in other areas of the Upper Midwest. Examples include prairies (Felton, Bluestem Prairie, and Santee Prairie Scientific and Natural Areas), fens (Gully Fen Scientific and Natural Area), sand dunefield (Agassiz Dunes Scientific and Natural Area), and forests (Badoura Jack Pine Scientific and Natural Area). We are currently determining the specific sites that we will visit and will provide that information soon. In addition to the sampling excursions, attendees are encouraged to submit a 10–15 minute presentation for the evening presentations.


Badoura Jack Pine Scientific and Natural Area (Minnesota Department of Natural Resources, Kelly Randall)


Gully Fen, Scientific and Natural Area (Minnesota Department of Natural Resources, Tyler Janke)

Bluestem Prairie Scientific and Natural Area (MN DNR, Kelly Randall)

The biological station offers housing and meals in a common facility that is a reasonable distance from sampling locations. We plan to have different options for lodging including less expensive bunkhouses and more private cabins (https://cbs.umn.edu/itasca/staying-station/lodging). Dining is also provided on site (https://cbs.umn.edu/itasca/staying-station/dining) and 3 meals per day will be provided plus dinner on the arrival day (June 15) and breakfast on the departure day (June 19).

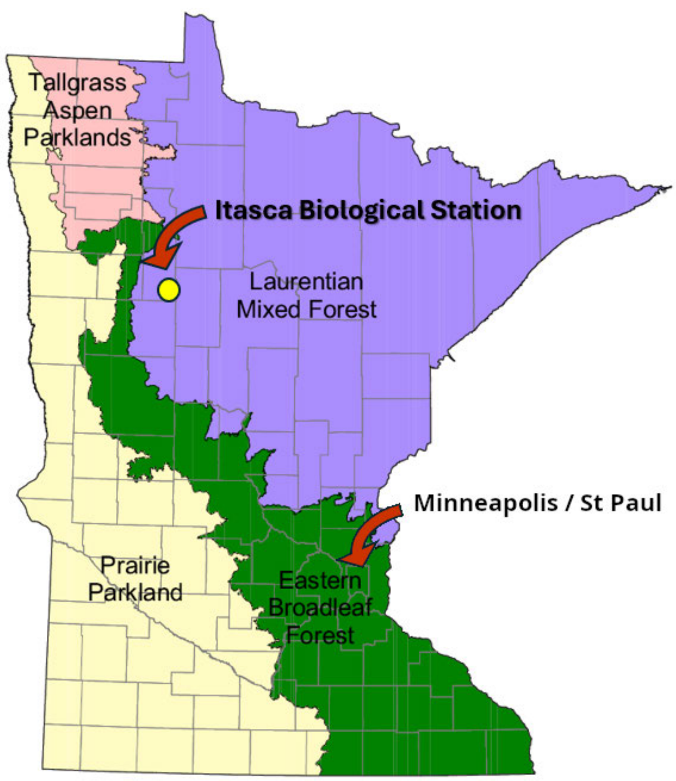
General information for the Itasca Biological Station can be found here: https://cbs.umn.edu/itasca/staying-station/life-station.

Housing options include both shared bunkhouses and private cabins with kitchen and bathroom facilities.

Travel to the station: Attendees traveling by air will likely want to fly into the Minneapolis–Saint Paul International Airport (MSP). The station itself is fairly remote and requires a 4 hr drive from Minneapolis. The organizers are working to arrange shuttle vans to the station. Vehicle rental is also available at the airport. If you are driving, there is plenty of parking, but you will need to purchase a Minnesota state park pass.

Cost estimates: There is some flexibility in accommodations and meal plan options (https://cbs.umn.edu/itasca/staying-station/rate-information). Plans are available for individual meals, or as a full day meal plan. Options for eating out are limited, but also do exist. Additionally, most cabins have full kitchens and allow for you to plan your own meals. Cabins range from bunkhouses with multiple beds, to larger studio or 1-4 bedroom cabins. We estimate a registration cost of ~$350-$500 for a four night stay and three full day meals depending on the lodging type selected.

There are also many options for excursions while visiting Minnesota outside of the meeting. If you are interested we can arrange a visit to the University of Minnesota Insect Collection (https://insectcollection.umn.edu/).



There are also opportunities to visit museums in the Twin Cities, hiking in one of the many state parks, or if you are adventurous embarking on a trip into the Boundary Waters Canoe Area.

If you are interested in attending please let us know by filling out the form at https://dipterists.org/field_meetings.html. This will help us plan this meeting and ensure the accuracy of the registration costs. You can also contact one of us if you have any questions.

We hope to see you in Itasca!

Keep an eye on the same website for updates, and for registration when it opens. Further announcements will be made through the dipterists mailing list.

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Dipterists Society Meeting wrap up: ESSA 2025

John Midgley[1,2] & Burgert Muller[3]

[1] KwaZulu-Natal Museum, Pietermaritzburg, South Africa; jmidgley@nmsa.org.za
[2] Dipterists Society, P.O. Box 231113, Sacramento, California, 95823, USA
[2] National Museum, Bloemfontein, South Africa

For the first time, a Dipterists Society meeting was held at the Congress of the Entomological Society of Southern Africa (ESSA). The meeting was jointly organized by Burgert Muller and John Midgley.

While the Dipterists Society, and its predecessor the North American Dipterists Society, has a long history of holding meetings at the Entomological Society of America annual meeting, the new global reach of the Society means that meetings can now be held at any congress, conference or other meeting where Dipterists are gathered. This year saw the 1st Dipterists Society General Meeting at ESSA, which we hope will not only be the first of many at ESSA, but also the first at many other meetings!

The meeting proved to be very popular, with 21 talks submitted for the session, which ended up filling an entire day. A range of topics was covered, including talks on taxonomy and systematics, endosymbionts, biodiversity, agricultural entomology, medical entomology, and niche modelling. Several of these talks were videoed and can be viewed on the Dipterists Society YouTube channel (https://www.youtube.com/@dipterists).

The full playlist, with links to each presentation, is available at:
https://www.youtube.com/playlist?list=PLAgh5E0fAQ5rNmLWWUzSdpc37VxU72aQB

Figure 1. John Midgley explaining the virtues of joining the Dipterists society ESSA 25 at the University of the Free State, Bloemfontein, South Africa. (Photo by Kurt Jordaens)

The ESSA programme was already quite full, so there was no opportunity for a formal meet and greet, but delegates managed to meet up during the happy hours and at the Society table, which was kindly sponsored for the ESSA organizers.

Figure 2: John Midgley manning the stand at ESSA 25 at the University of the Free State, Bloemfontein, South Africa. (Photo by ESSA)

ESSA meetings are held every two years, and we look forward to the 2nd Dipterists Society General Meeting at ESSA, which will be held in 2027 in Mpumalanga, South Africa!

Full list of talks:

John Midgley	Introduction to the Dipterists Society
Privilege Tungamirai Makunde	Tachinid diversity in *Euproctis terminalis*: a step towards augmentative and conservation biocontrol.
Jarmaine Magoai	An overview of the *Atherigona* of southern Africa.
Cassandra Barker	Exploring Bombyliidae diversity in South African biodiversity hotspots.
Robyn Manuel	Investigating species boundaries and pollination dynamics of *Rhigioglossa nitens* Chainey, 1987 and *Rhigioglossa edentula* Wiedeman, 1828 (Tabanidae) along the Saldanha peninsula.
Kirstin Williams	The state of the genus *Hybomitra* Enderlein (Diptera: Tabanidae) in the Afrotropics and the discovery of a new species from South Africa.
Genevieve Theron	New records of endosymbiotic bacteria in spider flies (Diptera: Acroceridae).

John Midgley	New records and species of hover fly (Diptera: Syrphidae) shines a light on the forest canopy habitat.
Cassandra Barker	Integrative approach to species delimitation of the South African Bombyliidae genus *Corsomyza* Wiedemann, 1820.
Arjan Engelen	Research proposal: Sexual dimorphism in fly visual function and its influence on flower visiting behaviour and floral evolution in Cape daisies.
Burgert Muller	Revision of Afrotropical *Suragina* Walker, 1859 (Diptera, Athericidae).
Kevin Malod	Past and current temperatures affect tephritid fruit fly movements: will climate change favour range expansion of pest species?
Louise Eypert	Exploring and characterizing factors influencing cucurbit-fruit fly damages and diversity in vegetable crop production systems in Cote d'Ivoire.
Rigardt van Rooyen	Seasonal population dynamics and fruit damage of *Dacus ciliatus* in commercial butternut plantations.
John-Henry Daneel	Ecology and thermal physiology of *Ceratitis quilicii* and *Ceratitis rosa* (Diptera: Tephritidae).
Nombasa Qangule	Organic, food-grade hydrophobic coatings and silicon suppress oviposition into mangos by oriental fruit flies, *Bactrocera dorsalis* (Hendel) (Diptera: Tephritidae) in laboratory trials.
Onkgopotse Seabi	Cold tolerance of fruit flies developing in different Citrus types: do microbes play a role?
Wendy Rasikhanya	Assessing the impact of rural and urban pollution on the major malaria vector *Anopheles arabiensis* Patton (Diptera: Culicidae).
Ashley Burke	Safeguarding endectocides for malaria control: strain-specific and generational responses to Ivermectin in *Anopheles funestus* s.s.
Kayla Noeth	The effect of sublethal Temephos exposure on longevity and the gut microbial composition of *Anopheles arabiensis* (Diptera: Culicidae).
Kurt Jordaens	Syrphidae and the diversity of pollinating Diptera in Afrotropical biodiversity hotspots.
Burgert Muller	Invasion patterns and niche comparison of the hoverfly *Toxomerus floralis* (Diptera: Syrphidae) between its native and non-native range.

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Dipterists Society 34th General Meeting at ESA Wrap-up!

Matt Bertone

Plant Disease and Insect Clinic, North Carolina State University,
Raleigh, North Carolina 27606, USA; maberto2@ncsu.edu

The Dipterists Society 34th General Meeting at ESA, held at the 2025 Annual Meeting of the Entomological Society of America (Portland, Oregon, USA; November 11 from 7-9pm), was a success! Other than a few technical difficulties encountered (mostly caused by the host, me) it was a fun and educational session. The entire program and links to the recorded portions can be found at the end of this article.

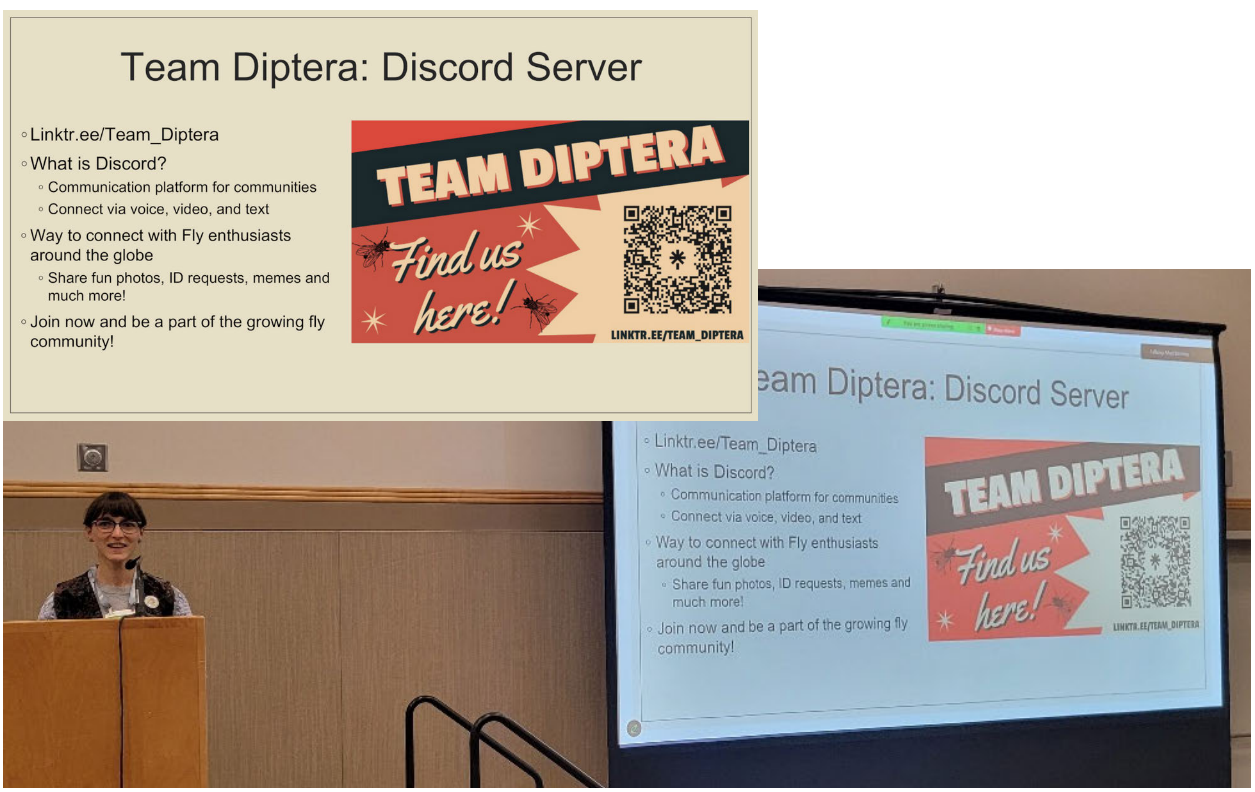
The night started with refreshments and light fare accompanying a social session. Folks gathered to meet new people, catch up with dear friends and colleagues, or discuss "rigorous scientific things". Luc Leblanc was also kind enough to hand out commemorative pins he made for attendees.

Attendees enjoying refreshments and socializing. Photo by Luc Leblanc. Inset, Pin made for attendees by Luc Leblanc. Photo by Matt Bertone.

After the social session, the meeting began with a few announcements. The first was by Matt Petersen (University of Minnesota), who discussed the upcoming Field Meeting (https://dipterists.org/field_meetings.html) to be held at the Itasca Biological Station and Laboratories in northern Minnesota (June 15-19, 2026). We hope to see you there!

Matt Petersen (University of Minnesota) discussing the next Field Meeting. Photo by Luc Leblanc.

Next was Brittany Kohler (University of California, Davis) who shared information on how to join other fly enthusiasts on Team Diptera. She highlighted one way to connect with the group through their discord (see image below for details).



Brittany Kohler (UC Davis) discussing the Team Diptera discord. Photo by Luc Leblanc. Inset: Team Diptera announcement.

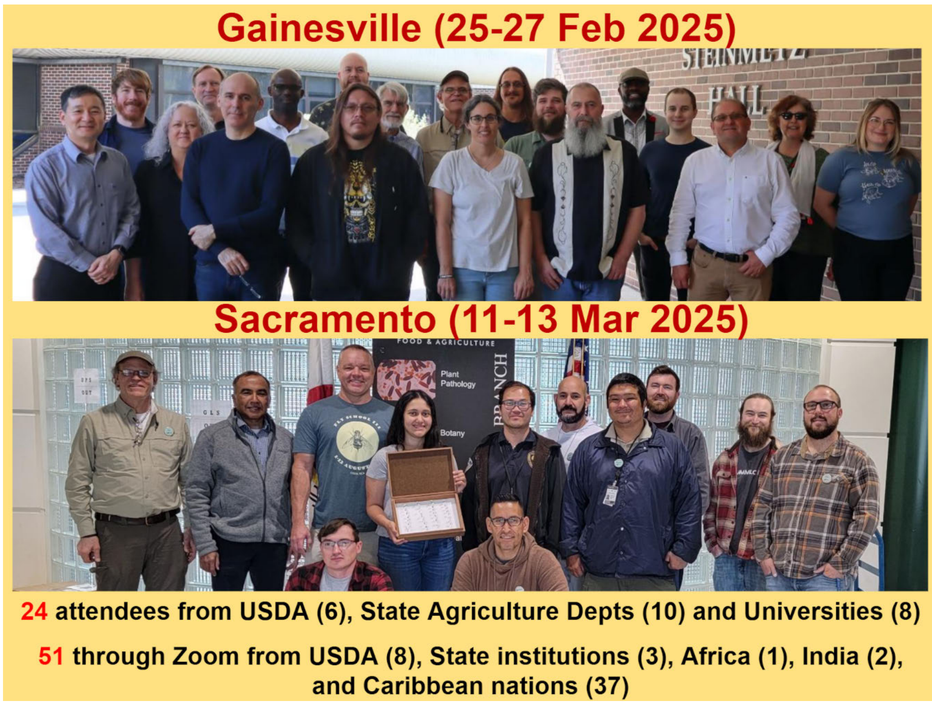
Brian Brown (Natural History Museum of Los Angeles County, emeritus) announced Fly School IV, to be held at the Soltis Center in Costa Rica, July 27th through August 9th, 2026. This two-week course dives deep into Diptera to train those interested in fly biology, ecology, and identification. For more information, please look for updates and ways to apply on the Fly School website (https://dipterists.org/fly_school.html).

Brian Brown (Natural History Museum of Los Angeles County) announcing the next Fly School. Photo by Luc Leblanc.

The next announcement was about a new journal, presented by John Stireman (Wright State University). The journal, *Journal of Lost Species*, aims to document species that are at risk of extinction, those that have been lost to extinction, or those found after being considered gone. For more information, please see the journal's website (https://journaloflostspecies.org/index.php/jls).

John Stireman (Wright State University) announcing a new journal, Journal of Lost Species. Photo by Luc Leblanc.

The final announcement was by Luc Leblanc (University of Idaho) regarding highlights from the USDA-funded 2025 Tephritidae Identification Course. Luc walked us through the program for the course showing familiar people and flies, learning or teaching various techniques for preparing specimens and identifying fruit flies. The course will be held again in 2027 in Texas.

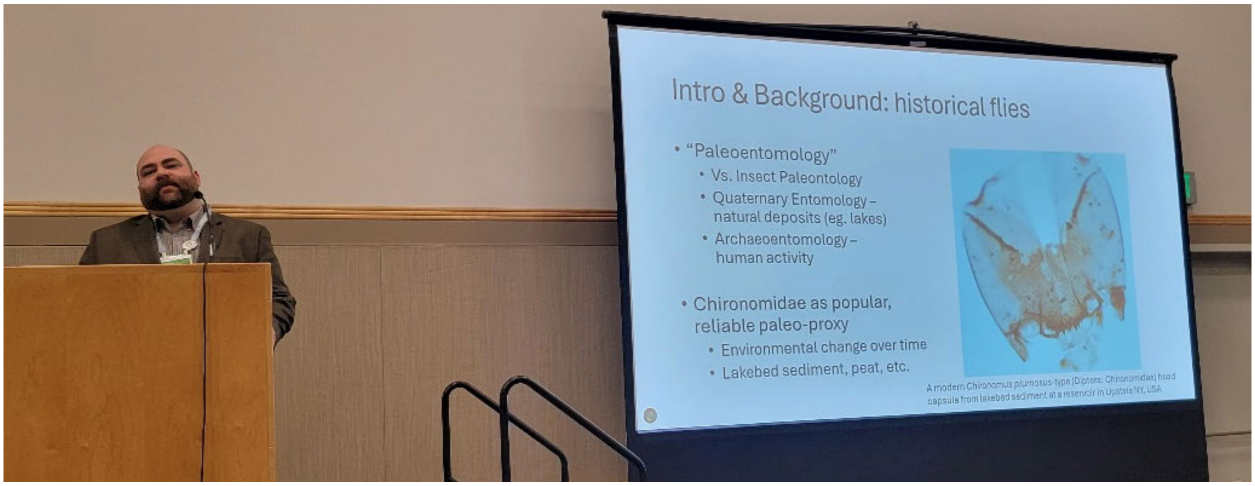


Slide from Luc Leblanc's (University of Idaho) announcement and discussion of the Tephritidae Course.

After the announcements we heard presentations from several speakers. First up was Riley Hoffman (UC Davis) who presented *Blow Flies Under the Lens: Forensic DNA Retrieval Post-ESEM*. Riley discussed her work recovering genetic material from larval blow flies that had been used for ESEM imaging.

Riley Hoffman (University of California, Davis) presenting on her work recovering blow fly DNA from specimens after ESEM imaging. Photo by Luc Leblanc.

We then heard from Michael Monzon (Purdue University) about his work on flies at two archaeological sites in the US (Archaeological Dipterology: Flies as Evidence from Past Civilizations). These sites were buildings from the 1600s and 1800s (respectively), both with a rich history where the fly and other insect specimens added important information.



Michael Monzon (Purdue University) discussing his work at two archaeological sites in the US. Photo by Luc Leblanc.

Next we a pre-recorded presentation by Chan Tsz Ying (Elaine) (The University of Hong Kong) on the biodiversity of Chironomidae in Hong Kong. This presentation was filled with exciting finds and new discoveries showing how little we know about midges in Hong Kong. Keep an eye out for her work!

Title slide for the first recorded talk shown during the meeting.

After that, we heard a recorded presentation by Jean de Dieu Nsenganeza (University of Rwanda/UR-CoEB) titled *Distribution of Flies (Insecta: Diptera) Across Altitudinal Gradient in Nyungwe National Park, South-Western Rwanda*. Jean discussed his work to identify fly communities found across altitudinal gradients in Rwanda, finding patterns in their diversity.

Title slide for the second recorded talk shown during the meeting.

Finally, I closed out the meeting with a photo presentation showing some of the amazing flies I saw in Costa Rica during Fly School III, as well as interesting flies I found in other locations. If you would like to see my photos, feel free to visit my Flickr page (https://www.flickr.com/photos/76790273@N07/), or for only the Fly School III photos you can find them in their own album (https://flic.kr/s/aHBqjBFxWq).

Matt Bertone showing photos of flies from Fly School III and other places. Photo by Luc Leblanc.

I am grateful for the help organizing and moderating the meeting provided by Dr. Luc Leblanc (University of Idaho). I am also grateful for those who contributed presentations or recordings, as the meeting would not be quite as interesting without all that good fly content – thank you! Finally thank you to all the people who came to the meeting: it was attended by about 28 people in person and 15 people virtually through Zoom (total = 43 attendees).

Virtual attendees joining by Zoom. Photos by Luc Leblanc (top) and Matt Bertone (bottom)

If you missed the meeting or have a burning desire to present in the 35th Dipterists Society General Meeting of the held at ESA, keep an eye out for announcements for next year's meeting to be held at the Annual Meeting of the Entomological Society of America in Columbus, Ohio, USA!

2025 Program:

7:00 PM – Social
Finger foods and cash bar provided; Limited free drink tickets offered

7:30 PM – Welcome & Announcements (https://youtu.be/mVu44t78atw)
Field Meeting – Matt Petersen (University of Minnesota)
Team Diptera Outreach – Brittany Kohler (UC Davis)
Fly School IV: Back to the Tropics – Brian Brown (Natural History Museum of Los Angeles County)
Tephritid Course – Luc Leblanc (University of Idaho)
Journal of Lost Species – John Stireman, Matt Forister, Hollis Woodard

8:00 PM – Blow Flies Under the Lens: Forensic DNA Retrieval Post-ESEM
Riley B. Hoffman (UC Davis) and Nicholas J. Miller (Northeastern University)
https://youtu.be/9GbScmdoWXQ

8:12 PM – Archaeological Dipterology: Flies as Evidence from Past Civilizations
Michael A. Monzon (Purdue University) and Krystal R. Hans (Purdue University)
https://youtu.be/Jywkl_zC_Gk

8:24 PM – The Biodiversity of Chironomidae (Diptera) in Hong Kong
Chan Tsz Ying (Elaine) (The University of Hong Kong)

8:36 PM – Distribution of Flies (Insecta: Diptera) Across Altitudinal Gradient in Nyungwe National Park, South-Western Rwanda
Jean de Dieu Nsenganeza (University of Rwanda/UR-CoEB)
https://youtu.be/B51OGGIxz_4

8:48 PM – Photos of Diptera from Fly School III and Beyond
Matt Bertone (North Carolina State University)
https://youtu.be/mpD3Bistimo

9:00 PM – Closing Remarks

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Opportunites and Requests

Fly School IV

Giar-Ann Kung

Dipterists Society, P.O. Box 231113, Sacramento, California, 95823, USA

Fly School will be returning to the Soltis Center (https://global.tamu.edu/soltis) in San Juan de Peñas Blancas, San Ramón, Costa Rica, from 27 July through 9 August 2026. This will be the fourth offering of the course, which is now an event of the Dipterists Society.

The two-week course is open to all who are interested, covering family-level identification, morphology, natural history, ecology, collecting methods, specimen preparation, curation, and other topics through lectures, fieldwork, and lab. On the return to San José, we plan to stop at the Costa Rica National Museum to visit the Entomology collections—their Diptera curator is an alumna of Fly School II!

The Soltis Center has an air-conditioned classroom where lectures and labs are held. Accommodations are shared cabins, each with four beds and with its own shower, sinks, and toilet. The cabins are not air-conditioned; however, they have ceiling fans and two large screen doors.

Costs are estimated to be approximately $1,500 USD, which includes $1,250 station fees (lodging and food). Participants are responsible for their own transportation to San José, Costa Rica, and any lodging before or after the course.

The application will be available in January 2026, and will be announced on the dipterists mailing list (https://lists.dipterists.org/mailman/listinfo/dipterists), the Fly School mailing list (https://groups.google.com/u/1/g/flyschool-L), and will be posted on the Fly School webpage. (https://dipterists.org/fly_school.html). Dipterists Society members will also be eligible to apply for Dipterists Society grants if selected to attend.

If you have any questions, feel free to email me at dipteracourse@gmail.com.

We hope that you will join us!

If you would like to support Fly School, consider donating to the Dipterists Society (https://dipterists.org/support.html)! You may earmark a donation, or any part of a donation, to Fly School in the “Other Information” field.

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Call for grant applications from the Dipterists Society!

Stephen D. Gaimari

Dipterists Society, P.O. Box 231113,
Sacramento, California, 95823, USA; sgaimari@dipterists.org

The Dipterists Society is here announcing the first grant competition for 2026!

From 2022 to date, we have awarded 36 grants to dipterists from 17 different countries, totaling more than $30,000 across five grant competitions for research, travel, and educational opportunities. Our intention moving forward is to run two standard grant competitions per year, subject to available funding, with their announcements and details given in the *Fly Times* (June and December) each year, and with application deadlines in the following August and February.

Guidelines and details for each competition will be given in the announcement, but generally speaking our grants program is divided into "travel" grants (e.g., meetings, educational opportunities) and "research" grants (e.g., visiting museums/collections, doing field work), and the funded activities must be specific to dipterology. Note, travel as part of a research project would qualify for a research grant, not a travel grant.

From time-to-time, we may also run special grant competitions for particular events that may fall outside of our regular schedule or for a specific purpose. For example, we may have a closed grant competition only for those individuals selected to attend Fly School, which will be announced once the list of attendees is finalized.

To be eligible to apply for Dipterists Society grants, you must be a member-in-good-standing, with dues up to date. (https://dipterists.org/membership.html)

Current call for grant applications for Diptera-specific travel or research!

The Dipterists Society here announces the opening of a grants competition to support **TRAVEL** or **RESEARCH** activities. All requested elements making up a full proposal must be submitted for full consideration. Please specify in your application whether yours is for Travel or Research.

The Dipterists Society expects to be able to grant a total of US$10,000 in this round, with awards typically not exceeding **$2000 (for research grants)** or **$1000 (for travel grants)** per successful applicant.

Proposals should be submitted as a combined PDF file to grants@dipterists.org no later than 9 February 2026.

Final decisions and notifications to successful applicants can be expected by early March 2026.

Successful applicants must use the awarded funds within one year of the award under normal circumstances, with short extensions to be requested and considered by the Society's Board of Directors.

As general guidance, here is some relevant information to help you develop your proposal. If you have questions about eligible vs. ineligible expenses, please contact us at grants@dipterists.org.

Examples of eligible expenses

i. travel costs
ii. per diem (food, lodging) for the applicant
iii. registration costs [meetings, educational opportunities]
iv. bench fees
v. purchase of necessary equipment and supplies [Research grants]
vi. field work expenses [Research grants]

Examples of ineligible expenses

i. salaries
ii. travel costs or per diem for additional people
iii. costs related to DNA sequencing [Research grants]
(costs related to acquiring specimens for DNA work is acceptable)

Application procedure

Following are the elements requested, to be submitted as a single combined PDF.

(a) Applicant information, including name, academic level, institution (if applicable), mailing address, email address, and phone number.
(b) Itemized budget in support of the proposal, including any expenses for which this grant funding will be used (e.g., anticipated costs for transportation, per diem, supplies, etc.).
(c) Budget justification, per line item.
(d) Current *Curriculum vitae*.
(e) **If a student**, a letter from an academic advisor, or other faculty member, confirming relevance of the proposed activity or travel.
(f) **If a Research Grant**, use the following guidelines:
  i. What are the aims of the proposed activity? [*maximum 300 words*]
  ii. What is already known about the study system? [*maximum 300 words*]
  iii. What is the plan of action, including schedule and approach? [*maximum 300 words*]
  iv. What will be the benefit to dipterology? [*maximum 300 words*]
  v. References cited. [*maximum 10*]
(g) **If a Travel Grant**, provide a brief account describing the benefit of the meeting or educational opportunity for the applicant (for a meeting, state proposed participation in the meeting, e.g., presenting a talk or talks, poster, running a symposium, etc.), including how funding from the Dipterists Society will help accomplish their goals for participating. [*maximum 750 words*]

A few things to note.

- This is a good opportunity to apply for funds to attend the next Field Meeting of the Dipterists Society! (see the announcement in this issue of *Fly Times*)
- This grant competition does not include funding for Fly School IV. There will be a separate call for grants from among the final list of attendees.
- This grant competition does not include funding to attend ICD 11 (too far out), but note that there will be two opportunities to apply – one in the next round to be announced in the *Fly Times* issue 76, and then the round to be announced in *Fly Times* issue 77.
- See https://dipterists.org/grants_awards.html for more information.

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21st Arbovirus Surveillance and Mosquito Control Workshop

Rui-De Xue

Anastasia Mosquito Control District, 120 EOC Drive,
St. Augustine, Florida 32092, USA; xueamcd@gmail.com

The 21st Arbovirus Surveillance and Mosquito Control Workshop will be held in Anastasia Mosquito Control District (AMCD), 120 EOC Drive, St. Augustine, FL, March 4–6, 2026 after the Florida Mosquito Control Association's Fly In class, AMCD, March 2-3, 2026. For more information about the workshop and the Fly In class, please visit the website at www.amcdsjc.org or contact Dr. Rudy Xue at rxue@amcdfl.org

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DIPTERA BLOOPERS

Those pesky mythicosquitoes!

Submitted by Steve Gaimari

popsci.com/environment/natural-mosquito-trap-fungus/

Systema Diptero... Dipterists Society All Bookmarks

POPULAR SCIENCE

ENVIRONMENT / ANIMALS / INSECTS

This mosquito death trap is all-natural and very deadly

The power of flowers and fungi is no match for these insects.

LAURA BAISAS / PUBLISHED OCT 31, 2025 1:33 PM EDT

Flowers provide mosquitoes with nectar, a critical food source. Photo by Marianna Armata via Getty Images

Never underestimate the power of fungi. It can turn ants into "zombies," help fictional plumbers grow, and even look like creepy fingers. One newly engineered strain of fungus uses the power of smell to kill Earth's deadliest animal—mosquitoes.

Mosquito-borne diseases, including malaria and dengue, kill thousands of people per year. In 2023 alone, malaria killed over 500,000 people in 83 countries. These illnesses are often difficult to control, and mosquitoes have gotten better at resisting chemical pesticides that used to work.

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DIPTERA ARE AMAZING!

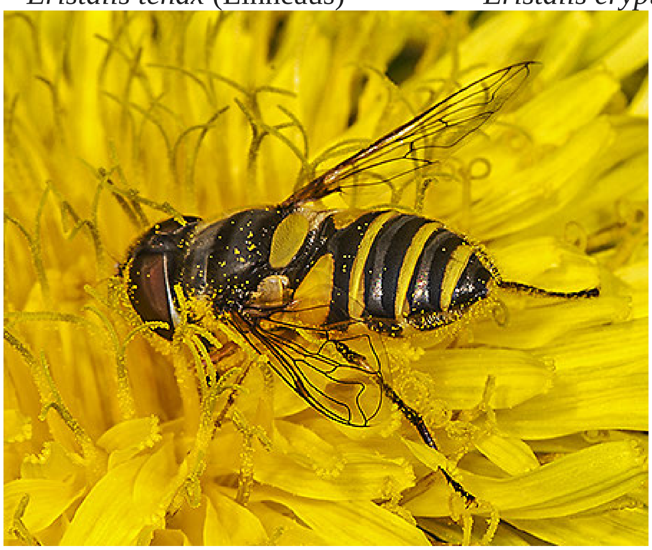
A female *Leptopeza antennalis* Melander (Hybotidae) photographed *in situ* near The Purchase in Great Smoky Mountains National Park, NC, USA. Until recently, this species was only known from specimens collected in the early- to mid-20th century. As a summer intern for Discover Life in America this year, one of my goals was to "rediscover" this species and collect new specimens from the Smoky Mountains. Success! Photograph by Zachary Dankowicz.

A pair of *Ochthera anatolikos* Clausen (Ephydridae) *in copula*, photographed on a sandbar of the Little River at the intersection of the Foothills Parkway and the Old Walland Highway in Tennessee, USA. This locality (and the entirety of the Foothills Parkway) is part of the Great Smoky Mountains National Park, and has some of the most interesting flora, fauna, and geology in the park. Photograph by Zachary Dankowicz.

Next is a series of local Syrphidae photographed by Anthony Thomas in New Brunswick Canada.

Eristalis tenax (Linneaus)

Eristalis cryptarum (Fabricius)



Eristalis transversa (Wiedemann)

Eristalis sp.

Sericomyia militaris (Walker)

Sphaerophoria sp. (*philanthus*-complex)

Spilomyia fusca Loew

Xylota sp.

Editor's Note:
In the previous issue of *Fly Times* (#74), in the Diptera Are Amazing section on page 59, the photograph labeled as a male *Diastracus prasinus* was miscaptioned. The correct caption for that photo is:

Male *Nepalomyia nigricornis* (V.D.) (Dolichopodidae), photographed at the Dark Hollow Falls in Shenandoah National Park, Virginia, USA. This species is ubiquitous in the Appalachians but individuals are extremely rarely photographed by amateur naturalists due to their minute size and restricted habitat, as they are only found in "shaded habitats in which a thin film of water is running over moss-covered rocks" (Runyon & Hurley 2003, revision, p. 410). Photograph by Zachary Dankowicz.

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BOOKS AND PUBLICATIONS

The resurrection of *Myia*, and an index to volume 12

Stephen D. Gaimari

Dipterists Society, P.O. Box 231113,
Sacramento, California, 95823, USA; sgaimari@dipterists.org

The journal *Myia* became a publication of the North American Dipterists Society starting with volume 9. Volumes 1–6, and the incomplete volume 7, were produced and distributed by Paul Arnaud, Jr., supported in part by the California Academy of Sciences in San Francisco. (There was no volume 8). The topics were rather broad, from biographical or autobiographical accounts, to type catalogs, and to more standard series of individual scientific papers. The list of the contents for volumes 1–7 (but unfortunately not most of the PDFs, yet!) can be seen on the *Myia* webpage (https://dipterists.org/myia.html).

The volumes published after transfer to the North American Dipterists Society are listed, along with PDFs for most of their contents, on the "Issues" tab of the *Myia* webpage. Volume 9–11 were published in 1998, 1999, and 2001, respectively, all by Bachuys in Leiden. The focus of volume 9 was a systematic database of tephritid fruit fly names, by Allen Norrbom and colleagues. Volume 10 was an annotated catalog and bibliography of family-group names in Diptera, by Curt Sabrosky. Volume 11 was a world catalog of Stratiomyidae, by Norm Woodley.

It was another 10 years before the next volume was published, this time by Pensoft in Sofia. Volume 12 (2011) was a series of family catalogs and a couple of biographical accounts, titled "Contributions to the *Systema Dipterorum* (Insecta: Diptera)", edited by Irina Brake and Chris Thompson. The introduction and taxonomic catalogs are all available on the *Myia* webpage as PDFs. Following are the links to the catalogs:

- Brake — Carnidae catalog, pp 113–169.
  (https://dipterists.org/assets/PDF/myia12-brake2011-carnidae.pdf)
- Mathis & McAlpine — Coelopidae catalog, pp 171–205.
  (https://dipterists.org/assets/PDF/myia12-mathis_mcalpine2011-coelopidae.pdf)
- Mathis & Sueyoshi — Dryomyzidae catalog, pp 207–233.
  (https://dipterists.org/assets/PDF/myia12-mathis_sueyoshi2011-dryomyzidae.pdf)
- Mathis & Barraclough — Diastatidae catalog, pp 235–266.
  (https://dipterists.org/assets/PDF/myia12-mathis_barraclough2011-diastatidae.pdf)
- Mathis — Helcomyzidae catalog, pp 267–280.
  (https://dipterists.org/assets/PDF/myia12-mathis2011-helcomyzidae.pdf)
- Mathis — Heterocheilidae catalog, pp 281–289.
  (https://dipterists.org/assets/PDF/myia12-mathis2011-heterocheilidae.pdf)
- Gaimari & Mathis — Odiniidae catalog, pp 291–339.
  (https://dipterists.org/assets/PDF/myia12-gaimari_mathis2011-odiniidae.pdf)
- Mathis & Rung — Periscelididae catalog, pp 341–377.
  (https://dipterists.org/assets/PDF/myia12-mathis_rung2011-periscelididae.pdf)

- Woodley — Supplement to Stratiomyidae catalog, pp 379–415. (https://dipterists.org/assets/PDF/myia12-woodley2011-stratiomyidae-supplement.pdf)
- Woodley — Xylomyidae catalog, pp 417–453. (https://dipterists.org/assets/PDF/myia12-woodley2011-xylomyidae.pdf)
- Woodley — Xylophagidae catalog, pp 455–500. (https://dipterists.org/assets/PDF/myia12-woodley2011-xylophagidae.pdf)

Although an excellent series of catalogs, one thing that was lacking in the volume was an index. So Neal Evenhuis took on this challenge and produced a taxonomic index, which has has made available as a PDF (https://dipterists.org/assets/PDF/myia12-evenhuis2025-index.pdf). Thank you to Neal for making this resource available to everyone!

After an even longer hiatus, closing in on 15 years, the Dipterists Society is planning to again resurrect *Myia*! So this is an announcement for a slow relaunch, with more details to come. We have had several inquiries from dipterists wanting to publish larger catalogs, which is among the topic areas that would be appropriate for *Myia*. The plan is for *Myia* to be peer-reviewed, and for its publication to be in accordance with the rules of the International Code of Zoological Nomenclature. Publication could come as single large works (as in volume 11) or as series of articles (as in volume 12), depending on submissions. So start thinking about the kinds of works you might like to publish in *Myia*! Stay tuned…

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SOCIETY BUSINESS

On the back pages of *Fly Times*, Dipterists Society business is recorded, as is desired for Society transparency.

Several documents are here provided on the following pages for the record. They are:

1. Current list of Directors, Officers, and Auxiliary Officers (1 page)
2. Approved minutes of the 1st Quarterly meeting of the Board of Directors, held 16 April 2025
3. Approved minutes of the 2nd Quarterly meeting of the Board of Directors, held 01 July 2025

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As of this writing, following are the Directors, Officers, and Auxiliary Officers of the Society. This list is also presented in the Society's webpage on governance (https://dipterists.org/governance.html)

Directors

Christopher Borkent
Neal Evenhuis
Stephen Gaimari
Martin Hauser
Ashley Kirk-Spriggs
Giar-Ann Kung
Erica McAlister
John Midgley
Sarah Oliveira
Thomas Pape

Officers

Stephen Gaimari, President
Martin Hauser, Vice President
John Midgley, Secretary
Giar-Ann Kung, Treasurer, Education Chairperson
Christopher Borkent, Assistant Treasurer
Matthew Bertone, Meeting Chairperson
Ashley Kirk-Spriggs, ICD Chairperson

Auxiliary Officers

Dalton Amorim, ICD Councilor
Yuchen Ang, ICD Councilor
Daniel Bickel, ICD Councilor (Vice Chair)
Will Bouchard, Field Meeting Co-Chair
Xiaolin Chen, ICD Councilor
Netta Dorchin, ICD Councilor
Stephen Gaimari, ICD Councilor
Valery Korneyev, ICD Councilor
Luc Leblanc, General Meeting Co-Chair
Kazuhiro Masunaga, ICD Councilor
Erica McAlister, ICD Councilor
Ximo Mengual, ICD Councilor
John Midgley, ICD Councilor (Secretary)
Sarah Oliveira, ICD Councilor
Matt Petersen, Field Meeting Co-Chair
Jeffrey Skevington, ICD Councilor

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Dipterists Society
DIRECTOR'S MEMO

Minutes of Directors Meeting

Prepared and filed 22 May 2025 by John Midgley, Secretary

Notice for this Quarterly Meeting was given by Steve Gaimari by email to all Directors on 10 April 2025, after polling all for availability.

Meeting held Tuesday, 16 April 2025, call to order at 10:00 PM (SAST) using Microsoft Teams.

Presiding: Stephen Gaimari
Secretary: John Midgley

Attendance: Virtual: Stephen Gaimari (Director/President) (SG), Martin Hauser (Director/Vice President) (MH), Neal Evenhuis (Director) (NE), Ashley Kirk-Spriggs (Director) (AKS) Giar-Ann Kung (Director) (GAK), John Midgley (Director) (JM), Sarah Oliveira (Director) (SO), Thomas Pape (Director) (TP).

Apologies: Christopher Borkent (Director) (CB), Erica McAlister (Director) (EM).

Call to order:
The President welcomed everyone to the meeting.

Item 1. Approval of minutes of the Board meeting of 10 December 2024 (distributed 10 April 2025).
No corrections were made. **NE moved to approve the minutes** (seconded MH, passed unanimously).

Item 2. Old business

- **Membership**
  SG summarized the membership, we have 157 members, most renew in a timely manner. Seven new members joined so far in 2025. Currently, there are 247 people in the Dipterists Directory, 881 on the mailing list and 1800 followers on Facebook. Our membership does not include everyone interested in Diptera, so we should think of ways to encourage them to join.

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- **Grants Task Force recommendation (distributed 10 January 2025, redistributed 10 April 2025)**
  At the previous meeting, SG, EM and TP were tasked to develop a document to guide funding calls made by the Society. This was distributed to the Directors ahead of the meeting. Comments were received from AKS via email:
  (1) that we should be clear about what any given call for proposals aims to fund. SG referred the meeting to Item (3)(b), where this is already covered. After the discussion, it was agreed that the guidance document has sufficient detail and more detail should be included in a call when it goes out, to allow each call to be tailored to the current goals of the Society. MH also said that this allows us to be flexible and fund areas of research that are underfunded.
  (2) We should be clear that there is no geographic bias. Adding ", worldwide" to item (2) was agreed as a solution to this. In specific calls, this should be reiterated.
  (3) to give preference to conference travel where the applicant is (a) convening a symposium, (b) giving an oral presentation, and (c) giving a poster presentation. JM suggested that other criteria may also be considered, such as prioritizing students, early career researchers or dipterists from lower and lower-middle income countries. SG suggested that the criteria should be decided by the reviewers on a call-by-call basis. **AKS moved to accept the recommendation as amended** (without any stated preferences) **to finalize a document guiding our grants program** (Seconded MH, passed unanimously).

- **CICD merger (3 documents distributed 10 April 2025)**
  A subcommittee of the CICD and DS has met to discuss the possible merger. The details are in the previous minutes. The subcommittee recommended merging the organizations, and SG, AKS and JM have developed a formal merger framework (distributed to the Directors on 10 April 2025). CICD met on 8 April 2025 to discuss the merger, but as there was no quorum, they needed to get email votes to finalize their decision. For a merger to proceed, both organizations must vote in favor. **NE moved that the Council for International Congresses of Dipterology formally merges entirely with the Dipterists Society and functions forthwith according to the Society's amended Bylaws and structure** (TP seconded, passed unanimously).

  Furthermore, actions need to be taken by the Board to establish CICD as set out in the vote above. After discussion, **TP moved that, effective only if and when the CICD vote on the merger passes, the Board of Directors takes the following actions. The Board creates a new Officer position titled ICD**

Chairperson (short for "International Congresses of Dipterology Chairperson") and a new Auxiliary Officer position titled ICD Councilor (short for "International Congresses of Dipterology Councilor"). Two members of the new Auxiliary Officer class ICD Councilor shall each have a special parenthetical title, namely, "ICD Councilor (Vice Chair)" and "ICD Councilor (Secretary)". Further, the Board establishes an Executive Committee of fifteen (15) people, titled "Council for International Congresses of Dipterology" (shortened to Council). All fifteen (15) members are voting members, and all are in the Officer and Auxiliary Officer positions previously named. Should fewer than two (2) members of this 15-person Council be concurrently Dipterists Society Directors, up to two (2) Directors shall be selected by the Board as nonvoting members of Council to supplement the Executive Committee in order to conform to Executive Committee rules. These actions are allowed under Society Bylaws Article 5 Sections 1 and 3 and Article 6 Section 1. To provide background, guidance, and definitions to this Executive Committee, the language of the formal proposal will be used to finalize such a document (seconded MH, passed unanimously).

TP moved that, to promote continuity of the Council, effective only if and when the CICD vote on the merger passes, we hereby appoint the following current Councilors to the created Officer and Auxiliary Officer positions, along with their current terms of service.

Ashley Kirk-Spriggs – ICD Chairperson, current term ending 2027
Daniel Bickel – ICD Councilor (Vice Chair), current term ending 2027
John Midgley – ICD Councilor (Secretary), current term ending 2031

The following ICD Councilors with terms ending in 2027

Dalton Amorim
Xiaolin Chen
Netta Dorchin
Stephen Gaimari
Jeffrey Skevington

The following ICD Councilors with terms ending in 2031

Yuchen Ang
Jessica Gillung
Valery Korneyev
Kazuhiro Masunaga
Erica McAlister
Ximo Mengual

Sarah Oliveira
(seconded NE, passed unanimously)

Education updates
GAK has booked and signed a contract with the Soltis Center for Fly School IV for 27 July to 9 August 2026.

Currently JM teaches (with others, including AKS) short courses in Africa. Funding for these is secure until 2027 but after this we may consider running a course similar to Fly School in Africa. For African students, these courses should be funded or they will struggle to travel and attend the events. SG said that an advantage of being associated with the Dipterists Society is that the society can help source funding.

- **Meetings updates**
  Meeting at ESA – Matt Bertone is organizing the meeting in November and will run it with help from Luc Leblanc.

  Meeting at ESSA – JM is organizing a meeting with help from Burgert Muller. There will be 23 talks, filling a day at the congress. Because it fills a day, there will not be time afterwards for a reception. Talks in Taxonomy, Agricultural Entomology, Biological Control, general Ecology and Biodiversity. A stall and putting pens into the conference pack will cost $500, which is not good use of money. A better option is to put promotional material in the session, which will be free. We can have unedited footage for the YouTube channel for $200 or edited footage for $500. AKS suggested a pull up banner advertising the society, which JM can produce at the KZN Museum. JM requested $650 for edited video and promotional material[1]. **NE moved to approve the requested budget** (seconded GAK, seven in favor, one abstention)

  We should try to have events at other congresses. AKS suggested an advert for society newsletters. SG and MH sent a letter to AK Diptera in Germany which was circulated to the members. AKS suggested a standardized document. NE suggested an advert for Diptera journals as well. MH supported the standardized letter. SG volunteered to drive the process with JM's support, aiming to put something in the ESSA newsletter first. NE suggested reaching out to the Lorquin Society. GAK will engage with the Lorquin Society to promote the

[1] SO left the meeting at this time (22:38 SAST). Her further votes are recorded as abstentions.

Dipterists Society. MH suggest the UC Davis Entomological Club. AKS suggested the UK Amateur Entomologists Society. **NE moved to allow for using society funds to promote the society through printed and electronic materials in journals and other resources with fees no more than $200 without further board approval** (seconded GAK, seven in favor, one abstention).

We should all consider options for field meetings. For the North American field meeting, two options are under consideration, one is Allerton Park in central Illinois and the other is the Itasca Field Station in Minnesota. Matt Bertone will make the decision and refer it to the board.

- **Engaging other societies (1 document distributed 11 April 2025)**
  Discussed under the previous point.

Item 3. New business

- **Financial review**
  Since the last meeting, we have paid $6272.42 plus wire transfer fees for seven grants.
  We have paid the following bills:

Recipient	Details	Amount
Pasquesi Sheppard LLC	Tax services	$1350.00
OVHcloud	Virtual private server	$127.91
Dreamweaver	Web hosting services	$39.98
USPS	Post office box rental	$182.00
California Attorney General's Office	Charity registration	$50.00

Our tax return has been filed on time.

Currently, our bank account balance is $55,154.61 and our gifting account balance is $47,826.84. This account has lost about 4% of its value, which is above the performance of most metrics such as S&P 500.

The payment of a grant to a Russian student was suspended by the US Treasury Office of Foreign Assets Control. The individual was not on any sanction list and the bank did not indicate that there would be a problem sending money to Russia. In future, we will apply for a license from OFAC to make these kinds of payments.

- **Treasurer updates**
  GAK is not yet fully functional as the Treasurer, as the bank has not approved her as a signatory. Part of the problem is that the bank is tied to California, and moving to a US wide bank may help going forward. At this point, no decision has been made and the Board will be updated if anything changes. This does mean that in future, the treasurer will need to be based in the USA.

  CB still has access to telephone banking for check deposits, as the previous treasurer. This is a convenient arrangement, and appointing CB as Assistant Treasurer will allow this to continue. This would be his only role. **TP moved to appoint CB as Assistant Treasurer** (seconded GAK, seven in favor, one abstention).

- **Grants**
  Agreed to table this for discussion at the next meeting.

- **Email aliases**
  There have been several issues with the email alias not forwarding emails to everyone. These were meant to make communication easier, but this is not the case. **NE moved to drop the aliases and remove the email addresses from the list of directors** (seconded TP, seven in favor, one abstention).

Item 4. New business (from the floor)
GAK has set up a Bluesky account, but has not posted anything. Perhaps we need a Social Media officer. TP suggested continuing the discussion at the next meeting.

Item 5. Date of next Directors meeting
AKS moved to have the next meeting in the last week of June or first week of July 2025, the final date to be decided closer to the time (seconded TP, seven in favor, one abstention).

Item 6. Adjourn
GAK proposed to adjourn the meeting (seconded MH, seven in favor, one abstention).
Meeting adjourned at 11:15 PM SAST.

Submitted by:
John Midgley
Secretary

Dipterists Society
DIRECTOR'S MEMO

Minutes of Directors Meeting

Prepared and filed 01 October 2025 by John Midgley, Secretary

Notice for this Quarterly Meeting was given by John Midgley by email to all Directors on 06 June 2025, after polling all for availability.

Meeting held Tuesday, 01 July 2025, call to order at 10:00 PM (SAST) using Microsoft Teams.

Presiding: Stephen Gaimari
Secretary: John Midgley

Attendance: Virtual: Stephen Gaimari (Director/President) (SG), Martin Hauser (Director/Vice President) (MH), Christopher Borkent (Director/Assistant Treasurer) (CB), Neal Evenhuis (Director) (NE), Ashley Kirk-Spriggs (Director/ICD Chairperson) (AKS), Giar-Ann Kung (Director/Treasurer, Education Chairperson) (GAK), Erica McAlister (Director) (EM), John Midgley (Director/Secretary) (JM), Thomas Pape (Director) (TP).

Apologies: Sarah Oliveira (Director) (SO).

Call to order:
The President welcomed everyone to the meeting.

Item 1. Approval of minutes of the Board meeting of 16 April 2025 (distributed 26 June 2025).
No corrections were made. **AKS moved to approve the minutes** (seconded MH, passed unanimously).

Item 2. Old business

- **Membership**
  SG summarized the membership, we have 157 members, most renew in a timely manner. One new member joined since our previous meeting. Members are renewing on time for the most part, with 24 membership payments, three renewals were paid at the Founding Member level, five renewals were student memberships, three regular member renewals gave a little extra, and we had

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only one new membership, a student. On 28 April SG sent out emails to members whose memberships were in arrears or renewal was due soon. So far 36 of them have not yet renewed, and we have another nine that were due in June to write reminders to.

SG raised the fact that we have had the Founding Member category available for almost five years. As the Society is becoming more established, this category seems to no longer apply, but the additional income from this higher level is welcome. We should consider how we phase this out, either by limiting the total number of memberships available in this category or setting a cut off date. The category could be replaced with Sustaining Member at the same rate.

After a discussion about the potential value of Sustaining Membership, the final decision was deferred to the next meeting.

- **Grants Program**
  SG noted that the grants program guidance document was now complete. This document should be viewed as a first version and can be amended if it is felt to not be serving its purpose.

  SG asked for opinions on running a grant program in the near future. MH noted the value of meeting grants to spread awareness about the society. While ad hoc grants can allow flexibility for members to attend many meetings, the funding program has been designed for competitive grants. Opening calls twice a year was suggested as a good starting point, as it aligns well with placing calls in *Fly Times*. EM suggested that the funding deadlines could be in February and August, giving applicants about six weeks to finalize applications. **TP moved to have two combined calls every year, so that announcements can be sent out in *Fly Times*. EM added that the deadlines should be in February and August to coincide with the announcements** (seconded MH, passed unanimously).

- **CICD Executive Committee**
  The merger of CICD into the Dipterists Society was officially completed on 8 May 2025. Two documents were circulated: the defining document for the executive committee and the congress organization guide.

- **Education updates**

GAK stated that no further progress had been made on Fly School IV as planning was ahead of schedule. EM suggested that the Society could consider funding a student for the Dipterists Forum, but after discussion, it was agreed that this should be part of the grants program.

- **Meetings updates**

  The report from Matt Bertone was circulated before the meeting and was accepted by the meeting.

  NE suggested that *Fly Times* should include meeting notices. SG will include any relevant meetings if the details are shared.

  JM reported on the ESSA meeting. The Society has a complimentary table to exhibit at, where stickers will be available. All delegates will receive a flyer in their conference packs. There are 23 scheduled talks, which will be recorded and uploaded to the Society's YouTube channel.

- **Engaging other societies**

  The advert for other society newsletters is still pending.

- **Social media**

  To be discussed via email.

Item 3. New business

- **Treasurer update**
  GAK is still not registered as a signatory on the accounts, this will happen next time she visits Sacramento.

- **Financial review**
  GAK presented the review. We have taken in $1459.35, that is $1330 in membership fees, and $129.35 in donations associated with membership payments. The memberships included 3 Founding Members renewing at the full Founding level, 10 Founding Members renewing at the regular level, 8 individual members, and 8 at the student rate.

  Our expenses since the last meeting totalled $3,246.70, broken down as follows:

John M. Midgley
01 October 2025
Page 4

a) Nonprofit Insurance Alliance, annual payment for General Liability and Directors & Officers coverage - $1,600
b) Carlamani Conferences for the approved ESSA expenses - $537
c) MailmanLists, annual payment for our dipterists list server - $58.65
d) Grant payment to one of the grant recipients from the 2024 grant program who needed the check reissued - $1,000
e) bank fees - $51.05

Our current balance at California Bank & Trust is $53,347.00.

Our current investment account balance at RBC is $52,173.29, so we are up $4346.45 since the end of last quarter. Like last time, this is no surprise given markets as they are. This should pick up steam if and when things get more normal.

- **Officer nominations**
  Matt Bertone, as Meeting Chairperson, has nominated two individuals for Auxiliary Officer positions in his purview. They are read as follows:
  1) "As Meeting Chairperson, I nominate Will Bouchard of the University of Minnesota to the Auxiliary Officer position of Field Meeting Chair, for the purpose of organizing and running the next Field Meeting."
  2) "As Meeting Chairperson, I nominate Luc Leblanc of the University of Idaho to the Auxiliary Officer position of General Meeting Co-Chair, for the purpose of helping me to organize and run the next General Meeting being held at ESA."
  MH moved that we vote on these nominations (seconded AKS, passed unanimously)

- **General meetings**
  Last year, at the ESA General meeting, the Society funded a social hour with drink tickets and finger foods, after the session of talks. It was suggested that we run a similar event, and preauthorize the same amount to fund it, that is $1,500 to cover the catering and drinks**. EM moved that we preauthorize $1,500 for the event at ESA (**seconded GAK, passed unanimously).

  In the past, General Meetings were only held at ESA, but this year there will be a meeting at ESSA as well. To avoid confusion, SG suggested that General Meetings are numbered within the context of the Congress at which they are held. A General Meeting is any gathering of Dipterists that is organized or supported by the Society in some way. AKS moved that we use sequential numbers for regional General Meetings in other parts of the World (seconded TP, passed unanimously).

Item 4. New business from the floor

- **Advertising the Society**
  AKS raised that we should put more effort into advertising the society, to promote membership.

- **Partnership with Buglife**
  EM approached by Buglife to do a Year of the Fly campaign. This would involve partnering with Buglife, providing information about species and families of flies which Buglife would promote. The focus will be British species, but additional information will be provided for the families globally.

- **Social media**
  Generally, our social media is poor. There is content on YouTube, but we do not get good engagement. We should establish a position as Social Media Chairperson. This person would manage people engaging with different platforms, rather than doing the work themselves. **NE moved that we establish this position** (seconded MH, passed unanimously).

Item 5. Date of next Directors meeting
CB moved to have the next meeting in the last week of September or first week of October 2025, the final date to be decided closer to the time (seconded MH, passed unanimously).

Item 6. Adjourn
MH proposed to adjourn the meeting (seconded EM, passed unanimously).
Meeting adjourned at 11:22 PM SAST.

Submitted by:
John Midgley
Secretary

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