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Fly Times is simultaneously distributed in PDF and printed format twice yearly, with spring and fall issues.

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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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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, and hope that this will be an attractive medium for students through retirees to showcase their activities.

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

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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).
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 the middle of May and the middle of November, although this is flexible up to the time of publication (which will generally be early June for the spring issue and early 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

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The North American Dipterists Society is a 501(c)(3) nonprofit organization (EIN 84-3962057), incorporated in the state of California on 27 November 2019. We are an international society of dipterists and Diptera-enthusiasts, serving the needs of the worldwide dipterist community. Note, as of the Directors meeting held 10 December 2023, the Society has been renamed **The Dipterists Society**, with subtext **An International Society for Dipterology**. Performing the tasks that go along with such a change (updating legal documents, the website, our logo and seal, etc.) are still in progress.

> *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 resources to those who need them, 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 for the next issue, which is slated for spring of 2024. And for larger works, please consider the *Fly Times Supplement* series, found at https://dipterists.org/fly_times_supplement.html.

Also note, I am still working on improving the front and back covers of the *Fly Times.* Here is an early attempt at something new! From here forward, I will try to use one of the images from the issue, or a special image that someone sends me, for the cover. The back page may or may not stay like this, but the color is similar to recent Society publications such *Myia* and some of the *Fly Times Supplements*. Some of you clever dipterists might have good ideas for a cover – please consider submitting them! My one stipulation (besides it being dipterological!) is that it is exactly 8-1/2 X 11 inches (*Fly Times* page size). For now, I'll be changing up the covers issue to issue, so please feel free to send your design ideas to me at sgaimari@gmail.com (cc sgaimari@dipterists.org).

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NEWS AND RESEARCH

Catotricha americana (Felt) (Diptera: Cecidomyiidae: Catotrichinae) newly confirmed from Canada

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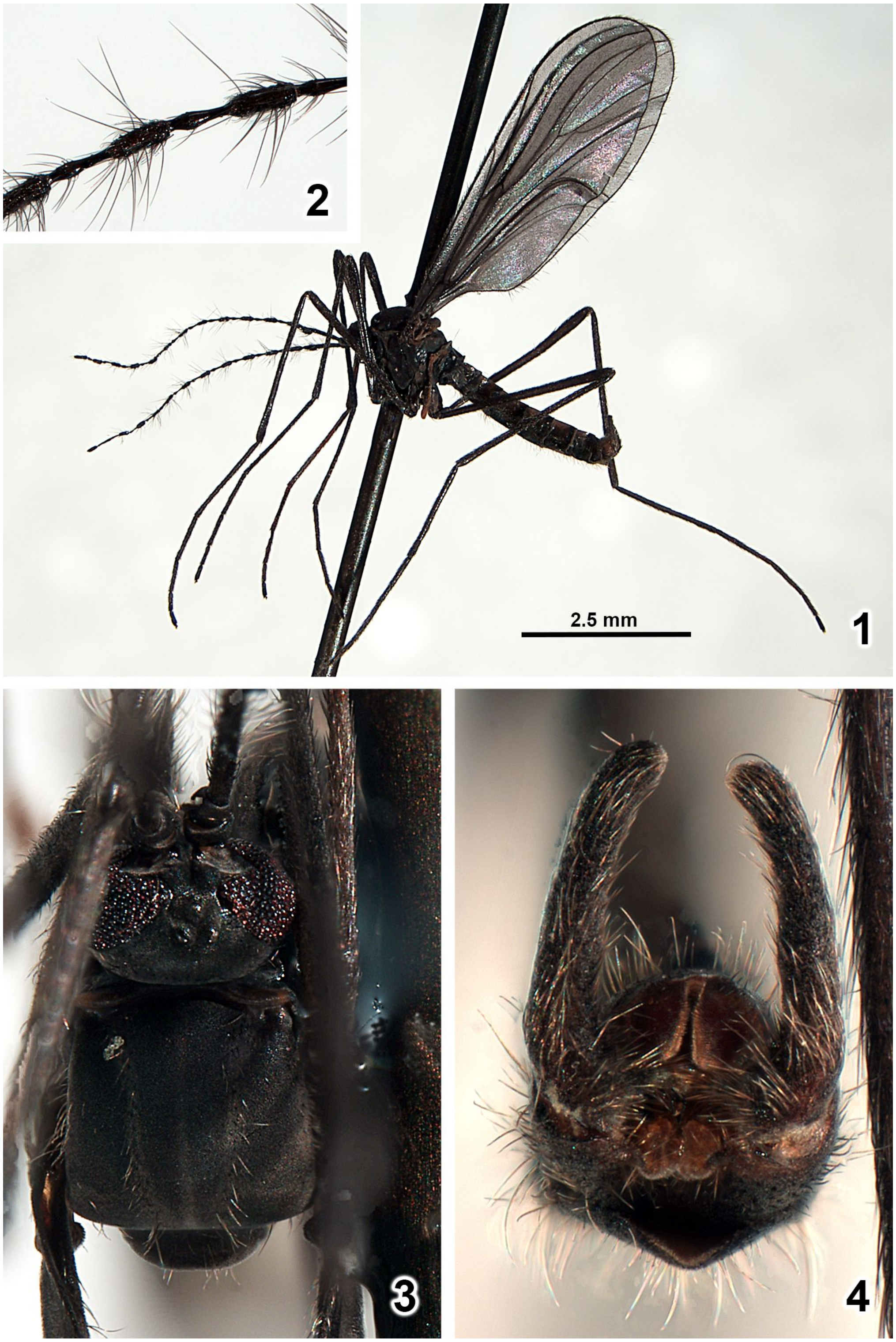
The subfamily Catotrichinae is a rarely collected group of Cecidomyiidae, with two genera and three species in North America (Jaschhof & Fitzgerald 2016). The subfamily is characterized by distinct plesiomorphic wing venation and includes eight species worldwide. Specimens have been collected in mature forests with trees of various age classes and layers of rotting leaves and wood. Larvae appear to develop in well decayed tree trunks (Jaschoff & Jaschhof 2008).

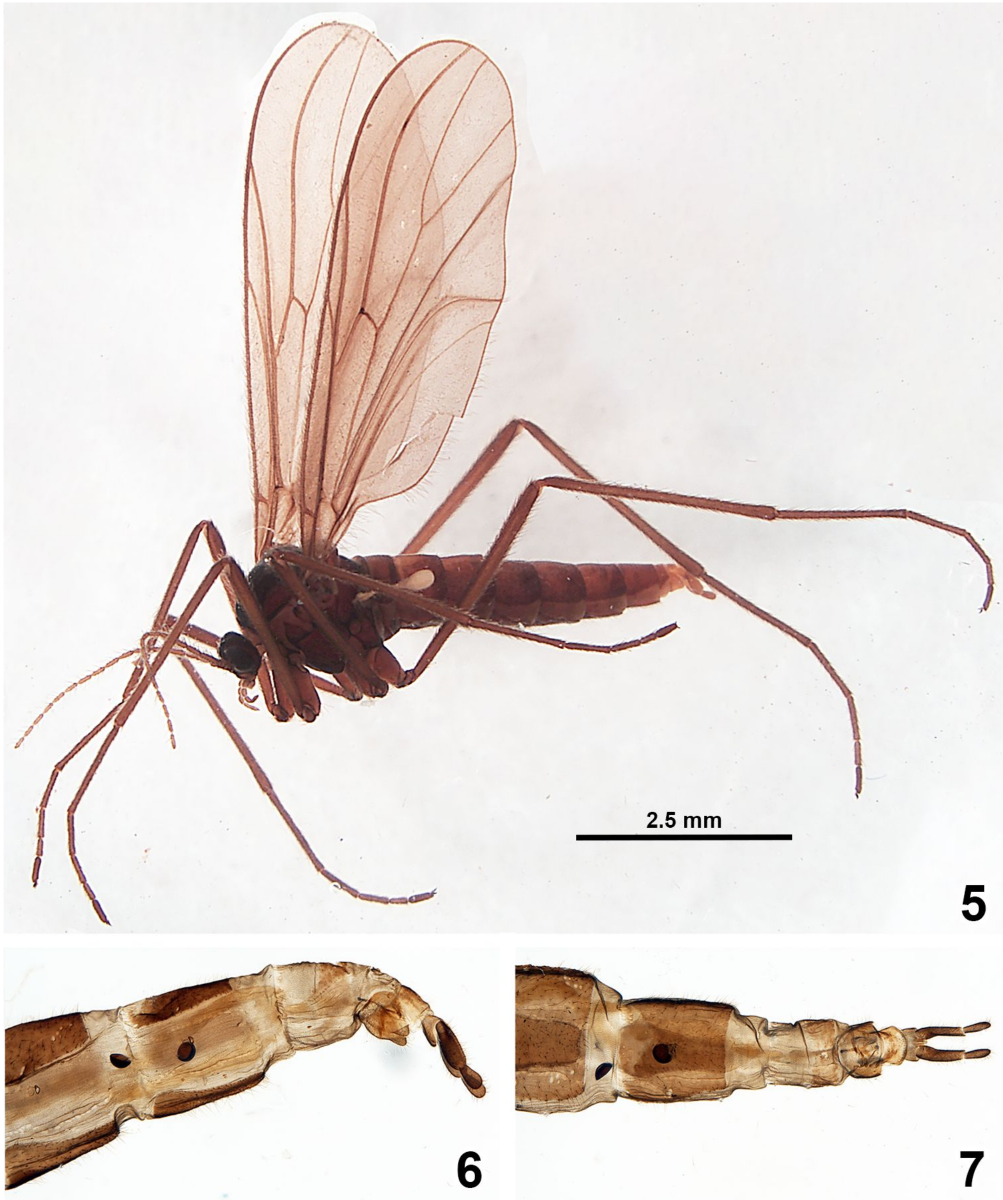
Jaschhof & Fitzgerald (2016) published the first Canadian records of the genus *Catotricha* Edwards, based on four dark brown female specimens (wing length ca. 6 mm) captured in Malaise traps set in Gatineau Park, Quebec. The traps were set-up from late September to mid-October, 2012, with the goal of capturing autumn flying Diptera. Although they left the identification incomplete, it was presumed to be *Catotricha americana* (Felt), the only species known from northeastern North America. Highlighting this rare genus led to the identification of three additional female specimens from among unidentified Diptera that were collected in a flight intercept trap in mixed forest (9–16.x.2011) in Aylmer, Quebec.

Since the 2016 publication, efforts were made to collect additional specimens of this species. On October 13, 2023, a female specimen was collected again in Gatineau Park (Meech Lake, 45.555, -75.871, T. Nakamura leg.) and two days later a male specimen was collected in Ontario at Ardoch Lake (15.x.2023, North Frontenac Twp., Frontenac Co., 44.934, -76.864, T. Nakamura leg.). The female specimen is housed in the Nakamura personal collection (Hirosaki, Japan) and the male is deposited in the Canadian National Collection of Insects, Ottawa. The discovery of the male specimen confirms the Canadian record of this species and also represents the first Ontario record of the genus and species.

In the original description of *C. americana*, Felt (1908) illustrated the male sixth flagellomere and Edwards (1938) provided the first illustration of the wing and male terminalia. Jaschhof (1998, 2001) illustrated the male terminalia, fourth flagellomere, palpus and provided a key to the six species of *Catotricha*. A photo of a live female specimen was uploaded to BugGuide (Wilson 2013) and later reproduced in Plakidas (2017). We provide the first digital images of the male of *C. americana*, including the antenna, head, thorax and male terminalia (Figs 1–4). The first photos of the female terminalia are also provided (Figs 6, 7).



Figs. 1–4. *Catotricha americana*, male. 1. habitus. 2. antenna flagellomeres. 3. head and thorax, dorsal view. 4. male terminalia, posterior view.



Figs. 5–7. *Catotricha americana*, female. 5. habitus (in alcohol). 6. terminalia, lateral view. 7. terminalia, dorsal view.

Jaschhof (2001) did not have a female specimen of *C. americana* available to create the key to species. The female of *C. americana* has a pair of well sclerotized spherical spermathecae, and consequently would key to *C. subobsoleta* (Alexander) in Jaschhof (2001), but may possibly be distinguished by its darker brown colouration (Fig. 5). No females of *C. subobsoleta* were available for comparison so further work is still needed to determine how the females of the two Nearctic *Catotricha* species can be reliably separated.

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The flower flies' airline, a stowaway on my back: The foreigner louse-fly, *Ornithoica vicina* (Walker, 1849) (Diptera: Hippoboscoidae) taking a ride on the back of the rare Colombian endemic flower fly species, *Lycopale magnifica* (Bigot, 1880) (Diptera: Syrphidae)

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In 2015, Martin Le-May, photographer and amateur birdwatcher captured a remarkable image of a weasel riding on the back of a woodpecker while it was flying (Le-May 2017). The weasel is not the first animal to taking a ride on the back of another creature. Photographers have recorded funny photos of lazy hitchhiking animals clutching on the backs of unprepared hosts (Jones and News 2019).

This phenomenon is not oblivious to insects, particularly flies. Female of *Anopheles konderi* Galvão & Damasceno (Diptera: Culicidae) has been recorded carrying eggs of the Robust Bot Flies, *Dermatobia hominis* (Linnaeus Jr. in Pallas, 1781) (Diptera: Oestridae), an ectoparasite fly that causes cutaneous myiases in mammals. Females of *Dermatobia hominis* that occur primarily in forests, exhibit a characteristic reproductive behavior, namely, the use of phoretic vectors such as mosquitoes (mouth parts) to transmit eggs and partially hatched larvae to their potential vertebrate victim, including humans (Alencar *et al*. 2017). Louse flies (Hippoboscidae) have been recorded carrying avian skin mites (Epidermoptidae) and bird lice (Phthiraptera) hitching a ride on its abdomen. The author pointed out that these small parasites are wingless and poor dispersers but can conveniently move from bird to bird by riding on the back of Louse flies (Kautz 2015).

While conducting an insect survey at the Tatamá National Park, located in the Andean Western Cordillera in Colombia, four female of *Lycopale magnifica* (Bigot, 1880) (Syrphidae: Eristalinae: Eristalini) were collected using aerial nets. The revision of one specimen under stereomicroscopic revealed the discovery of the foreigner "Louse flies" species, *Ornithoica (Ornithoica) vicina* (Walker, 1849) (Hippoboscoidae: Ornithoicinae: Olfersiini) hitchhiking on the abdomen dorsum of this rare Colombian endemic flower fly species (Fig. 1). This peculiar interaction motivated this short communication.

Ornithoica (Ornithoica) vicina is an obligate bird ectoparasite (Bequaert 1954, Fig. 22, Graciolli & de Carvalho 2003, Figs. 4, 19, 27, Maa 1969 and Wood 2010, Figs. 2, 11, and 22), associated with 10 orders, 25 families, and 86 genera of Birds, most of them Passeriformes of small size, several of them migratory, which could explain its wide distribution from Canada to Argentina and Brazil (Bequaert 1954, Maa 1969). *Ornithoica vicina* has been exclusively reported in the Northwestern and eastern Cordilleras in Colombia (Graciolli 2016: 772) and is here recorded from the Western Cordillera for the first time.

Lycopale magnifica is a Colombian endemic flower fly species, that is apparently restricted to pristine high Andean Forest ecosystems in the Western and Eastern Cordilleras between 1690–2400 m (Montoya 2016). Although its flight patterns are not very well known, it could have considerable flight capacity, flying long distances, moving up to 2 km per day as has been reported for other species (Schweiger et al. 2007).

Figure 1. The Louse fly, *Ornithoica (Ornithoica) vicina* (Walker, 1849) hitchhiking on the abdominal dorsum of the Colombian endemic flower fly species, *Lycopale magnifica* (Bigot, 1880).

Although, it is impossible to assure how this *O. vicina* specimen got on the abdomen of *L. magnifica*, for sure it was not feeding on *L. magnifica* since no sign of damage to its abdomen is noticeable. Maybe, *O. vicina* was just taking a ride over the abdomen of *L. magnifica*, as an intermediate strategy to economize energy while comfortably moving long distances from one bird to another, traveling on the flower flies' airline.

Acknowledgments

We are grateful to Tatamá National Park team and to Montezuma Rainforests for all their help during the field trip. Thanks are due to the Laboratorio de Colecciones Entomológicas Universidad de Antioquia for their support.

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Notes on *Lispocephala vitripennis* Ringdahl and *Caricea* Robineau-Desvoidy (Muscidae) in North America

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In the course of research, three male specimens of *Lispocephala vitripennis* Ringdahl, 1951, housed in the Canadian National Collection of Insects (CNC), were identified from Nepean, Ottawa (Ontario) and Gatineau Park (Quebec) (Fig. 1). At first this species appeared to represent a new North American record, until further study revealed a more complex story.

Huckett (1977) collected one male and two female specimens from New Hampshire between 1954 and 1956 that were identified as *Lispocephala spuria* (Zetterstedt, 1838), the only known record of the species in North America. Recently, the junior synonym of *L. spuria*, *L. vitripennis* Ringdahl, was found to be a good species based on the study of the genitalia of the holotypes (Hellqvist 2021). This change in status has resulted in a re-assessment of previous identification records. A recent review of British "spuria", including those collected in Scotland, has proven that they all belong to "vitripennis", indicating that *L. vitripennis* is a more southern species and *L. spuria* more northern in Europe. Furthermore, the records of *L. spuria* from continental Europe (Gregor *et al*. 2016) appear to be that of *L. vitripennis* based on comparisons with the illustrations of the male genitalia (Hellqvist 2021). The North American record of the species remains unresolved because the male specimen of *L. spuria* is apparently nonexistent in Huckett's private collection housed in the USNM.

There has been some confusion over the years with the names *Caricea* Robineau-Desvoidy, 1830 and *Lispocephala* Pokorny, 1893, for which a brief summary is given here. The genus *Caricea* was established for several species that are now placed in *Coenosia* Meigen, 1826, and *Lispocephala* was established for *Anthomyia alma* Meigen, 1826 and related species. Over the years, however, there was a dispute over what was the type-species of *Caricea*. Stein (1908: 11) considered *Coenosia alma* to be the type-species and synonymized *Lispocephala* with *Caricea*. Huckett (1934: 82–83) gave a comprehensive review of *Coenosia*, *Caricea* and the various treatments of their type-species, and he concluded that *Coenosia tigrina* (Fabricius, 1775) was the type-species of *Caricea*. Several authors followed this, in particular Emden (1940: 154–156) who treated *Caricea* as a good genus and described a large number of African species. Hennig (1961: 318–519), in his monograph of Palaearctic Muscidae, also reviewed the various type-species designations for *Coenosia* and *Caricea* and formally designated *Caricea communis* Robineau-Desvoidy, 1830 (= *Coenosia tigrina* (Fabricius, 1775)) as the type-species of *Caricea*, which he synonymized with *Coenosia*. In the Catalogue of Palaearctic Diptera, Pont (1986: 193) followed Stein (1908) as the earliest type-species designation, as a result of which *Lispocephala* became a junior synonym of *Caricea* and all the species of *Lispocephala* were transferred to *Caricea*. Most recently, an even earlier designation of *Caricea communis* as type-species of *Caricea* by Duponchel in d'Orbigny (1842: 172) was found by Evenhuis *et al*. (2010: 53), and consequently (and, we hope, definitively) *Caricea* with the *tigrina*-group of species returns to the synonymy of *Coenosia* and *Lispocephala* stands as a valid genus.

Material examined. Canada: 1♂ Ontario, Nepean, 24 Gervin Street, 45.317°N 75.720°W, 12.v.2017, 90m, Malaise trap, J. E. O'Hara, CNC698576; 1♂ Ottawa, 28.x.1956, J. R. Vockeroth; 1♂ Quebec, Lac Phillipe, 45.37°N 76.00°W, 22.viii.1959, J. R. Vockeroth.

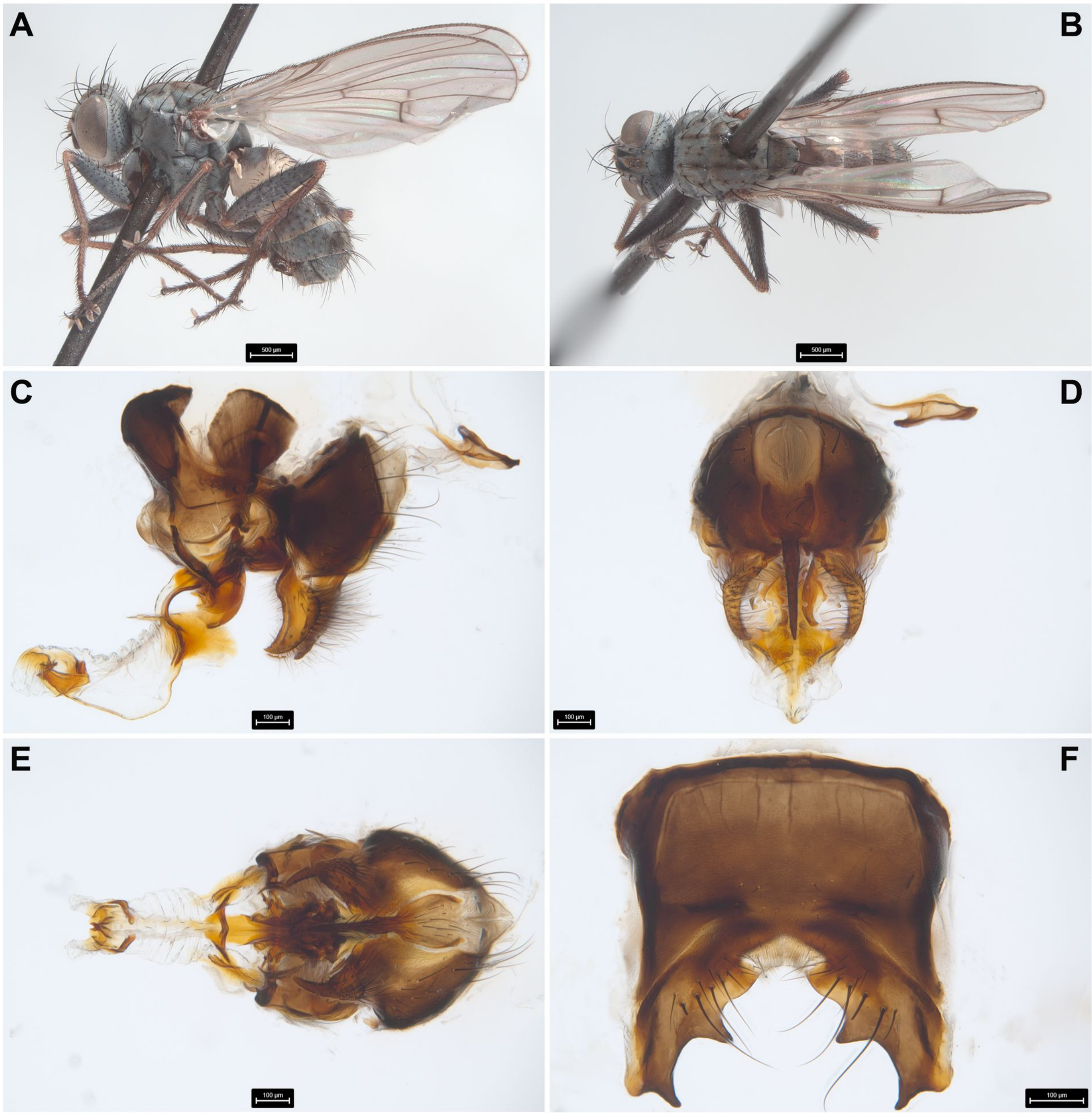

Figure 1: Male *Lispocephala vitripennis* Ringdahl (Nepean). habitus: (A) lateral, (B) dorsal; postabdomen: (C) lateral, (D) posterior, (E) ventral, (F) sternite 5.

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Phorid flies from the high desert near Los Angeles

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During the COVID-19 pandemic, when foreign travel was unavailable, I spent some time looking at local phorid fly faunas. The area within the city of Los Angeles (LA), where I work, is relatively well-sampled because of the BioSCAN project (Brown et al. 2014) and the intensive work done by the BioSCAN team. Therefore, I concentrated on some habitats nearby, whose species composition would be expected to be similar to that of Los Angeles but might reveal something about the source of the species in the better-known urban area.

One such divergent habitat is the high desert (Figs 1–2), just to the north of the Los Angeles basin, on the other side of the San Gabriel Mountain range. This area has on average, higher high temperatures, lower low temperatures, and less rainfall than LA (Figs. 3–4). There are many subregions in the high desert, and the flora varies remarkably over short distances. We chose, out of convenience, to sample at a site near Juniper Hills (JH), located at 34.44°N, 117.94°W, with an elevation of 1324 m.

Figs. 1–2. Malaise trap at the Juniper Hills site (photos by G. Kung). 1 (left). Southward-facing view. 2 (right). Northward-facing view.

We operated one Townes lightweight style Malaise trap (Townes 1972; purchased from Sante Traps) more or less continuously during the spring and summer of 2020. Specimens were captured in ethanol and identified using morphology (Borgmeier 1963, 1964, 1966, Brown and Hartop 2014, Hartop et al. 2015, 2016a, Hartop et al. 2016b).

Phorid catches from this trap were sparse. In general, these flies are more abundant in areas with more moisture. The most moisture-loving phorids, those feeding on fungi, are much more common in irrigated urban Los Angeles. For instance, *Megaselia agarici* Lintner makes up about one-quarter of the urban phorid catch, but is an insignificant fraction of the JH fauna. On the other hand, the most abundant species of *Megaselia* at JH, *M. arizonensis* Borgmeier is near the middle of the LA pack, comprising only 0.4% of the catch.

The most abundant overall species at JH was *Pseudacteon amuletum* Plowes et al., a parasitoid of native fire ants *Solenopsis xyloni*. These ants are common at JH but have been extirpated from most of the Los Angeles basin by the invasive Argentine ant, *Linepithema humile*.

The most remarkable catch from this site is the previously unknown males and second female specimen of the species *Apocephalus hirtifrons* (Peterson and Robinson). This species was originally described in its own genus, *Zyziphora* (Peterson and Robinson 1976), because of its remarkable, flattened, cockroach-like female with numerous supra-antennal setae (Fig. 5), but was synonymized with *Apocephalus* by Brown (1992). The holotype is from Colorado (Fig. 6), so it was somewhat surprising to see it in the high desert of California. There are a further two specimens from Arizona's Catalina State Park (near Tucson), and many more from Volcan Mountain and San Diego Zoo Safari Park sites in the San Diego Barcode of Life (SDBOL) collections/projects, and some from the Burns Piñon Ridge Reserve (from the state funded California Insect Barcode Initiative) all under BIN number BOLD:ABY1058 in the Biodiversity of Life Database (BOLD). We have also found males from light trap samples from Joshua Tree, near Joshua Tree National Park (see map, Fig. 6).

Table 1. Catch from Juniper Hills Malaise trap 2020. Abbreviations: sampling periods (in bold): **1)** 22 March–5 April; **2)** 5–19 April; **3)** 19 April–3 May; **4)** 8–17 May; **5)** 14–28 June**; 6)** 28 June–12 July; **7)** 12–27 July; **8)** 27 July–9 August; **9)** 9–16 August. Taxonomic abbreviations: All species are *Megaselia* except *Apo – Apocephalus*; *Phal – Phalacrotophora*; *Ps – Pseudacteon*.

	1	**2**	**3**	**4**	**5**	**6**	**7**	**8**	**9**	**total**	**percent**
Ps amuletum	0	0	0	0	5	5	8	8	10	36	38.30%
arizonensis	3	5	7	3	0	1	0	0	0	19	20.21%
Apo hirtifrons	0	0	0	0	8	4	0	2	4	18	19.15%
hirticaudata	1	11	5	0	0	0	0	0	0	17	18.09%
sydneyae	1	0	3	4	2	0	0	0	0	10	10.64%
sordida	7	1	0	1	0	0	0	0	0	9	9.57%
tecticauda	0	0	4	3	0	0	0	0	0	7	7.45%
agarici	1	0	2	1	0	2	0	0	0	6	6.38%
stoakesi	0	2	4	0	0	0	0	0	0	6	6.38%
defibaughorum	0	1	0	0	1	0	1	0	0	3	3.19%
Phal halictorum	0	0	0	0	2	1	0	0	0	3	3.19%
largifrontalis	0	2	0	0	0	0	0	0	0	2	2.13%
lombardorum	0	0	0	0	1	0	1	0	0	2	2.13%
albizona	0	0	0	0	0	0	0	0	1	1	1.06%
brejchaorum	0	0	0	0	0	0	1	0	0	1	1.06%
ciancii	0	0	1	0	0	0	0	0	0	1	1.06%
francoae	0	0	0	0	0	0	1	0	0	1	1.06%
hardingorum	1	0	0	0	0	0	0	0	0	1	1.06%
hentschkeae	0	0	0	1	0	0	0	0	0	1	1.06%
modesta	0	0	0	0	0	0	0	0	1	1	1.06%
nigra	1	0	0	0	0	0	0	0	0	1	1.06%
unknown BB9	0	0	1	0	0	0	0	0	0	1	1.06%
										147	

Table 2. Ten most common species of Phoridae (all of genus *Megaselia*) among 42,480 specimens collected from 30 sites in Los Angeles, California.

Species	**# collected**	**% of catch**	**way of life**
agarici	10,890	25.64	fungivore
sulphurizona complex	4233	9.96	unknown
nigra	3815	8.98	fungivore
lombardorum	3074	7.24	buried carrion
wiegmanae	2244	5.28	unknown
marquezi	1715	4.04	fungivore
oxboroughae	1509	3.55	unknown
armstrongorum	1428	3.36	fungivore
halterata	1349	3.18	fungivore

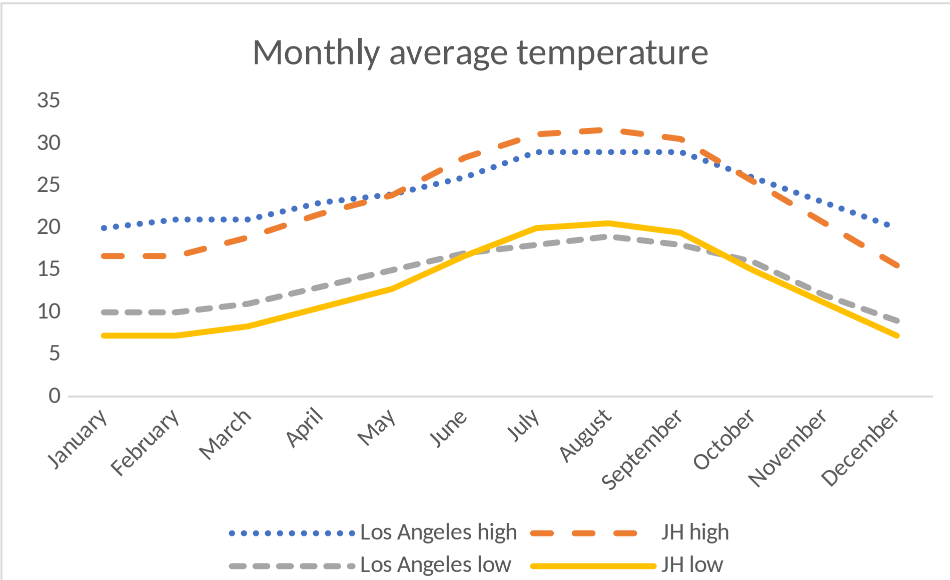


Fig. 3. Comparison of temperature means at Los Angeles and Juniper Hills.

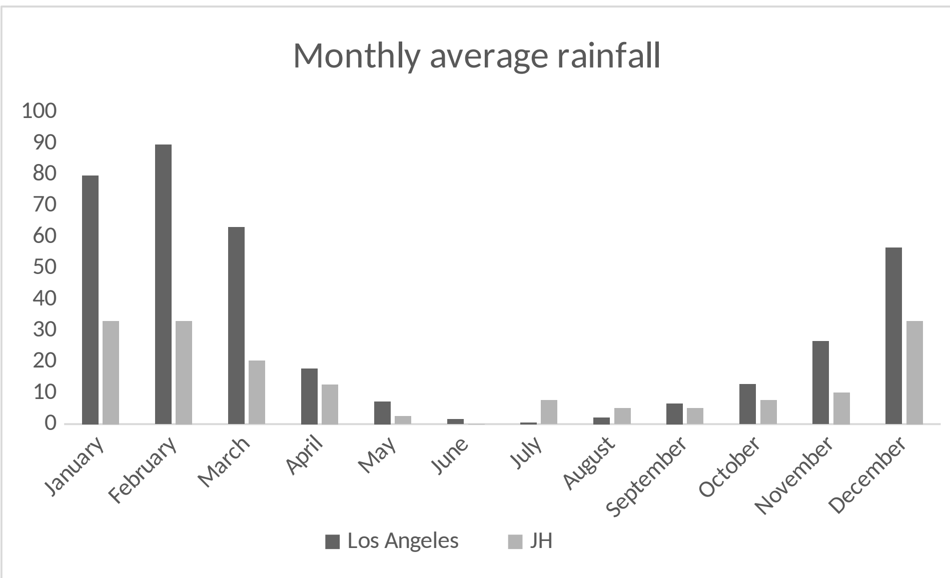
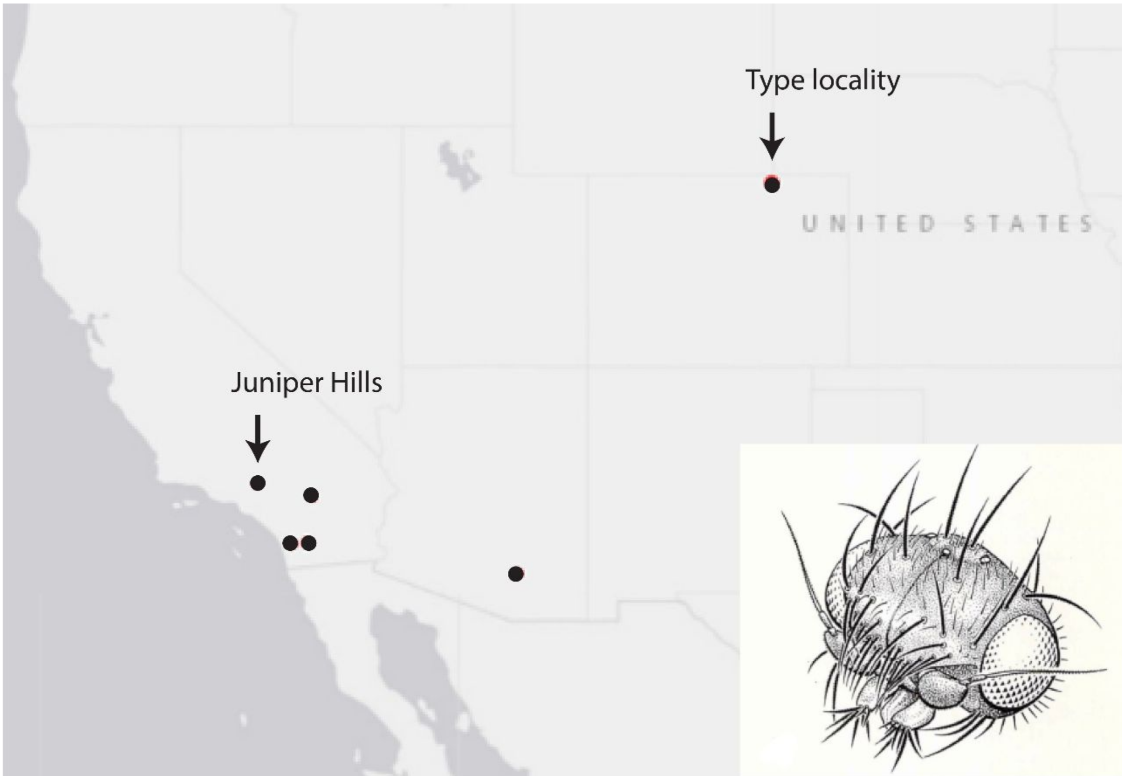


Fig. 4. Comparison of rainfall means at Los Angeles and Juniper Hills.



Figs. 5–6. *Apocephalus hirtifrons* (Peterson & Robinson). 5 (inset). Head (from Manual of Nearctic Diptera, volume 2). 6. distribution map.

Acknowledgements
I thank Kimball Garrett for operating our trap on his property, the state of California for funding the CIBI project (administered through the California Institute for Biodiversity by Dan Gluesenkamp), Bradley Zlotnick and Joshua Kohn for sharing data collected in the San DIego Barcode of Life's San Dieguito River Valley Conservancy project,and Giar-Ann Kung for her comments on an earlier version of this note and her relentless pursuit of the female of *A. hirtifrons* over the last decade.

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Photogrammetric 3D models of insects extended for interactive photos and their use

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Visualizations are an important part of research in the biological sciences, where the description of the studied object is a significant part of the published results. In recent decades, fundamental progress has been achieved in the use of electron microscopes, which enable the observation and 2D imaging of objects at very high magnification. Development of X-ray based imaging technologies (microtomography) brought 3D imaging of small biological objects in high magnification at high resolution. However, the disadvantage of these methods is the high price of the equipment, difficult operation and maintenance, and last but not least, the absence of natural colors of the object.

An interesting way of creating 3D visualizations is the use of photogrammetric imaging, which was originally used in cartography and geodesy, and nowadays has also found application in other fields. To achieve a 3D output, it uses series of digital images with their subsequent processing by relevant software. The object is scanned in several planes and angles. For each position, the object is captured in a series of successive images, by combining them, stacked image with full depth of field is created. The stacked images obtained in this way are subsequently used for the creation of a 3D model (Fig. 1). Compared to other methods of 3D imaging of insects, the photogrammetric method has several advantages. The colors of the created 3D models are identical to the natural coloring of the displayed objects, the costs of creating a 3D model are many times lower and the device is easy to operate. A detailed procedure for creating photogrammetric insect models is described by Nguyen et al. (2014).

Fig. 1. Photogrammetric 3D model of horse fly *Tabanus spodopterus*[1].

Despite of many advantages, photogrammetric 3D imaging is used very rarely. This imaging method has several constrains that largely limit its wider use. The most common are the following: 1) Imaging of some very fine structures can be problematic. Current 3D model assembly software cannot capture some very fine structures, especially the fine hairs. 2) a round shiny surface with a different refractive index (compound eyes, structural colors of beetles) causes problems during model creation, resulting in missing areas (“holes”) in the model. 3) The thin wings of insects present a challenge to folding software, especially the highly transparent and shiny wings of Diptera and Hymenoptera. The resulting 3D models have insufficiently folded wings with empty spaces between the veins. Some of the mentioned problems can be eliminated by optimizing the light during photography, minor failures can be at least partially corrected with the help of other software.

[1] https://sketchfab.com/3d-models/tabanus-spodopterus-b7dc30408a894ccf97d46f413e42954c

Figs. 2–3. *Glossina* spp. (tsetse flies). 2 (top). Interactive stacked photography of tsetse fly *Glossina austeni*[2]. 3. (bottom). Photogrammetric 3D model of *Glossina morsitans* extended for interactive photographs[3].

3D models of insects are excellent for viewing in any plane or angle, but they still do not achieve as high a resolution of microscopic details and structures as stacked photographs. The use of more powerful optics may help achieve higher quality images of tiny details, but it is unlikely that a resolution comparable in quality to stacked photography will be achieved. This problem can be solved by inserting stacked photographs into the model. High resolution stacked photos can be combined with viewing software that allows zooming and manipulation of displayed object. Such interactive photographs (IP) make it possible to observe the object at magnification comparable to lower magnification of a scanning microscope, but in its natural colors (Fig. 2). Interactive photographs are suitable for publication. To achieve a high quality display, the photos must be in the highest possible resolution. IPs have a very good use in taxonomic publications, as they allow display of key details in a quality comparable to direct observation of the object under a microscope (see Kozánek et al, 2021). Interactive photographs of key characters can be inserted in 3D models as benchmarks. By adding interactive photographs, the visual and informative value of the model significantly increases, as they allow displaying even those details of key structures that may disappear in the process of creating the model (Fig.3).

The initial impulse for the use of photogrammetric method of 3D imaging of insects was their use for museum documentation, especially type specimens. However, 3D models are also widely used for other purposes. 3D models of insects and other biological objects, their videos and models supplemented by interactive photographs, are valuable extensions of presentations and lectures, provide better visual information and, last but not the least, increase the attention of audience. 3D models have an interesting application in museum exhibitions, where they can provide interesting additional information about small organisms that, due to their small dimensions, are difficult to display as real exhibits. The large application of 3D models of insects and other small biological objects has a future, especially for educational purposes. They bring the microworld of nature closer in an interesting way and allow to get to know it in a new attractive way (Fig. 4).

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[2] https://products.virnat.sk/interactive-photographies/Glossian_austeni_habitus/index.html
[3] https://sketchfab.com/3d-models/glossina-morsitans-eadbc7de0396469498e95a4686c1aaaa

Fig. 4. Photogrammetric 3D models have very good use for educational purposes and in interactive museum exhibitions[4].

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[4] https://virnat.sk/virnat-s-r-o-presented-at-the-night-of-museums-and-galleries-at-the-slovak-national-museum/

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California comeback: *Stonemyia velutina* reappears in the West Sierra foothills

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I'm a nomadic naturalist based in California, and had the absolute pleasure of living in the foothills of the west Sierra this summer, in a little town called Tollhouse, outside of Fresno. Here, I wandered in a variety of habitats after work, chasing mostly after rare plants, but really after anything I could get my hands on. On May 26 2023, I rattled my car down a beaten up dirt road to an area called the Jose Basin, a fascinating area containing a lot of decomposed granite – granite that has decomposed into a fine sand that many plants specialise in inhabiting – and a site of some very peculiar plants, including the relictual *Carpenteria californica*.

I had checked this site several times the past few weeks, looking for a particularly rare monkeyflower – the slender-stalked monkeyflower (*Erythranthe gracilipes*) – that resides in slightly disturbed decomposed granite. Today, I was hoping things would be different, but I checked several known sites and had no luck. It was getting late, but I decided to try one last area, hopping back in my car and driving towards the final site. Unfortunately, a bridge was out, as the area had burned severely a few years ago and many of the roads were totally wrecked from last winter's huge storms. Determined, I left my car, jumped across the stream, and set out. I found the site, but looked around…no luck. It dawned on me that I wouldn't get to see this special plant this year, as I was leaving the area at the end of the week, and surely it's too late for it to still be in bloom? Just as I was about to walk back to my car, I swung around some manzanitas for one last check in a granite area. And all of a sudden I spotted a small population of these plants, near a swale with decomposed granite. What luck!

I nabbed some nice shots of the plant, and set back to the car. I usually have a long lens on tap for these walks back, in case I spot some interesting pollinators or other insects. But I left it at home today, thinking "what could be out there in the burned chaparral". Nevertheless, I kept my eyes open as I made my way back, in case something leapt out at me. As I was nearing my car, I spotted a large horsefly-like fly dashing through the air. There was little in the way of vegetation owing to the severe burn, but there were a few sapling Kellogg's oaks in the vicinity. The fly landed on one of the leaves, but I assumed it would be off in a jiffy. I tiptoed my way nearer to the leaf, hoping it would stay around long enough for me to get a macro shot on it, as it looked peculiar and I had seen little in the way of larger flies in the Jose Basin in my 4–5 visits down here. To my surprise, it was an obliging specimen, and I snapped several shots, before making my way to my car, and puzzling over what it may be on the drive back to Tollhouse.

I was later connected with A.W. Thomas on Bugguide, who indicated that this was *Stonemyia velutina*, a species last recorded in California (Middlekauff et al.1980) in 1942, and (IUCN 2007) listed as extinct! Now, a deserved word about this enigmatic fly.

Stonemyia flies are members of the Tabanidae, a family of conspicuous Orthorrhaphan flies found worldwide, familiar for their painful bites and commonly referred to as "Horse and Deer Flies". Tabanids wield long proboscises and feed on nectar, with the anautogenous females delivering the oft-painful bites. They exhibit little host-specificity in their subjects – any blood will do in providing the ingredients to successful reproduction – but they tend to mostly feed on larger mammals. Tabanids lay their eggs near water, with varying wetness depending on the genus. Their larvae are carnivorous, feeding on worms, insect larvae, and other arthropods. Once pupated, the larvae metamorphosize in around 2 weeks, and an adult emerges.

In California where we are based, there are 73 species and 7 subspecies of tabanids (Middlekauff et. al 1980), in 11 genera. *Stonemyia* reside within the *Pangoniinae* subfamily of the *Tabanidae*, a group characterized by the possession of ocelli and by the antennal flagellum usually having eight rings. *Stonemyia* are not known to be haematophagous, and little is known about their biologies, leaving plenty of room for amateur speculation. My curiosity lingers about whether this special species may have a delicate relationship with California chaparral fires, which may explain its rarity. Whatever the case may be, I'm delighted to have had an encounter with this special species and to share it with others.

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Does the Sante Malaise trap really catch more insects than the Bugdorm Malaise trap?

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An article by Sharkey & Brown (2022) in the Spring 2022 issue of *Fly Times* caught my attention. They put a Sante Traps Malaise trap head to head (literally) with a Bugdorm EZ Malaise trap at a location in Bernardino County, California. Partway through their three-week test they reversed the positions of the two traps. They concluded on the basis of both the volume of the catch and the number of braconids that the Sante trap was superior. It was a simple test and the authors concluded: "As far as we are concerned, there is no comparison. The Sante Traps model is a far superior design, and the one we use in our field work" (p. 64).

The Sharkey & Brown article was published on 17 June 2022, just about a week before I was leaving for New Brunswick, Canada, on a two-week collecting trip for tachinid flies. I was taking along five Malaise traps to set up in various places on the rural property of long-time friends who were accustomed to my collecting interests. My traps consisted of one Sante Traps Malaise trap[1], three Bugdorm EZ Malaise traps[2], and one 6-meter Gressitt and Gressitt-style Malaise trap[3]. It seemed as though I had the makings for a second comparison between Sante and Bugdorm Malaise traps.

Fig. 1. Google Earth aerial view of trap locations. A 6-meter trap was placed in location 1, a Bugdorm and a Sante trap in locations 2 and 3 (reversed halfway through survey), and two Bugdorm traps in locations 4 and 5.

The property on which I placed the five traps is a rural property situated along Route 170 in the community of Oak Bay in southeastern New Brunswick, Canada. Full details about trap placements, dates, and results for Tachinidae, were published in O'Hara (2023). Trap positions are shown in Figure 1 and trap types are explained in the caption. Of interest in this article are traps 2 and 3 that were set against the north-facing side of a dense row of trees, mostly alders. This was not what I considered the best spot on the property for Malaise traps but it suited the purpose I had in mind, which was to position the Sante and Bugdorm traps 12 meters apart against the tree line and switch them halfway through the collecting period. The Sante trap was in position 3 during the first half of the collecting period and in position 2 during the second half (Figs. 2–5). Trap heads were filled with 75% ethanol and emptied twice in each position, over four intervals: June 27–29, June 30–July 3, July 3–5, and July 6–8. Trap heads were usually emptied at dusk except on July 3rd when they were emptied during early afternoon and each trap switched to the original location of the other. Samples were preserved in fresh 75% ethanol in 300 ml jars.

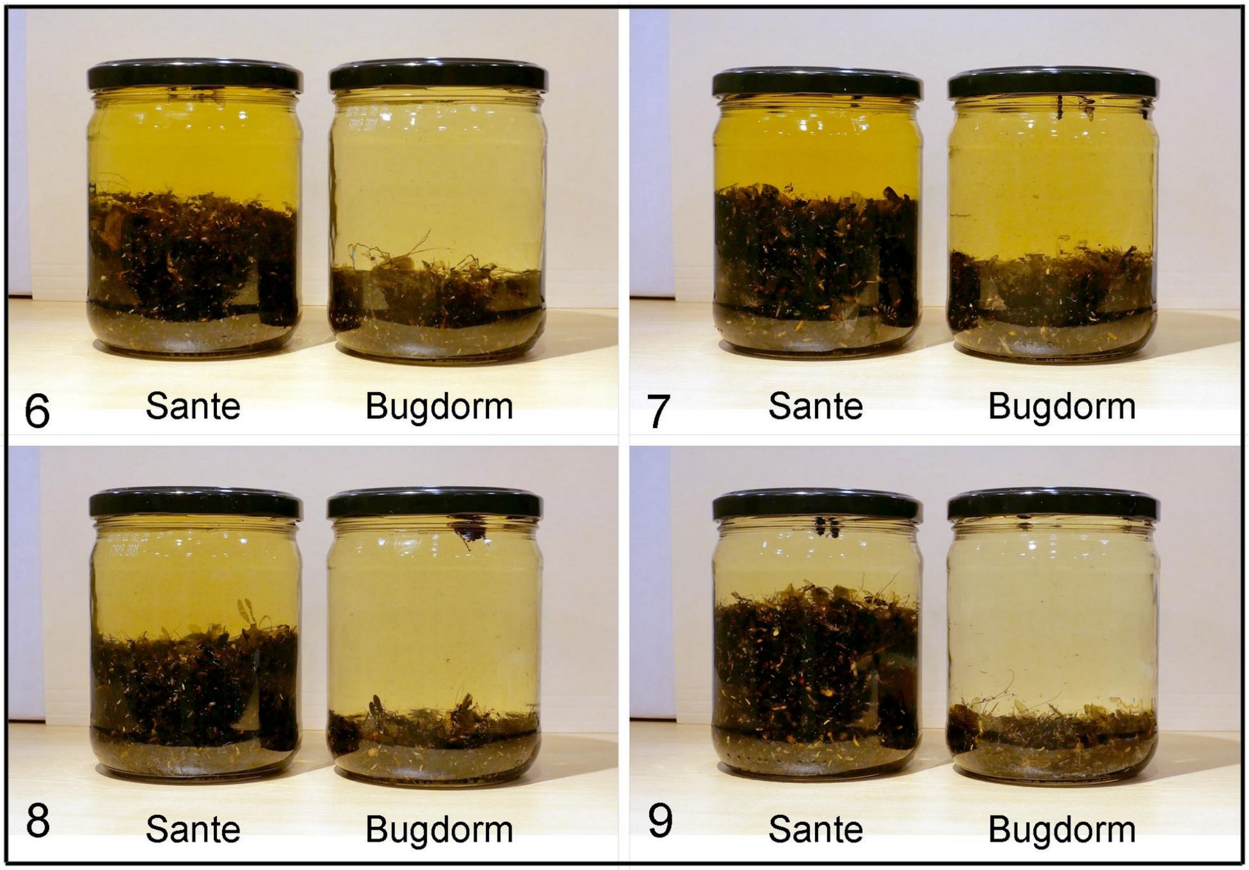
[1] https://santetraps.com/
[2] https://shop.bugdorm.com/product_info.php?products_id=326
[3] https://www.johnwhock.com/products/other-entomological-traps/standard-6-meter-malaise-trap/

Figs. 2–5. Sante and Bugdorm Malaise traps in their positions along a tree row (traps 2 and 3 in Fig. 1), with back ends against the vegetation and heads pointing north. 2. Trap positions from June 27 to July 3, with Sante trap on left and Bugdorm trap on right. 3. Close-up of Sante trap. 4. Trap positions from July 3 (early afternoon) to July 8, with Bugdorm trap on left and Sante trap on right. 5. Close-up of Bugdorm trap.

Results

The eight samples over the four intervals are shown in Figs. 6–9. Samples in Figs. 6 and 7 are from the traps in their original positions (Fig. 2) and samples in Figs. 8 and 9 are from the traps in their switched positions (Fig. 4). The samples were not quantitively compared except for Tachinidae, but qualitatively the volume of insects collected in the Sante trap was noticeably greater than in the Bugdorm trap during the four collecting periods regardless of trap position or date of collection. These results are the same as reported by Sharkey & Brown (2022); i.e., the Sante trap outperformed the Bugdorm trap.

The results for Tachinidae were only slightly in favor of the Sante trap and probably not statistically significant: 79 specimens comprising 26 species (Sante) vs. 59 specimens comprising 24 species (Bugdorm), with one species, *Siphona* (*Siphona*) *geniculata* (De Geer), dominating the results with 24 specimens (Sante) vs. 21 specimens (Bugdorm).



Figs. 6–9. Trap samples from Sante and Bugdorm Malaise traps over the four collecting periods in 2022, preserved in 300 ml jars. 6. June 27–29. 7. June 30–July 3. 8. July 3–5. 9. July 5–8. Trap positions were switched in early afternoon on July 3.

The full results of the New Brunswick tachinid survey can be found in O'Hara (2023). To briefly summarize, 736 specimens of Tachinidae were collected and pinned (fresh or from alcohol) and comprised 98 species. All have been deposited in the Canadian National Collection of Insects in Ottawa. The 6-metre Malaise trap, which I placed in a spot where I thought it would catch the most tachinids (at forest/lawn edge and facing east, Fig. 3 in O'Hara 2023), caught 424 specimens belonging to 72 species. It is a phenomenally good trap for collecting many kinds of insects.

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Plastics and microplastics and their impacts on mosquitoes

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Plastic has replaced steel in cars, paper in packaging, wood in furniture, and even homes. In the 1950's, plastic opened a new frontier. The over-consumption, lack of knowledge of its end use, and the impacts it has on the ecosystem and potential health impacts on animals and humans has been identified as a global problem with need for remediation. There are now plastic islands floating in the Pacific Ocean, known as Garbage Patches, impacting marine animals and other organisms that have now been found with microplastics inside them (Ribeiro et al. 2019). What is most shocking is that every piece of plastic that has ever been made still exists in some form.

Plastic has been identified as having improved our quality of life by reducing stress and certain costs associated with other materials. For example: Plastic dishware and cutlery have alleviated cleaning dishes when on the go or in a hurry. Takeout orders account for 269,000 tons of plastic waste a year. Most people take advantage of this without realizing the impact plastic is having on the environment. The majority of plastic waste gets shipped to another country to sit in mountains of plastic or ends up in ditches impacting water quality and impeding ditch function for water drainage, thus resulting in an ideal environment for mosquito production. Discarded plastic containers also serve as breeding site for mosquitoes after rain events and can produce hundreds of mosquitoes in a week or two. A bottle cap of water can hold around hundreds of mosquito eggs. So how many mosquitoes may be produced from the mountains of discarded plastic containers? In the U.S., 35 billion plastic bottles have been consumed annually, one can only imagine how many mosquitoes have been produced.

Over its life, plastic slowly disintegrates into tiny pieces called microplastics that are easy for birds, insects, and marine life to mistake for food. Microplastics are fragments of any type of plastic less than 5 mm (0.20 in) in length and they are microfibers from clothing, cosmetics, food packaging, industrial process, microbeads, pellets, and arise from the degradation (breakdown) of larger plastic products, such as water and soda bottles, fishing nets, plastic bags, microwave containers, tea bags and tire wear in the environment and contamination in organisms (Ribeiro et al. 2019). Polyethylene microplastics have been demonstrated to induce biochemical changes in the fresh water mosquito species, *Culex quinquesfaciatus* Say (Malafaia, et al. 2020). Do microplastics and commercial products impact mosquito life cycle and the possibility of transmission of pathogens? Preliminary studies have demonstrated ontogenic transfer (the transferring between the different life stages in different habitats) of microplastics in our ecosystems. However, more research is needed to better understand how microplastics are impacting the food chain, from insects to animals and finally into human beings (Barrett 2019, Ai-Jaibachi, et al. 2018, Gopinath, et al. 2022). One study evaluated the ingestion of microplastics in larval *Culex pipiens* (complex) mosquitoes and evaluated if these microplastics would be carried from larval stage to adulthood (Ai-Jaibachi, et al. 2018 and 2019). This study demonstrated the transfer of microplastics to the adult stage identifying as a potential aerial pathway to contamination of new environments. This could lead to the top of the food chain, either directly by mosquito bites or through the ingestion of their predators. Recently there are more publications about the impact of microplastics on mosquito development.

A laboratory colony of *Aedes aegypti* Linn. (Orland strain) at Anastasia Mosquito Control District, St. Augustine, Florida has been used for an experiment evaluating popular consumer personal care products rated with very high or zero microplastics on mosquito survival. These products were selected and purchased from local stores including: whitening toothpaste, sport sunscreen, organic toothpaste, and baby natural sunscreen as the testing material. Bioassay trials using 2nd-instar *Ae. aegypti* larvae to a series of concentrations of each product were conducted. Controls were just reverse osmosis water (Fig. 1). Mortality was recorded until all the larvae were dead or adults had emerged.

Fig. 1. Experiment set-up: Each container (100 mL stalled water in 120 mL Volume) had introduced 10 2nd and 3rd instar larvae of *Aedes aegypti*. Each product had 3 concentrations (low, middle, and high) and each concentration with 3 replications. Also, each treatment had untreated cup for control.

Through the preliminary trials, the results showed that the product, sport sunscreen appeared to stop growth in the immature stages and impacted adult emergence. The sport sunscreen resulted in the tested larvae to appear transparent with black anomalies throughout all immature stages, even into adulthood (Figs. 2, 3). In the sport sunscreen-treated group, the most staggering observation was the black spots left on the container after the adults emerged. These spots were also found in the dead larvae and pupae suggesting that the microplastic could have been discharged upon adult emergence.

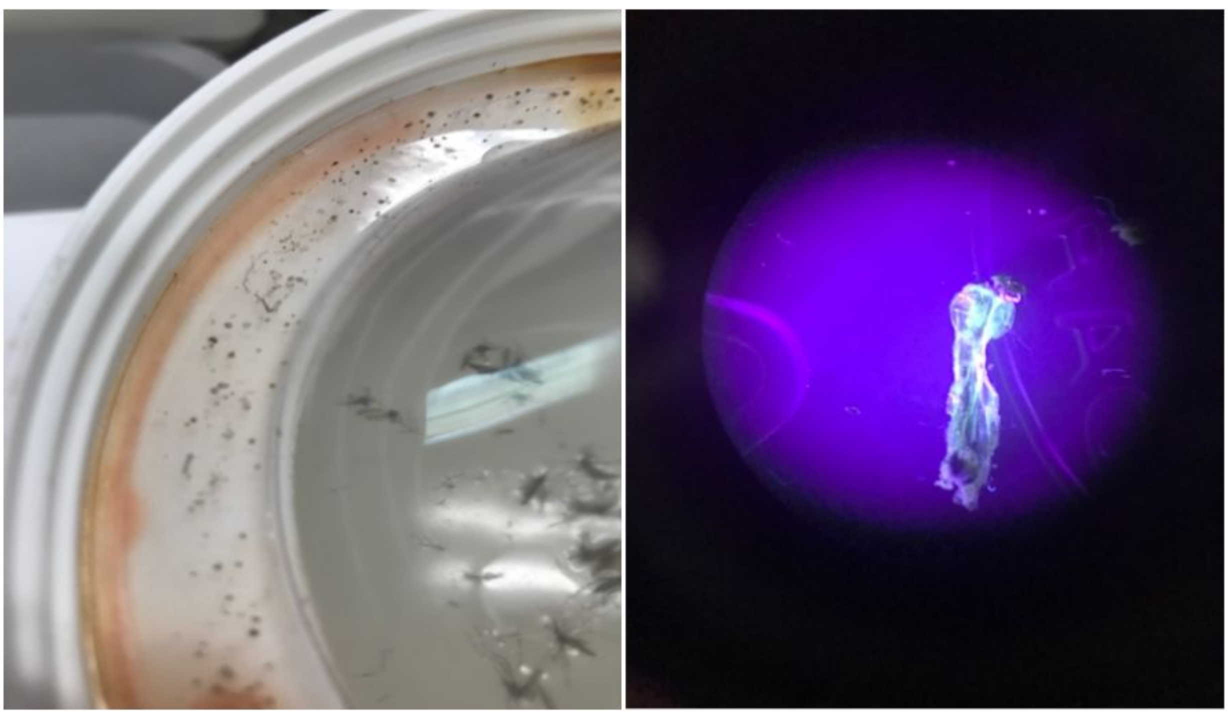

Fig. 2. Inhibited adult mosquito emerging from pupae and dead adult mosquitoes after uncompleted emerging after 9–10 day exposures of the high concentration of the products, Sport Sunscreen and Whitening Toothpaste in the laboratory.

Fig. 3. Damaged and dead larvae of *Aedes aegypti* after 7-day exposures in the high concentrations of the product, Sport Sunscreen, from early stage of larvae: Top left: damaged abdomen and siphon of larvae after exposure of the product, Sport Sunscreen. Top right: Damaged head of larvae. Bottom left: dead larvae after exposure of the product, Whitening Toothpaste. Bottom right: dead and color changed larvae and pupae after exposure of Whitening Toothpaste from early larvae stages.

The whitening toothpaste evaluation resulted in 100% mortality at all test concentrations suggesting survivorship was inhibited by the product. The larval and pupal stages were observed to be very dark in color with a swollen thorax. The other products resulted in 100% mortality but no visual observations of color change or abdomen extension.

The popular consumer personal care products included with the highest and lowest amounts of microplastic resulted in *Ae. aegypti* larvae with different colors after exposure. Upon adult emergence it was observed that more males died in all experiments than the females (Fig. 2). Observations suggested that microplastics may go from larval stages to adulthood. Environmental plastic pollution caused more breeding sites for mosquitoes and may increase the threats of public health due to female mosquitoes as a potential vector for transporting plastic residues to humans (Gopinath, et al. 2019).

Several research studies have documented that mosquito could transfer microplastic after ingestion from larval stages to adults and did not significantly impact larval development and adult behavior, size, and longevity (Barrett 2018, Ai-Jaibachi, et al. 2018, 2019, Thormeyer and Tseng, 2023). Recently, a couple of studies showed that microplastic ingestion during the larval stage may impact their bacterial microbiota (Edwards, et al, 2023) and biochemical changes in their mid-gut (Malafaia, et al. 2020). Our preliminary experiment demonstrated that the exposure larval mosquitoes to the products included microplastics impacted on larval development and survivorship. In addition, abnormal characteristics of larvae and adults were observed and potentially caused by other chemical mixtures with the products. The microplastics in the products may also serve as synergists for unknown chemistries of the products, sport sunscreen and whitening toothpaste that resulted in larval mortality. The mode of action of these two products and microplastic composition are worth addressing in future studies. The successful transference of microplastics from immature stages to adult mosquitoes may pose a public health risk and needs to be further investigated. Additionally, further studies are needed to understand how these microplastics interact with other insecticides to determine their role in mosquito behavior and control.

Contact information: Trish Becker is a Commissioner at Anastasia Mosquito Control District (AMCD) and this is her college intern project which has been carried out at AMCD in 2020. Rui-De Xue is Director at AMCD and supervisor for this project. Any commercial products mentioned in the article is research purpose only and does not mean any endorsement by AMCD.

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[Some of my] Phoridological errors

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Some errors in the Manual of Central American Diptera (MCAD) key and other works by me are corrected.

Figs. 15, 16, 18. Change female symbol to male.

1) ***Aemulophora* Borgmeier**. In 2016, Wendy Porras sent me specimens of a distinctive species of phorid from emergence traps in the ZADBI (Zurquí All Diptera Biodiversity Inventory) project in Costa Rica. We identified these flies as belonging to a new species of the Brazilian genus *Lenkoa* Borgmeier, described by Borgmeier in 1969 (Brown and Porras 2016).

Borgmeier (1969) did not recognize that he had already described this genus, and Brown (2010) did not recognize that Borgmeier had described this Costa Rican species as *Aemulophora reichenspergeri* long ago (Borgmeier 1938). The error was further perpetuated by Disney (1994) at couplet 75 in his key to females of the world genera. The Brazilian species, *Lenkoa aurita* Borgmeier is doubtlessly (in my mind) congeneric with the Costa Rican *L. phillipei*, necessitating the following actions:

Lenkoa Borgmeier 1969 is a junior synonym of *Aemulophora* Borgmeier 1938 **new synonymy**

Aemulophora aurita (Borgmeier 1969) is a **new combination**

Lenkoa phillipei Brown & Porras 2016 is a junior synonym of *Aemulophora reichenspergeri* Borgmeier 1938 **new synonymy**

Aemulophora was not included in the key to Central American phorids in the MCAD (Brown 2010), but the following will allow the females to be identified:
[to be placed after couplet 122, from the second lead in couplet 120, which should be changed by adding an “A” to “… I22A”]

122A. Posterior margin of head with pointed posterolateral processes in dorsal view (Brown and Porras 2016, fig. 2) ………………………… *Aemulophora* Borgmeier
– Posterior margin of head rounded …………….…………………………………. 123

The following should be added to the synopsis:
Aemulophora Borgmeier. This genus was proposed for the species *A. reichenspergeri* (Borgmeier 1938) from Costa Rica. Borgmeier (1969) later described the genus *Lenkoa* from Brazil, which was synonymized by me in this current paper. Brown and Porras (2016) mistakenly redescribed *A. reichenspergeri*, as noted above. The bizarre females are likely found in termite nests, although we do not have any information on them specifically. The female specimens occurred in a single emergence trap sample and have not been seen otherwise by us in Costa Rica. Brown and Porras provided a description of a co-occurring phorid that might be the otherwise unknown male.

2) ***Beckerina* Malloch**. The genus *Beckerina* was omitted from the phorid key. Only the genera similar to *Beckerina* — *Brownphora* Disney and *Enderleinphora* Disney — were included. Following is a modification to the key to include *Beckerina*.

33. Flagellomere 1 elongate pointed; wing vein R_{2+3} absent …..…. *Brownphora* Disney
– Flagellomere 1 round; wing vein R_{2+3} present ………………………….…..… 33A
33A. Supra-antennal setae on frontal margin ………………………. *Beckerina* Malloch
– Supra-antennal setae removed dorsally from frontal margin (MCAD fig. 52.16) ……
………………………………………………………...*Chaetaspidia* Borgmeier

This genus, in its more restricted form (Disney 2004) is found nearly worldwide. It is represented by several undescribed species in Costa Rica, and its way of life is completely unknown.

3) It is ironic and vexing that in a paper in which I was providing guidelines for describing new genera, I made a mistake myself! Genus *Aurisetiphora* is clearly the same as that described as *Danumphora* by Disney (2002) from Malaysia. The species name "*maggiesnowae*" is possibly still valid, as we will need to obtain much more information on these rarely encountered species.

Aurisetiphora is a junior subjective synonym of *Danumphora* **new synonymy**
Danumphora maggiensnowae is a **new combination**

Acknowledgment
I thank Soraya Uribe Celis for pointing out some of these errors.

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Systema Dipterorum Version 4.5 – update

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Thanks to the help of many users, *Systema Dipterorum* (SD) is now at Version 4.5 (posted in November 2023). When we took over management of the database from Chris Thompson in July 2018, it had (198,687 species-group names; 23,485 genus-group names; 4,671 family-group names; and 33,070 references. After removing a few thousand duplicate entries and striving to keep as up-to-date as possible with the literature, we are now at 216,304 species-group names; 24,341 genus-group names; 4,388 family-group names; and 40,199 references.

The current stats are as follows:

	Total	**Available**	**Taxonomically Valid**	**Valid Extant Spp.**
Species	216,304	209,761	174,097	**169,408**
% Reference linked	86.44%			
% Authority linked	73.70%			
Genus	24,3410	20,871	12,730	
% Reference linked	84.90%			
% Authority linked	60.22%			
Family	4,388	2,033	560	

The family-group names numbers need a bit of explanation. These names were entered into SD directly from Curt Sabrosky’s 1999 Family-Group Names Catalog, which included numerous subsequent usages and incorrect spellings (virtually all of which were entered into SD as available names). The decrease in total entries from 2018 to now includes removing duplicate entries and supra-familial names. There was also a substantial decrease in available names. We recently went through all the family-group names in SD and changed many names previously considered as available names (mostly synonyms) to subsequent usage, nomina nuda, or unavailable names. The database of family-group names now better reflects reality, whereas before, it was primarily a raw data entry of names straight from Sabrosky (1999) and treating all as available names.

The vast majority of the work that has been conducted in the last few months has been to make SD as accurate as possible. We periodically export our versions to the Catalogue of Life (CoL) and those folks have provided both feedback and tools to make our data cleaner and more accurate. The results from working “under the hood”, as it were, does not show up in the statistics, but users can be assured of having more accurate data than ever before. This does not mean that all errors have been found and corrected. We get almost daily feedback from users whenever a potential error is spotted.

Most of these are typographical errors or references not yet linked to a record (and easily fixed), but some are more serious, involving a dive into the original literature to solve a particular nomenclatural problem that may take anywhere from minutes to hours or days to solve. Our data is online for a wide variety of users, and the comments and feedback from these users are our portal to how good (or bad) we are doing. To that end, we reiterate our plea to all users to feel free to let us know if there is something missing, wrong, or misleading.

A shout-out and thank you to those who have reported errors to us 2023, assisted with database queries, and helped with proving us with papers we did not have access to – including (in no particular order): Brad Sinclair, Mark Mitchell, Tony Rees, Zachary Dankowicz, Elisabeth Stur, Pierre-Yves Gloaguen, Ximo Mengual, Sander Bot, Arthur Frost, Jere Kahänpäa, Doug Yanega, Jim O'Hara, Daniel Whitmore, Stephen Smith, Ray Gagné, Steve Gaimari, Pjotr Oosterbroek, Richard Pyle, Jostein Kjaerandsen, Patrice Bouchard, Thalles Pereira, Mathias Jaschhof, Jeff Skevington, Patrick O'Grady, Gabriel Neve, Aimee Ward, Andres Duarte, Hauteng Huang, Jorge Almeida, Gunilla Ståhls, Yury Roskov, Bill Murphy, Vlad Blagoderov, Ralph Harbach, Russell Cox, Marc Pollet, Verner Michelsen, Geoff Ower, Luciana Musetti, Art Borkent, Kevin Moran, Peter Uetz, Natasha Dreis, Ashley Kirk-Spriggs, Mihaly Földvári, Marc De Meyer, Jens-Hermann Stuke, Chris Angell, Tina Gopalan, Niyan Shehan, Stuart Longhorn, Shannon Henderson, Al Norrbom, Socrates Letana, Adrian Pont, Gary Steck, Spencer Pote, John Midgley, Dimitar Bechev, Oscar Sánchez Molina, Iestyn Jealous, Torsten Dikow, Masaaki Suwa, Christian Kehlmaier, Alessandro Camargo, J.M. Landa, Carlos Lamas, Robert Douglas, Paul Rude, Terry Whitworth, Stefan Ungricht, Nina Krivosheina, Emilia Nartshuk, Xiaolong Lin, Diego Fachin, Pavel Sánchez, Nikolaus Szucsich.

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Recently launched: Catalogus dipterorum Germaniae

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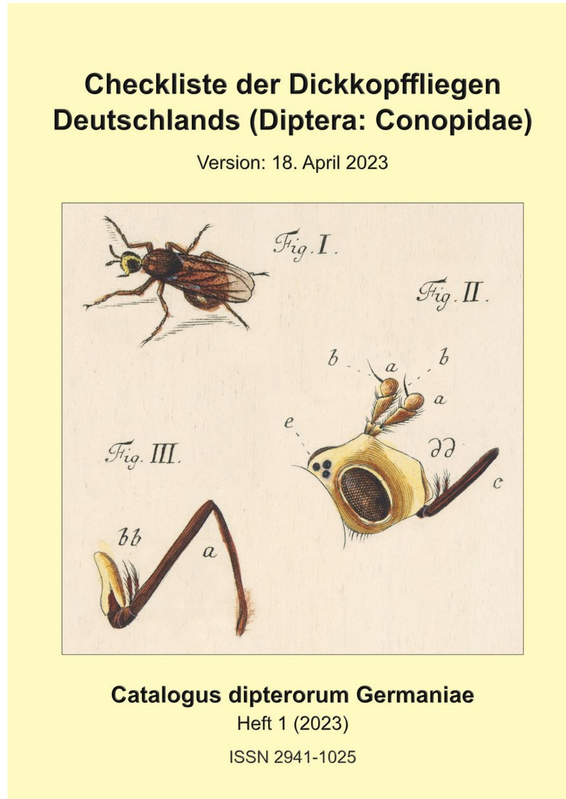

Twenty-four years ago, a 354 page strong compendium was published by a team of dipterologists, presenting the first checklist of Diptera for modern Germany (Schumann et al. 1999). Today, Schumann et al. (1999) is largely outdated due to many nomenclatorial changes, first citings of additional species, and new species descriptions. It is thus inevitable, that a revised version needs to be compiled. Fortunately, Germany not only has a long dipterological tradition but also still a reasonably ample reservoir of people interested in Diptera. The German Diptera Study Group AK DIPTERA (https://www.ak-diptera.de) currently has 130 registered Dipterists that share their passion and constantly add to the knowledge of the Diptera fauna of Germany. In order to spread the workload on many shoulders and guarantee a swift completion, it was decided not to pursue a monograph but instead to break down the task and publish each family as a separate paper in a new open access online-journal, explicitly initiated for this project, named Catalogus dipterorum Germaniae (CdG). Being an online-only journal, manuscripts can be published quickly after review and acceptance. The fundamental change to the old checklist is the addition of a bibliography for Germany that aims to be as complete as possible. In order to enable simple updates for individual families, each family checklist is published with a version number, meaning that an updated version of the family checklists can be published at any time if substantial new data accumulate. Publication language is German or English, and everyone is welcomed to participate — check out the current state at http://www.ak-diptera.de/catalogus/bearbeitungsstand/. With a forerun of less than one year, the editorial team of the CdG was able to publish the first volume on 10 June 2023. By the end of this year, the first ten family checklists will be available. All numbers of the CdG can be downloaded, distributed and used under the Creative Commons CC BY 4.0 from the journal's website (http://www.ak-diptera.de/catalogus/archiv/) and several other online repositories (https://d-nb.info/1292645261/, https://bonn.leibniz-lib.de, https://www.zobodat.at).

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Comparison of fungus gnat (Mycetophilidae and Keroplatidae) catches at two mountain sites in north central Nevada during 2021 and 2023

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Since 2017 I have been studying the mycetophilids of North Central Nevada. I became interested in this group of flies because they often turned up in EVS dry ice baited traps used for sampling adult mosquitoes as part of a mosquito abatement program. Once I started studying this group of insects the original question receded into the background in favor of many questions about their biology and the lives they led. Initially I put Malaise traps out in many different areas to discover what species might occur around here. At the same time I began looking to rear these insects out of mushrooms, moss, leaf litter and other possible sources. I learned a lot through these efforts, which caused me to realize that putting up a trap in a location for a few days or a week was not going to show me what was really in a given locality. So in 2021 I began putting traps up in different plant communities and leaving them there for the entire insect season. Depending on elevation this might run from March through December. The study sites I chose were in different mountain ranges here in northern Nevada, and most of the plant communities were islands in a sea of sagebrush, often for miles around. In 2021 I put up six Malaise traps in six different plant communities and locations, in three mountain ranges. The insect season began in March and went through the second week in December, when I took all of them down. It was a dry year, and I saw almost no mushrooms. I collected 18 different genera of Mycetophilidae in these island habitats during 2021.

The 2023 season was considerably different than the two preceding years of extreme drought. The winter of 2022/23 saw heavy snowfall, this meant that getting up into the mountains to set up traps at various sites came a month or more later than in the previous years. Small streams from snowmelt and springs, dry during 2021 were numerous and flowed until late June in many places. As spring advanced the snowfall turned to rain, sometimes torrential downpours. This continued through the summer and into fall. There were a lot more mushrooms everywhere in 2023 than in 2021, of those that were collected fungus gnats emerged from about 10% of them.

I initially put out traps in seven sites in two mountain ranges, the Bloody Runs and the Santa Rosas. An eighth trap was put up at a site on the valley floor near the Santa Rosa Mountains. By the end of the season only five of these remained. Although all but one of the traps were damaged by wind, animals and Mormon crickets, three were so severely traumatized they had to be removed well before the end of the insect season. A fourth made it almost to the end of the season before it to succumbed to animal mauling. Following are brief descriptions of the sites I selected in 2023.

Valley Floor

Big Thicket (Fig. 1) – At 4650 feet elevation, the lowest site. It consisted of a large dense thicket of buffaloberry, willows and wild rose, situated among other such thickets separated by small grassy areas. This was the first trap that went up, March 30, when snow was on the ground. Despite a mauling by horses it was not severely damaged and it remained up until November 27.

Santa Rosa Mountains

Singas Creek Riparian Area – Situated along a creek that never dries up. This was the only trap that was not damaged in some way during the season. Elevation 5240 feet.

Pussywillow (Fig. 2) – Situated at the head of a bowl shaped valley that had many springs and small streams coming out of it. I put the trap up near a cluster of pussywillows on a little creek. The trap went up on April 20, came down on October 28 as winter advanced in the area. Elevation 5800 feet.

Figs. 1–2. 1 (left). Valley Floor, Big Thicket site, early spring. 2 (right). Santa Rosa Mountains, Pussywillow site.

Granite Peak (Fig. 3) – This site, at 9400 feet, did not become accessible until June 29. During my previous effort here the trap blew away and I never saw it again. I put this trap up next to a stunted limber pine so it would get some shelter from the wind. I put guy lines on the trap to make it more secure in high winds. The site was overrun with huge numbers of Mormon crickets, this should have told me that putting up a trap here was not a good idea, but I was determined to do it and see what I could catch. Then came the 10th International Congress of Dipterology, after which I came down with Covid, followed by several days of a visit from an acquaintance – this resulted in me not returning until August. The trap had been damaged by wind, and cut to tatters by Mormon Crickets. They filled the killing jar and turned the rest of the catch to fragments in their death throes. I took the trap down, little was gained from this effort. I took a bag of needle leaf litter from underneath the limber pine and ran it through a Berlese funnel – many interesting arthropods, no mycetophilids. I have not lost interest in this site.

Buffaloberry (Fig. 4) – situated along a small creek in a side canyon, the site was surrounded by huge old buffaloberry bushes. The trap was set up on April 24, taken down June 5 when it was discovered that a major part of one wing of the canopy of the trap was torn out. It appeared that an animal had decided it was something to eat. This was a very interesting site, I was sad at taking down the trap. 5650 feet elevation.

Bloody Run Mountains

Aspen Forest – Situated in an aspen forest that runs along a small creek that originates in springs higher up. Elevation 5440 feet.

Willow Triangle (Figs. 5–6) – This site is situated on a south facing slope in the Bloody Run Mountains. It is a triangular shaped island of willow thickets interspersed with lush grass. The area had many small springs and streams. The triangle was surrounded by dry, rocky mountainsides, so it

Figs. 3–4. 3 (top left). Santa Rosa Mountains, Granite Peak site. 4 (top right). Santa Rosa Mountains, Buffaloberry site.

was a moist island in this landscape. The trap was put up in a willow thicket on May 22, taken down on October 27. It did not suffer any real damage during this period. Elevation 5710 feet.

Paradise Creek Headwaters (Fig. 7) – This site was at the headwaters of Paradise Creek, a major stream in the Bloody Runs. It was situated in a large bowl shaped basin with many springs and small streams that come together to form Paradise Creek. Mountain peaks rise 1400 feet around this area, and when I got there large areas of snow were still present. I put the trap up near the stream on May 18. I returned on June 8 at which time I found the trap apparently blown into a willow bush, all the anchor rings had been torn out apparently from wind. Whatever it had caught was gone, the killing jar had been invaded by ants. I took the trap down. 6240 feet elevation.

Comparison of the 2021 and 2023 Seasons – Singas Creek, Santa Rosa Mountains
This site was situated along Singas Creek, (Figs. 8–9), a stream that never dries up, even in drought years. It forms from the confluence of many small streams originating in springs and snowmelt high in the mountains. The trap was situated in a narrow band of riparian vegetation consisting of willow, chokecherry, wild rose and creek dogwood. Outside of this narrow band steep dry mountainsides rose. Two years of extreme drought preceded 2023, which in contrast had a wet winter, spring. Summer and fall. A wet 2019 and an average 2020 preceded a very dry 2021. Between 2021 and 2023 there was little change in this site except that the vegetation had gotten denser and more difficult to get through. In 2021, a drought year, there was still a lot of water flowing in the creek, in 2023 there was a much greater flow, in early spring I was afraid the creek would overflow it’s banks and sweep away the trap, This did not happen, however. In 2021 the trap was at the site from

Figs. 5–8. 5 (top left). Bloody Run Mountains, Willow Triangle area. 6 (top right). Bloody Run Mountains, Willow Triangle site. 7 (bottom left). Bloody Run Mountains, Paradise Creek Headwaters site. 8 (bottom right). Santa Rosa Mountains, Singas Creek site.

May 7–December 7, in 2023 it was there from April 15October 28. This was because of the weather, when the winter retreated in spring, and when it advanced again in the autumn. I had expected to see a lot of mycetophilids in 2023 because of all the moisture, but as Table 1 shows, not only were there fewer genera caught at the Singas Creek site in 2023, there were also fewer individuals. Mushrooms were far more abundant everywhere in 2023 than 2021, and I reared adults out of many of them.

Table 1. Singas Creek Riparian Area, Santa Rosa Mountains, Nevada – Comparison of Mycetophilidae and Keroplatidae caught in Malaise traps during 2021 and 2023.

2021	**2023**
Orfelia Costa 0	*Orfelia* Costa 7/8–8/12: 1 female
Megalopelma Enderlein 9/2–9/16: 1 female	*Megalopelma* Enderlein 0
Sciophila Meigan 11/16–12/2: 1 female	*Sciophila* Meigan 9/16–10/12: 2 females
Boletina Staeger 5/7–5/29: 1 female	*Boletina* Staeger 4/15–4/30: 6 females 4/30–5/16: 2 females 5/16–5/30: 4 females
Docosia Winnertz 5/7–5/29: 1 female 11/16–12/2: 1 female	*Docosia* Winnertz 5/16–5/30: 1 female
Leia Metgan 8/13–8/19: 1 female	*Leia* Meigan 0
Garrettella shermani Vockeroth 0	*Garrettella shermani* Vockeroth 4/15–4/30: 1 male
Mycetophila Meigan 9/28–10/19: 1 female 10/19–11/2: 8 females 11/2–11/16: 2 females 11/16–12/2: 1 female	*Mycetophila* Meigan 6/27– 15 females, 2 males from mushroom 8/12–8/31: 1 female 9/16–10/2: 2 females 10/12 –10/28: 1 female
Zygomyia Winnertz 9/28–10/19: 1 female 10/19–11/2: 8 females 11/2–11/16: 2 females 11/16–12/2: 1 female	*Zygomyia* Winnertz 10/12–10/28: 2 females
Anatella Winnertz 11/2–11/16: 1 female	*Anatella* Winnertz 0
Brevicornu Marshall 10/19–11/2: 1 female, 1 male 11/16–12/2: 1 female	*Brevicornu* Marshall 4/15–4/30: 1 female 10/12–10/28: 7 adults
Cordyla Meigan 6/8–6/22: 1 female 6/22–7/6: 1 female 7/6–7/20: 1 female 7/20–8/3: 1 female 11/2–11/16: 1 female 11/16–12/2: 1 female	*Cordyla* Meigan 9/9–9/23: 1 female
Phronia Winnertz 10/19–11/2: 1 male	*Phronia* Winnertz 0
Epicypta Winnertz 10/19–11/2: 1 female	*Epicypta* Winnertz 0
Exechia Winnertz 9/28–10/19: 1 female 10/19–11/2: 1 male 11/16–12/2: 1 female 12/2–12/7: 2 females	*Exechia* Winnertz 10/12–10/28: 1 female
Rymosia Winnertz 10/19–11/2: 1 male	*Rymosia* Winnertz 0
Total Genera: 14	**Total Genera: 10**

I have reared *Rymosia* out of at least 12 genera of mushroom, *Mycetophila* out of four, and *Exechia* out of one. It seems like there are very few *Rymosia* at the Singas site, despite the presence of mushrooms. Abundant mushrooms do not seem to have made for a bigger mycetophilid population at this site in 2023. The big factor in the difference between the two years seems to be the preceding two seasons. The seasons preceding 2021 were wet, maybe making for more favorable conditions for mycetophilid development, the effect of this carried over into 2021. Exactly the opposite happened in 2023, which was preceded by two extreme drought years that were probably hard on mycetophilid populations, and this showed up in 2023, a wet year. It will be interesting to see what 2024 will bring.

Comparison of 2021 and 2023 Seasons, Aspen Forest, Bloody Run Mountains

This site is situated along a small creek in a narrow gorge down which flows a small stream fed by several springs, (Figs. 10–11). There is a narrow gallery of forest composed mostly of aspen but with chokecherry and wild rose as well. In most years the near the trap location the stream goes dry by June, in 2023 it never did dry up. In 2021 the forest had an infestation of some kind of wood boring beetles – both buprestids and cerambycids have been caught in the trap there. At that time the infestation was heavier in the upper end of the forest, and trees were falling over or breaking at the midpoint of the tree. By 2023 the infestation had increased and had moved downstream to cover the entire forest (Fig. 12).Trees were falling over during the entire 2023 season, early on one fell on the trap itself. I was able to get the tree off and repair the trap. After that I moved the trap about forty feet to what seemed a safer location. No more trees fell on it, but on September 23 I found that the trap had been severely mauled by an animal – one of the poles was broken and the fabric of the trap was shredded, so I took it down that day. The elevation of the trap was 5440 feet.


Figs. 9–10. 9 (left). Santa Rosa Mountains, Singas Creek riparian area. 10 (right). Bloody Run Mountains, Aspen Forest site.

Figs. 11–12. Bloody Run Mountains, Aspen Forest site. 11 (left). Forest. 12 (right). Beetle-infested falling trees.

So this site was affected by the changes in the progress and nature of the seasons the same as the riparian site on Singas Creek (see Table 2). The beetle infestation at the aspen site was not present at all at the Singas site. It may be that two years of severe drought weakened the trees in the aspen

Table 2. Aspen Forest, Bloody Run Mountains, Nevada – Comparison of Mycetophilidae caught in Malaise traps during 2021 and 2023.

2021	**2023**
Boletina Staeger 4/7–4/20: 4 females, 3 males	*Boletina* Staeger 4/22–5/13: 2 females
Coelosia Winnertz 4/20–5/4: 1 male	*Coelosia* Winnertz 0
Docosia Winnertz 4/7–4/20: 1 male 4/20–5/14: 2 females 5/4–5/19: 1 male 5/31–6/15: 1 female	*Docosia* Winnertz 5/13–5/27: 3 females 5/27–6/19: 2 females 6/19–7/10: 1 female
Mycetophila Meigan 4/7–4/20: 2 females 4/20–5/4: 1 female	*Mycetophila* Meigan 4/22–5/13: 3 females
Zygomyia Winnertz 11/23–12/10: 1 female	*Zygomyia* Winnertz 0
Brevicornu Marshall 4/20–5/4: 1 female	*Brevicornu* Marshall 0
Cordyla Meigan 5/31–6/15: 2 females 6/15–6/29: 5 females 6/29–7/13: 2 females 8/10–8/26: 1 female 10/7–10/28: 1 female	*Cordyla* Meigan 9/9–9/23: 1 female
Exechia Winnertz 10/7–10/28: 1 female	*Exechia* Winnertz 0
Anatella Winnertz 5/31–6/15: 1 female	*Anatella* Winnertz 0
Total genera: 9	**Total genera: 4**

forest and allowed the beetle infestation to progress greatly. The change in the aspen forest was ongoing from 2021 to 2023, there may not be a forest there for many more years. There was no visible change in the Singas vegetation except that the vegetation had grown more dense between 2021 and 2023.

Conclusions

In comparing the two sites, in both there was a reduction in the number of genera captured, from 2021 to 2023 and for the most part, in the numbers of individuals taken. But when the data is examined the genera that are missing at each site do not correspond to those at the other. The only exception to this is *Anatella*, which vanished from each of the sites. Only one specimen of this genus was captured at each site in 2021, so it's absence in 2023 might be due just to chance. One male of this genus was captured at Willow Triangle near the end of May, so it is still around, maybe just not sampled well by Malaise trap.

In addition, the extent of reduction in genera between 2021 and 2023 was much greater in the Aspen Forest (55%) as compared to the Singas site (29%). It seems likely that the variability in the seasons from 2019–2023 has had an effect on the mycetophilid fauna of these two sites, but how much of the difference between them was due to that cannot even be guessed. All the work I have done over the past seven years has shown me that there is a very complex situation with regard not only to mycetophilids, but to all the organisms that live here in north central Nevada, with regard to which I have little real understanding.

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A list of English common names for the robber flies (Diptera: Asilidae) of North America north of Mexico

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A list of over 1000 English common names for the robber fly species of North America north of Mexico was developed by a community of professional and amateur entomologists interested in promoting the study of this diverse family amongst the general public. While not primarily intended as a taxonomic resource, this list also serves as a checklist of the robber flies of North America north of Mexico. It is updated when necessary.

Many believe that coining common names is unnecessary and counterproductive, given the value and utility of stable (usually), unique scientific names. Why add more? However, many of us have been forced to invent common names when publishers insist that such names be developed for field guides and other books. Online resources for the non-scientist (where common names are often used) have proliferated and frequently they demand common names. Similarly, government and non-governmental biodiversity and conservation agencies usually require common names in their management documents; *NatureServe* and the *Committee on the Status of Wildlife in Canada* (*COSEWIC*) are examples. Why not use common names coined by biologists and naturalists who know the group, so that the names represent the species as much as possible? Why not encourage everyone who wants to use common names to use these standardized ones?

Methods and Guiding Principles

In May 2022, an online spreadsheet was posted where interested participants could suggest common names (and their rationales) for genera or groups of related genera. Also included were general ideas about the coining of common names and the need for them. These ideas elicited many comments from participants. In March 2023, an online poll for assessing the popularity of the submitted names was posted. The resulting 55 genera/group names chosen were then used in the construction of 1008 species names. Again, suggestions for these species' common names were solicited from the community of robber fly enthusiasts and, where several names were suggested for a species, the compilers chose one. The completed list was posted in August 2023.

The list of candidate species was compiled from catalogs, taxonomic revisions, and other published literature. Species with reliable but not yet published occurrences north of the USA-Mexico border (such as on iNaturalist) were also included (e.g., *Pseudorus distendens* complex). Undescribed species, even if they are frequently identified or mentioned online, are not included.

Several trends in naming genera are evident. Short names are preferred over long ones. Some scientific names are used for common names because they are short, easy to pronounce, and well-recognized (e.g., *Itolia*, *Laphria*). Group names that encompass related, similar-looking genera are adopted (e.g., "Bandit" for many Cyrtopogonini, "Bladetail" for most Asilini). Names that do not describe all the taxa in a genus or larger group are fine – e.g., those that refer to one sex only (e.g., "Bladetail", "Hammertail") or those that use color as a descriptor (e.g., "Rusty Robber"). It's also all right if names relate to more taxa than the named group (e.g., "Longhorn Robber"). And there is agreement that it's good to have a mix of descriptive (based on morphology, habitat, behavior) and whimsical (e.g., the use of words similar to "robber" – "Bandit", "Ruffian", "Pirate") names.

The naming of the individual species within a genus or group of genera follows similar lines. Terms that are descriptive or otherwise relevant to the species' morphology, biology, or history are preferred, especially if they are memorable or evocative. A direct translation of the scientific specific epithet is not used if it seems illogical or obscure or if a better descriptor is available. Patronyms are frequently retained, especially in large genera with many similar species where another, more descriptive name is hard to come by. Names referring to geographical ranges or type localities are also used in these situations. And, just as in the generic names, those names that might relate to more than one taxon are not shunned; for example, using "Seashore Sandpirate" for *Lasiopogon actius* does not mean there are no other *Lasiopogon* species found along sea beaches.

Concluding Remarks

The list is a living document available at the following webpage, but the current version is copied below for the record.
https://www.robberfliesoftheworld.com/NA_CommonNames.php

The spreadsheet used in the development of these names, which contains useful information such as alternate name suggestions, the rationale for name choices, and so on, can be accessed below. Names cannot be changed, but comments can be made.

The authors give thanks to many colleagues who helped contribute name suggestions and thoughtful discussions, especially: Giff Beaton, Paul Bedell, Steve Collins, Eric Fisher, Karl McKnight, Sam O'Connell, Herschel Raney, and Richard Yank.

Additions or corrections to this list should be sent to Chris Cohen at myelaphus@gmail.com.

List

Ablautus arnaudi: Arnaud Prospector
Ablautus basini: Great Basin Prospector
Ablautus californicus: California Prospector
Ablautus coachellus: Coachella Prospector
Ablautus colei: Northwestern Prospector
Ablautus coquilletti: Coquillett Prospector
Ablautus flavipes: Yellow-legged Prospector
Ablautus linsleyi: Linsley Prospector
Ablautus mimus: Arizona Prospector
Ablautus rufotibialis: Texas Prospector
Ablautus schlingeri: Black-footed Prospector
Ablautus trifarius: Loew Prospector
Ablautus vanduzeei: Spot-winged Prospector
Amblyonychus trapezoidalis: Green-eyed Lion Fly
Andrenosoma corallium: Mexico Chiselmouth
Andrenosoma cruentum: Mangrove Chiselmouth
Andrenosoma fulvicaudum: Northern Chiselmouth
Andrenosoma hesperium: Golden-horned Chiselmouth
Andrenosoma igneum: Fiery Chiselmouth
Andrenosoma xanthocnemum: Texas Chiselmouth
Apachekolos confusio: Arizona Pixie
Apachekolos crinitus: Hairy-backed Pixie
Apachekolos scapularis: Hairy-footed Pixie
Apachekolos tenuipes: Thin-legged Pixie
Apachekolos weslacensis: Weslaco Pixie
Archilestris magnificus: Northern Brigand
Asilus sericeus: Butterflyhunter
Atomosia arkansensis: Arkansas Micropanther
Atomosia glabrata: Ringed Micropanther
Atomosia melanopogon: Blackbeard Micropanther
Atomosia mucida: Yellow-horned Micropanther
Atomosia puella: Common Micropanther
Atomosia punctifera: Pitted Micropanther
Atomosia pusilla: Little Micropanther
Atomosia rufipes: Black-booted Micropanther
Atomosia sayii: Yellow-legged Micropanther
Atomosia tibialis: White-brushed Micropanther
Atomosiella antennata: Metallic Micropanther
Atoniomyia duncani: Arizona Micropanther
Backomyia anomala: White Water Bandit
Backomyia hannai: Big Bear Bandit
Backomyia limpidipennis: Clear-winged Bandit
Backomyia schlingeri: Schlinger Bandit
Backomyia seminoensis: Wyoming Bandit

Beameromyia bifida: Notch-tailed Pixie
Beameromyia chrysops: Golden-eyed Pixie
Beameromyia disfascia: White-sided Pixie
Beameromyia floridensis: Florida Pixie
Beameromyia kawiensis: Kansas Pixie
Beameromyia lacina: Fringed Pixie
Beameromyia lunula: Madera Pixie
Beameromyia macula: Spot-legged Pixie
Beameromyia monticola: Santa Rita Pixie
Beameromyia occidentis: Chiricahua Pixie
Beameromyia pictipes: Painted Pixie
Beameromyia prairiensis: Prairie Pixie
Beameromyia punicea: Purple Pixie
Beameromyia silvacola: Woodland Pixie
Beameromyia vulgaris: Variable Pixie
Blepharepium priapus: Phoenix Hanging Thief
Blepharepium sonorense: Sonora Hanging Thief
Bohartia bromleyi: Nevada Longhorn Robber
Bohartia isabella: Sierra Longhorn Robber
Bohartia martini: Brown Longhorn Robber
Bohartia munda: Mariposa Longhorn Robber
Bohartia nitor: White-haired Longhorn Robber
Bohartia tenuis: Slender Longhorn Robber
Bromleyus flavidorsus: Yellow-backed Goggle Eye
Callinicus calcaneus: Red Goldenrobber
Callinicus pictitarsis: Halloween Goldenrobber
Callinicus pollenius: Northern Goldenrobber
Callinicus quadrinotatus: Four-spotted Goldenrobber
Callinicus vittatus: Stripe-backed Goldenrobber
Ceraturgus aurulentus: Golden Tiger Fly
Ceraturgus cornutus: Golden-horned Tiger Fly
Ceraturgus cruciatus: Ring-tailed Tiger Fly
Ceraturgus elizabethae: Red-backed Tiger Fly
Ceraturgus fasciatus: Banded Tiger Fly
Ceraturgus johnsoni: Florida Tiger Fly
Ceraturgus mitchelli: White Tiger Fly
Ceraturgus nigripes: Black-legged Tiger Fly
Ceraturgus oklahomensis: Oklahoma Tiger Fly
Ceraturgus similis: Appalachian Tiger Fly
Cerotainia albipilosa: White-faced Micropanther
Cerotainia macrocera: Yellow-faced Micropanther
Cerotainiops abdominalis: Ant-assassin Chiselmouth
Cerotainiops kernae: California Chiselmouth
Cerotainiops lucyae: Southwestern Chiselmouth
Cerotainiops mcclayi: Sedge Chiselmouth
Cerotainiops omus: Band-legged Chiselmouth
Cerotainiops wilcoxi: Wilcox Chiselmouth
Coleomyia albula: Washington Thornface
Coleomyia alticola: Colorado Thornface
Coleomyia crumborum: California Thornface
Coleomyia hinei: Northern Thornface
Coleomyia rainieri: Mount Rainier Thornface
Coleomyia rubida: Mount Hood Thornface
Coleomyia sculleni: Red-haltered Thornface
Coleomyia setigera: Silver-ringed Thornface
Comantella cristata: Crested Spotwing
Comantella fallei: Colorado Spotwing
Comantella pacifica: Okanagan Spotwing
Comantella rotgeri: Dark Spotwing
Cophura albosetosa: British Columbia Bandit
Cophura ameles: Carlsbad Bandit
Cophura arizonensis: Arizona Bandit
Cophura bella: Red Bandit
Cophura brevicornis: Common Bandit
Cophura caca: Otero Bandit
Cophura clausa: Autumn Bandit
Cophura dammersi: Salton Sea Bandit
Cophura daphne: Brown-winged Bandit
Cophura dora: Nebraska Bandit
Cophura fergusoni: Diablo Bandit
Cophura fisheri: Fisher Bandit
Cophura fur: Williston Bandit
Cophura getzendaneri: Sand-dune Bandit
Cophura hennei: San Nicolas Bandit
Cophura hesperia: Tucson Bandit
Cophura painteri: Painter Bandit
Cophura pollinosa: Baboquivari Bandit
Cophura powersi: Riverside Bandit
Cophura pulchella: Pretty Bandit
Cophura rozeni: Gray Bandit
Cophura scitula: Elegant Bandit
Cophura sculleni: Bicolored Bandit
Cophura sodalis: Sinaloa Bandit
Cophura stylosa: Oklahoma Bandit
Cophura tanbarki: Tanbark Bandit
Cophura texana: Texas Bandit
Cophura timberlakei: Timberlake Bandit
Cophura tolandi: Mojave Bandit
Cophura trunca: Tuberculate Bandit
Cophura vanduzeei: Southern California Bandit
Cophura vandykei: Blythe Bandit
Cophura vera: Stripe-tailed Bandit
Cophura vitripennis: Northern Bandit
Cyrtopogon ablautoides: Streamside Bandit
Cyrtopogon albifacies: Alberta Bandit
Cyrtopogon albifrons: White-faced Bandit
Cyrtopogon albovarians: Banff Bandit
Cyrtopogon aldrichi: Aldrich Bandit
Cyrtopogon alleni: Showy Bandit
Cyrtopogon anomalus: Spot-tailed Bandit
Cyrtopogon auratus: Golden Bandit
Cyrtopogon aurifex: Black-tufted Bandit
Cyrtopogon auripilosus: Red-horned Bandit
Cyrtopogon banksi: Banks Bandit
Cyrtopogon basingeri: Fuzzy Bandit
Cyrtopogon beameri: Madrean Bandit
Cyrtopogon bigelowi: Yellow-tailed Bandit
Cyrtopogon bimaculus: Two-spotted Bandit
Cyrtopogon caesius: Blue-gray Bandit
Cyrtopogon callipedilus: Fancy-foot Bandit
Cyrtopogon chagnoni: Quebec Bandit
Cyrtopogon curtipennis: Short-winged Bandit

Cyrtopogon curtistylus: Short-horned Bandit
Cyrtopogon cymbalista: Paddle-foot Bandit
Cyrtopogon dasyllis: Boreal Bumblebee Bandit
Cyrtopogon dasylloides: Western Bumblebee Bandit
Cyrtopogon dubius: Cascade Bandit
Cyrtopogon evidens: White-waisted Bandit
Cyrtopogon falto: Golden-faced Bandit
Cyrtopogon fumipennis: Smoky-winged Bandit
Cyrtopogon glarealis: Ridge-backed Bandit
Cyrtopogon idahoensis: Idaho Bandit
Cyrtopogon infuscatus: Bulbous-faced Bandit
Cyrtopogon inversus: White-brushed Bandit
Cyrtopogon jemezi: Jemez Bandit
Cyrtopogon laphriformis: Two-striped Bandit
Cyrtopogon leptotarsus: Thin-footed Bandit
Cyrtopogon lineotarsus: Slender-footed Bandit
Cyrtopogon longimanus: San Rafael Bandit
Cyrtopogon lutatius: Brush-faced Bandit
Cyrtopogon lyratus: Lyre-backed Bandit
Cyrtopogon marginalis: Shiny-edged Bandit
Cyrtopogon montanus: Mountain Bandit
Cyrtopogon nitidus: Shining Bandit
Cyrtopogon nugator: White-clawed Bandit
Cyrtopogon perspicax: Sharp-eyed Bandit
Cyrtopogon planitarsus: Flat-footed Bandit
Cyrtopogon platycaudus: Broad-tailed Bandit
Cyrtopogon plausor: Semaphore Bandit
Cyrtopogon praepes: Swift Bandit
Cyrtopogon princeps: Royal Bandit
Cyrtopogon profusus: Splendid Bandit
Cyrtopogon pulcher: Beautiful Bandit
Cyrtopogon rainieri: Mount Rainier Bandit
Cyrtopogon rattus: Sierra Bandit
Cyrtopogon rejectus: Calaveras Bandit
Cyrtopogon rufotarsus: Topknot Bandit
Cyrtopogon sabroskyi: Sabrosky Bandit
Cyrtopogon sansoni: Alberta Bandit
Cyrtopogon semitarius: Yellow-waisted Bandit
Cyrtopogon stenofrons: Narrow-faced Bandit
Cyrtopogon sudator: Yosemite Bandit
Cyrtopogon swezeyi: Bryce Canyon Bandit
Cyrtopogon tenuis: Slender Bandit
Cyrtopogon thompsoni: White-tufted Bandit
Cyrtopogon tibialis: Silver City Bandit
Cyrtopogon vanduzeei: Tahoe Bandit
Cyrtopogon vandykei: Van Dyke Bandit
Cyrtopogon varans: Golden-tailed Bandit
Cyrtopogon vulneratus: Reddish-tailed Bandit
Cyrtopogon willistoni: Sagebrush Bandit
Dasylechia atrox: Bumblerobber
Dicolonus medius: Tulare Longhorn Robber
Dicolonus nigricentrus: Black-bristled Longhorn Robber
Dicolonus simplex: Curly-haired Longhorn Robber
Dicolonus sparsipilosus: Orange Longhorn Robber
Dicranus jaliscoensis: Northern Talon Fly
Dicropaltum cumbipilosus: Flat-haired Bladetail
Dicropaltum humilis: Texas Bladetail
Dicropaltum mesae: Little Golden Bladetail
Dicropaltum rubicundus: Red Bladetail
Dioctria henshawi: Cascades Longhorn Robber
Dioctria hyalipennis: European Longhorn Robber
Dioctria pleuralis: Laguna Longhorn Robber
Dioctria pusio: Red-tailed Longhorn Robber
Dioctria vera: Southwestern Longhorn Robber
Dioctria wilcoxi: Tuolumne Longhorn Robber
Diogmites angustipennis: Slender-winged Hanging Thief
Diogmites basalis: Northern Hanging Thief
Diogmites bilobatus: Golden Hanging Thief
Diogmites coloradensis: Colorado Hanging Thief
Diogmites contortus: Yellow Hanging Thief
Diogmites crudelis: Giant Hanging Thief
Diogmites discolor: Silver-spotted Hanging Thief
Diogmites esuriens: Short-striped Hanging Thief
Diogmites fragilis: Delicate Hanging Thief
Diogmites herennius: Ohio Hanging Thief
Diogmites misellus: Little Hanging Thief
Diogmites missouriensis: Missouri Hanging Thief
Diogmites neoternatus: Plain-tailed Hanging Thief
Diogmites perplexus: New Mexico Hanging Thief
Diogmites platypterus: Black Hanging Thief
Diogmites pritchardi: Oklahoma Hanging Thief
Diogmites properans: Black-banded Hanging Thief
Diogmites sallei: Dark-winged Hanging Thief
Diogmites salutans: Brown-banded Hanging Thief
Diogmites ternatus: Cuba Hanging Thief
Diogmites texanus: Texas Hanging Thief
Diogmites unicolor: Arizona Hanging Thief
Eccritosia zamon: Northern Flare Fly
Echthodopa carolinensis: Carolina Longhorn Robber
Echthodopa formosa: Eastern Longhorn Robber
Echthodopa pubera: Western Longhorn Robber
Efferia aestuans: Eastern Hammertail
Efferia albibarbis: Sand Hammertail
Efferia anacapai: Anacapa Hammertail
Efferia antiochi: Antioch Hammertail
Efferia anza: Anza Hammertail
Efferia apache: Apache Hammertail
Efferia apicalis: Southeastern Hammertail
Efferia argentifrons: Silver-faced Hammertail
Efferia argyrosoma: Pewter Hammertail
Efferia arida: Aridlands Hammertail
Efferia armata: Golden Club-legged Hammertail
Efferia aurimystacea: Golden-faced Hammertail
Efferia auripila: Golden Hammertail
Efferia azteci: Aztec Hammertail
Efferia basingeri: Nevada Hammertail
Efferia basini: Eureka Hammertail
Efferia beameri: Beamer Hammertail
Efferia belfragei: Hine Hammertail
Efferia benedicti: Sagebrush Hammertail
Efferia bexarensis: Bexar Hammertail
Efferia bicaudata: Great Plains Hammertail

Efferia bicolor: Lesser Mesquite Hammertail
Efferia bryanti: Madera Canyon Hammertail
Efferia cabeza: Cochise Hammertail
Efferia caliente: Caliente Hammertail
Efferia californica: California Hammertail
Efferia cana: Silver-tailed Hammertail
Efferia candida: White Hammertail
Efferia canella: Sierritas Hammertail
Efferia carbonaria: Black-legged Hammertail
Efferia clementei: San Clemente Hammertail
Efferia completa: Rio Grande Hammertail
Efferia coquillettii: Coquillett Hammertail
Efferia costalis: Crested Hammertail
Efferia coulei: Northwestern Hammertail
Efferia cressoni: Metallic Hammertail
Efferia cuervana: Red-legged Hammertail
Efferia davisi: Davis Hammertail
Efferia deserti: Desert Hammertail
Efferia duncani: Dusty Hammertail
Efferia ehrenbergi: April Hammertail
Efferia femorata: Pine-trunk Hammertail
Efferia fisheri: Fisher Hammertail
Efferia frewingi: Columbia River Hammertail
Efferia gila: Gila Hammertail
Efferia grandis: Great Mesquite Hammertail
Efferia halli: San Bernardino Hammertail
Efferia harveyi: Harvey Hammertail
Efferia helenae: Helen Hammertail
Efferia imperialis: Emperor Hammertail
Efferia incognita: Juniper Hammertail
Efferia inflata: Broad-tipped Hammertail
Efferia jubata: Maned Hammertail
Efferia kansensis: Kansas Hammertail
Efferia kelloggi: Flagstaff Hammertail
Efferia kondratieffi: Kondratieff Hammertail
Efferia latruncula: Bristle Crested Hammertail
Efferia leucocoma: White-haired Hammertail
Efferia luna: Luna Hammertail
Efferia macroxipha: Long-sword Hammertail
Efferia mesquite: Mesquite Hammertail
Efferia monki: Bromley Hammertail
Efferia mortensoni: Portal Hammertail
Efferia nemoralis: Woodland Hammertail
Efferia neoinflata: Yosemite Hammertail
Efferia neosimilis: Ocotillo Hammertail
Efferia okanagana: Okanagan Hammertail
Efferia ordwayae: Gray Crested Hammertail
Efferia pallidula: Pale Hammertail
Efferia parkeri: Arizona Hammertail
Efferia peralta: Peralta Hammertail
Efferia pernicis: Los Angeles Hammertail
Efferia pilosa: El Paso Hammertail
Efferia pinali: Pinal Hammertail
Efferia plena: Yellow Hammertail
Efferia pogonias: Yellow-bearded Hammertail
Efferia prairiensis: Prairie Hammertail
Efferia prattii: Laredo Hammertail
Efferia producta: White Thorny Hammertail
Efferia prolifica: Autumn Crested Hammertail
Efferia rapax: Yellow-haired Hammertail
Efferia setigera: Hairy Thorny Hammertail
Efferia slossonae: Scrub Hammertail
Efferia snowi: Snow Hammertail
Efferia spiniventris: Spine-belly Hammertail
Efferia staminea: Straw-faced Hammertail
Efferia subarida: Tucson Hammertail
Efferia subcuprea: Chiricahua Hammertail
Efferia subpilosa: Beaver Creek Hammertail
Efferia tabescens: Sesqui Hammertail
Efferia tagax: Arizona Club-legged Hammertail
Efferia tapeats: Grand Canyon Hammertail
Efferia texana: Texas Hammertail
Efferia tolandi: Toland Hammertail
Efferia tricella: Silver Hammertail
Efferia triton: Triton Hammertail
Efferia truncata: Huachuca Hammertail
Efferia tuberculata: Thorny Hammertail
Efferia tucsoni: Reddish Thorny Hammertail
Efferia utahensis: Utah Hammertail
Efferia varipes: Colorado Hammertail
Efferia vertebrata: Mountain Hammertail
Efferia wilcoxi: Wilcox Hammertail
Efferia willistoni: Williston Hammertail
Efferia yermo: Yermo Hammertail
Efferia yuma: Yuma Hammertail
Efferia zonata: Banded Hammertail
Eucyrtopogon albibarbus: White-haired Spotwing
Eucyrtopogon calcaratus: Spurred Spotwing
Eucyrtopogon comantis: Chilcotin Spotwing
Eucyrtopogon diversipilosis: Northern Spotwing
Eucyrtopogon incompletus: Prairie Spotwing
Eucyrtopogon kelloggi: New Mexico Spotwing
Eucyrtopogon maculosus: Washington Spotwing
Eucyrtopogon nebulo: Common Spotwing
Eucyrtopogon nigripes: Black-footed Spotwing
Eucyrtopogon punctipennis: Northwestern Spotwing
Eucyrtopogon spiniger: Spiny Spotwing
Eucyrtopogon varipennis: Brown-tipped Spotwing
Eudioctria albius: Northeastern Longhorn Robber
Eudioctria beameri: Sequoia Longhorn Robber
Eudioctria brevis: Little Longhorn Robber
Eudioctria denuda: Spot-backed Longhorn Robber
Eudioctria disjuncta: Texas Longhorn Robber
Eudioctria dissimilis: San Jacinto Longhorn Robber
Eudioctria doanei: Pasadena Longhorn Robber
Eudioctria media: Pacific Longhorn Robber
Eudioctria monrovia: Monrovia Longhorn Robber
Eudioctria nitida: Shiny Longhorn Robber
Eudioctria propinqua: Appalachian Longhorn Robber
Eudioctria sackeni: Sacken Longhorn Robber
Eudioctria tibialis: Virginia Longhorn Robber
Eudioctria unica: Bare-faced Longhorn Robber

Hadrokolos cazieri: Chisos Twigsitter
Hadrokolos pritchardi: Pritchard Twigsitter
Hadrokolos texanus: Red-legged Twigsitter
Haplopogon bullatus: Brown Goggle Eye
Haplopogon dicksoni: Gray-backed Goggle Eye
Haplopogon erinus: New Mexico Goggle Eye
Haplopogon latus: Brownsville Goggle Eye
Haplopogon parkeri: Arizona Goggle Eye
Haplopogon triangulatus: Texas Goggle Eye
Haplopogon utahensis: Utah Goggle Eye
Heteropogon arizonensis: Arizona Twigsitter
Heteropogon cazieri: Cazier Twigsitter
Heteropogon chiricahua: Chiricahua Twigsitter
Heteropogon cirrhatus: Curly-headed Twigsitter
Heteropogon currani: Oklahoma Twigsitter
Heteropogon davisi: Sabino Twigsitter
Heteropogon divisus: Golden-tailed Twigsitter
Heteropogon duncani: Spring Twigsitter
Heteropogon fisheri: Fisher Twigsitter
Heteropogon johnsoni: Fuzzy-white Twigsitter
Heteropogon lautus: Elegant Twigsitter
Heteropogon ludius: Shining Twigsitter
Heteropogon macerinus: Eastern Twigsitter
Heteropogon maculinervis: Spot-veined Twigsitter
Heteropogon martini: NevadaTwigsitter
Heteropogon patruelis: Dark-winged Twigsitter
Heteropogon paurosomus: Fringe-footed Twigsitter
Heteropogon phoenicurus: Red-tailed Twigsitter
Heteropogon rubidus: Smoky-winged Twigsitter
Heteropogon rubrifasciatus: Red-banded Twigsitter
Heteropogon senilis: Hoary Twigsitter
Heteropogon spatulatus: Broad-tailed Twigsitter
Heteropogon stonei: Plateau Twigsitter
Heteropogon tolandi: Pinyon Twigsitter
Heteropogon wilcoxi: Wilcox Twigsitter
Hodophylax aridus: Aridlands Bandit
Hodophylax basingeri: San Bernardino Bandit
Hodophylax halli: Walker Pass Bandit
Hodophylax tolandi: New Mexico Bandit
Holcocephala abdominalis: Golden Goggle Eye
Holcocephala calva: Gray Goggle Eye
Holcocephala fusca: Dusky Goggle Eye
Holopogon acropennis: Pointed-winged Twigsitter
Holopogon albipilosus: White-haired Twigsitter
Holopogon atrifrons: Black-faced Twigsitter
Holopogon atripennis: Dark-winged Twigsitter
Holopogon caesariatus: ShaggyTwigsitter
Holopogon crinitis: Hairy Twigsitter
Holopogon currani: Curran Twigsitter
Holopogon guttulus: Gray-sided Twigsitter
Holopogon mica: Little Twigsitter
Holopogon mingusae: Mingus Twigsitter
Holopogon oriens: Eastern Twigsitter
Holopogon phaeonotus: Brown Twigsitter
Holopogon sapphirus: Sapphire-tailed Twigsitter
Holopogon seniculus: Yellow-veined Twigsitter
Holopogon snowi: Snow Twigsitter
Holopogon stellatus: Western Twigsitter
Holopogon umbrinus: Shadow Twigsitter
Holopogon vockerothi: Vockeroth Twigsitter
Holopogon wilcoxi: Wilcox Twigsitter
Itolia atripes: Black-legged Itolia
Itolia maculata: Spotted Itolia
Itolia timberlakei: Banded Itolia
Lampria bicolor: Black-backed Lampria
Lampria rubriventris: Gold-backed Lampria
Laphria aeatus: Northern Laphria
Laphria affinus: Autumn Laphria
Laphria aimatis: Western Orange-patched Laphria
Laphria aktis: Radiant Laphria
Laphria altitudina: Northeastern Laphria
Laphria apila: Bald Laphria
Laphria asackeni: Golden Laphria
Laphria astur: Western Yellow-backed Laphria
Laphria asturina: Red-banded Laphria
Laphria calvescenta: Bald Laphria
Laphria canis: Common Black Laphria
Laphria carbonaria: California Laphria
Laphria champlainii: Champlain Laphria
Laphria cinerea: Ashy Laphria
Laphria columbica: Columbia Laphria
Laphria coquillettii: Red-tailed Laphria
Laphria divisor: Black-waisted Laphria
Laphria engelhardti: Southwestern Laphria
Laphria fattigi: Georgia Laphria
Laphria felis: Variable Laphria
Laphria fernaldi: Red-spotted Laphria
Laphria ferox: Fierce Laphria
Laphria flavicollis: Black-tailed Laphria
Laphria franciscana: Western Black Laphria
Laphria gilva: Orange-patched Laphria
Laphria grossa: Giant Laphria
Laphria huron: Huron Laphria
Laphria index: Arrowhead Laphria
Laphria insignis: Remarkable Laphria
Laphria ithypyga: Southern Arrowhead Laphria
Laphria janus: Orange-tailed Laphria
Laphria lata: Goliath Laphria
Laphria macquarti: Cowboy Laphria
Laphria milvina: Hawkish Laphria
Laphria nigella: Texas Laphria
Laphria partitor: Two-toned Laphria
Laphria posticata: Boreal Laphria
Laphria rapax: Greedy Laphria
Laphria royalensis: Isle Royale Laphria
Laphria sackeni: Pacific Laphria
Laphria sacrator: Yellow-waisted Laphria
Laphria sadales: Red-legged Laphria
Laphria saffrana: Painted Laphria
Laphria scorpio: Scorpion Laphria
Laphria semitecta: Manitoba Laphria
Laphria sericea: Silky Laphria

Laphria sicula: Dagger Laphria
Laphria thoracica: Eastern Yellow-backed Laphria
Laphria trux: Silver-backed Laphria
Laphria unicolor: Yellow Laphria
Laphria ventralis: Orange-bellied Laphria
Laphria virginica: Pinewoods Laphria
Laphria vivax: Lively Laphria
Laphria vorax: Prairie Laphria
Laphria vultur: Golden-orange Laphria
Laphria winnemana: Winnemana Laphria
Laphystia albiceps: Texas Dunerobber
Laphystia annulata: Ringed Dunerobber
Laphystia bromleyi: Oklahoma Dunerobber
Laphystia brookmani: California Dunerobber
Laphystia canadensis: Canada Dunerobber
Laphystia cazieri: Cazier Dunerobber
Laphystia confusa: Golden-backed Dunerobber
Laphystia duncani: Tempe Dunerobber
Laphystia flavipes: Yellow-legged Dunerobber
Laphystia howlandi: Golden Dunerobber
Laphystia jamesi: Long Beach Dunerobber
Laphystia laguna: Brown Dunerobber
Laphystia lanhami: Colorado Dunerobber
Laphystia limatula: Orange-banded Dunerobber
Laphystia litoralis: Atlantic Dunerobber
Laphystia martini: Gray-tailed Dunerobber
Laphystia notata: Shiny Dunerobber
Laphystia ochreifrons: Ochre-faced Dunerobber
Laphystia opaca: Obscure Gulf Dunerobber
Laphystia rubra: Red Dunerobber
Laphystia rufiventris: Red-bellied Dunerobber
Laphystia rufofasciata: Red-banded Dunerobber
Laphystia sexfasciata: Six-banded Dunerobber
Laphystia sillersi: Mexico Dunerobber
Laphystia snowi: Kansas Dunerobber
Laphystia texensis: Gulf Dunerobber
Laphystia tolandi: Nevada Dunerobber
Laphystia torpida: San Joaquin Dunerobber
Laphystia utahensis: Utah Dunerobber
Laphystia varipes: Plains Dunerobber
Lasiopogon actius: Seashore Sandpirate
Lasiopogon albidus: Pale Sandpirate
Lasiopogon aldrichii: Subalpine Sandpirate
Lasiopogon anaphlecter: Yosemite Sandpirate
Lasiopogon apache: Apache Sandpirate
Lasiopogon apoecus: Mexico Sandpirate
Lasiopogon appalachensis: Appalachian Sandpirate
Lasiopogon arenicola: San Francisco Sandpirate
Lasiopogon asilomar: Asilomar Sandpirate
Lasiopogon bitumineus: Dark Pismo Sandpirate
Lasiopogon bivittatus: Two-striped Sandpirate
Lasiopogon californicus: California Sandpirate
Lasiopogon canningsi: Cannings Sandpirate
Lasiopogon canus: Beringian Sandpirate
Lasiopogon chaetosus: Bristly Sandpirate
Lasiopogon chrysotus: Golden Sandpirate
Lasiopogon cinereus: Ashy Sandpirate
Lasiopogon coconino: Coconino Sandpirate
Lasiopogon condylophorus: Mountain Lake Sandpirate
Lasiopogon currani: Glade Sandpirate
Lasiopogon delicatulus: Rainier Sandpirate
Lasiopogon dimicki: Oregon-beach Sandpirate
Lasiopogon drabicolum: Gray Sandpirate
Lasiopogon esau: Hairy Sandpirate
Lasiopogon flammeus: Fiery Sandpirate
Lasiopogon fumipennis: Smoky-winged Sandpirate
Lasiopogon gabrieli: San Gabriel Sandpirate
Lasiopogon hinei: Siberian Sandpirate
Lasiopogon karli: Gila Sandpirate
Lasiopogon lavignei: Lavigne Sandpirate
Lasiopogon littoris: Pale Pismo Sandpirate
Lasiopogon marshalli: Marshall Sandpirate
Lasiopogon martinensis: Snake River Sandpirate
Lasiopogon monticola: Mountain Sandpirate
Lasiopogon nelsoni: Nelson Sandpirate
Lasiopogon odontotus: San Joaquin Sandpirate
Lasiopogon oklahomensis: Ozark Sandpirate
Lasiopogon opaculus: Dusky Sandpirate
Lasiopogon pacificus: Pacific Sandpirate
Lasiopogon piestolophus: Gulf Coast Sandpirate
Lasiopogon polensis: Colorado Sandpirate
Lasiopogon primus: Northwestern Sandpirate
Lasiopogon pugeti: Puget Sound Sandpirate
Lasiopogon puyallupi: Salish Sea Sandpirate
Lasiopogon quadrivittatus: Great Plains Sandpirate
Lasiopogon ripicola: Columbia Basin Sandpirate
Lasiopogon schizopygus: Southeastern Sandpirate
Lasiopogon shermani: Red-legged Sandpirate
Lasiopogon sierra: Sierra Sandpirate
Lasiopogon slossonae: Streamside Sandpirate
Lasiopogon terricola: Little Reddish Sandpirate
Lasiopogon testaceus: Rust-tailed Sandpirate
Lasiopogon tetragrammus: Great Lakes Sandpirate
Lasiopogon trivittatus: Rocky Mountain Sandpirate
Lasiopogon tumulicola: Dune Sandpirate
Lasiopogon wilcoxi: Wilcox Sandpirate
Lasiopogon willametti: Willamette Sandpirate
Lasiopogon woodorum: Ohio Sandpirate
Lasiopogon yukonensis: Yukon Sandpirate
Lasiopogon zonatus: Banded Sandpirate
Leptogaster aegra: Red-backed Pixie
Leptogaster altacola: Highland Pixie
Leptogaster arborcola: Tree-twig Pixie
Leptogaster arenicola: Sandlands Pixie
Leptogaster arida: Common Western Pixie
Leptogaster atridorsalis: Spot-tailed Pixie
Leptogaster brevicornis: Short-horned Pixie
Leptogaster californica: California Pixie
Leptogaster carolinensis: Carolina Pixie
Leptogaster coloradensis: Colorado Pixie
Leptogaster cultaventris: Band-bellied Pixie
Leptogaster eudicrana: Southwestern Pixie

Leptogaster flavipes: Yellow-legged Pixie
Leptogaster fornicata: Northwestern Pixie
Leptogaster hesperis: Oak Creek Pixie
Leptogaster hirtipes: Tufted Pixie
Leptogaster incisuralis: Black-banded Pixie
Leptogaster lanata: Woolly-faced Pixie
Leptogaster lerneri: Florida Pixie
Leptogaster murina: Mousey Pixie
Leptogaster nitoris: Shiny-tailed Pixie
Leptogaster obscuripennis: Brown-winged Pixie
Leptogaster obscuripes: Cuba Pixie
Leptogaster panda: Panda Pixie
Leptogaster parvoclava: Melagra Pixie
Leptogaster patula: Atascosa Pixie
Leptogaster salvia: Sagebrush Pixie
Leptogaster schaefferi: Brownsville Pixie
Leptogaster texana: Texas Pixie
Leptogaster virgata: Stripe-backed Pixie
Leptopteromyia americana: Southern Pixie
Leptopteromyia mexicanae: Mexico Pixie
Lestomyia atripes: Black-legged Bristleback
Lestomyia fraudigera: California Bristleback
Lestomyia montis: Mountain Bristleback
Lestomyia sabulona: Northern Bristleback
Lestomyia strigipes: Wyoming Bristleback
Lestomyia unicolor: Arizona Bristleback
Machimus adustus: Sunburned Bladetail
Machimus antimachus: Yellow-legged Bladetail
Machimus aridalis: Aridland Bladetail
Machimus autumnalis: Autumn Bladetail
Machimus blantoni: Panhandle Bladetail
Machimus callidus: Western Montane Bladetail
Machimus citus: Arizona Bladetail
Machimus coleus: Azusa Bladetail
Machimus delusus: Grassland Bladetail
Machimus erythocnemius: White-spined Bladetail
Machimus fattigi: Red-legged Bladetail
Machimus floridensis: Florida Bladetail
Machimus formosus: Golden Bladetail
Machimus frosti: Carolina Bladetail
Machimus gilvipes: Colorado Bladetail
Machimus grantae: Oregon Bladetail
Machimus griseus: Gray Bladetail
Machimus hubbelli: Sandhill Bladetail
Machimus johnsoni: Pennsylvania Bladetail
Machimus latapex: Alhambra Bladetail
Machimus lecythus: Brown Bladetail
Machimus longipennis: Long-winged Bladetail
Machimus maneei: Black-legged Bladetail
Machimus notatus: Black-thighed Bladetail
Machimus notialis: Big Bear Bladetail
Machimus novaescotiae: Nova Scotia Bladetail
Machimus occidentalis: Western Bladetail
Machimus paropus: Black-spined Bladetail
Machimus polyphemi: Gopher Tortoise Bladetail
Machimus prairiensis: Prairie Bladetail
Machimus sadyates: Shiny-sided Bladetail
Machimus sestertius: Oregon Bladetail
Machimus snowii: Snow Bladetail
Machimus stanfordae: Stanford Bladetail
Machimus vescus: Little Western Bladetail
Machimus virginicus: Virginia Bladetail
Mallophora atra: Black Beebandit
Mallophora bomboides: Florida Beebandit
Mallophora fautrix: Golden-tailed Beebandit
Mallophora leschenaulti: Beelzebub Beebandit
Mallophora orcina: Southern Beebandit
Megaphorus acrus: Oklahoma Beebandit
Megaphorus clausicellus: Eastern Beebandit
Megaphorus frustra: Brown-winged Beebandit
Megaphorus guildiana: Prairie Beebandit
Megaphorus intermedius: Colorado Beebandit
Megaphorus laphroides: Kentucky Beebandit
Megaphorus lascrucensis: Las Cruces Beebandit
Megaphorus martinorum: Martin Beebandit
Megaphorus megachile: Baja Beebandit
Megaphorus minutus: Tiny Beebandit
Megaphorus pallidus: Pale Beebandit
Megaphorus prudens: Oracle Beebandit
Megaphorus pulcher: White-tipped Beebandit
Megaphorus willistoni: Northern Beebandit
Metadioctria parvula: Little Longhorn Robber
Metadioctria resplendens: Resplendent Longhorn Robber
Metadioctria rubida: Red Longhorn Robber
Metapogon amargosae: Armagosa Spotwing
Metapogon carinatus: Maned Spotwing
Metapogon gibber: Seabeach Spotwing
Metapogon gilvipes: Escondido Spotwing
Metapogon holbrooki: Arizona Spotwing
Metapogon hurdi: Spot-tailed Spotwing
Metapogon obispae: Obispo Spotwing
Metapogon pictus: Painted Spotwing
Metapogon punctipennis: Southwestern Spotwing
Metapogon tarsalus: Red-footed Spotwing
Metapogon tricellus: Three-celled Spotwing
Microstylum galactodes: Gray Titan
Microstylum morosum: Dark Titan
Myelaphus lobicornis: Northern Longhorn Robber
Myelaphus melas: California Longhorn Robber
Nannocyrtopogon antennatus: Funnel-horned Bandit
Nannocyrtopogon aristatus: Colorado Bandit
Nannocyrtopogon arnaudi: Arnaud Bandit
Nannocyrtopogon atripes: Western Bandit
Nannocyrtopogon bruneri: El Dorado Bandit
Nannocyrtopogon cerussatus: Sonoma Bandit
Nannocyrtopogon crumbi: Pinal Bandit
Nannocyrtopogon deserti: Desert Bandit
Nannocyrtopogon howlandi: Gavilan Bandit
Nannocyrtopogon inyoi: Club-horned Bandit
Nannocyrtopogon irvinei: Ridge-faced Bandit
Nannocyrtopogon jbeameri: San Benito Bandit

Nannocyrtopogon lestomyiformis: Bristle-backed Bandit
Nannocyrtopogon mingusi: Great Basin Bandit
Nannocyrtopogon minutus: Tiny Bandit
Nannocyrtopogon monrovia: Monrovia Bandit
Nannocyrtopogon neoculatus: Pinyon Bandit
Nannocyrtopogon nevadensis: Nevada Bandit
Nannocyrtopogon nigricolor: Black Bandit
Nannocyrtopogon nitidus: Shining Bandit
Nannocyrtopogon oculatus: Eyed Bandit
Nannocyrtopogon richardsoni: Butte Bandit
Nannocyrtopogon sequoia: Sequoia Bandit
Nannocyrtopogon stonei: Stripe-faced Bandit
Nannocyrtopogon timberlakei: Oro Grande Bandit
Nannocyrtopogon tolandi: Toland Bandit
Nannocyrtopogon vanduzeei: Gray-tailed Bandit
Nannocyrtopogon vandykei: Van Dyke Bandit
Nannodioctria albicornis: Sequoia Longhorn Robber
Nannodioctria seminole: Seminole Longhorn Robber
Negasilus astutus: Cunning Bladetail
Negasilus belli: Bell Bladetail
Negasilus gramalis: Alberta Bladetail
Negasilus platycerus: Broad-horned Bladetail
Neoitamus affinis: Pacific Bentbristle
Neoitamus brevicomus: Northwestern Bentbristle
Neoitamus coquillettii: Coquillett Bentbristle
Neoitamus flavofemoratus: Yellow-thighed Bentbristle
Neoitamus orphne: Dark Bentbristle
Neoitamus terminalis: California Bentbristle
Neomochtherus albicomus: Pale Bladetail
Neomochtherus angustipennis: Narrow-winged Bladetail
Neomochtherus auricomus: Golden-haired Bladetail
Neomochtherus californicus: California Bladetail
Neomochtherus comosus: Hairy Bladetail
Neomochtherus idahoae: Idaho Bladetail
Neomochtherus lassenae: Cascade Bladetail
Neomochtherus latipennis: Broad-winged Bladetail
Neomochtherus lepidus: Elegant Bladetail
Neomochtherus montanus: Mountain Bladetail
Neomochtherus pallipes: European Bladetail
Neomochtherus piceus: Black Bladetail
Neomochtherus willistoni: Williston Bladetail
Nevadasilus auriannulatus: Golden Western Assassin
Nevadasilus blantoni: Autumn Western Assassin
Nicocles abdominalis: Red-tailed Silvertip
Nicocles aemulator: California Silvertip
Nicocles argentatus: Silver-legged Silvertip
Nicocles bromleyi: Arizona Silvertip
Nicocles canadensis: Canada Silvertip
Nicocles dives: Western Silvertip
Nicocles engelhardti: Carolina Silvertip
Nicocles lomae: Loma Silvertip
Nicocles pictus: Winter Silvertip
Nicocles politus: Eastern Silvertip
Nicocles pollinosus: Banded Silvertip
Nicocles reinhardi: Texas Silvertip
Nicocles rufus: Red Silvertip
Nicocles utahensis: Utah Silvertip
Ommatius baboquivari: Great Plumetop
Ommatius beameri: Least Plumetop
Ommatius bromleyi: Yellow-legged Plumetop
Ommatius floridensis: Florida Plumetop
Ommatius gemma: Glittering Plumetop
Ommatius maculatus: Stripe-backed Plumetop
Ommatius oklahomensis: Oklahoma Plumetop
Ommatius ouachitensis: Ouachita Plumetop
Ommatius parvulus: Madrean Plumetop
Ommatius pretiosus: Red-tailed Plumetop
Ommatius texanus: Texas Plumetop
Ommatius tibialis: Northeastern Plumetop
Ommatius wilcoxi: Southeastern Plumetop
Omninablautus arenosus: Sand Bandit
Omninablautus nigripes: Black-legged Bandit
Omninablautus nigronotum: Black-backed Bandit
Omninablautus tolandi: Palm Springs Bandit
Orrhodops americanus: Arizona Red Eye
Orthogonis stygia: Blackstabber
Ospriocerus aeacidinus: Kansas Rusty Robber
Ospriocerus aeacus: Red-tailed Rusty Robber
Ospriocerus arizonensis: Clear-winged Rusty Robber
Ospriocerus brevis: Little Rusty Robber
Ospriocerus ebyi: Rio Grande Rusty Robber
Ospriocerus galadae: Galad Rusty Robber
Ospriocerus latipennis: Broad-winged Rusty Robber
Ospriocerus longulus: Long-tailed Rusty Robber
Ospriocerus minos: Black Rusty Robber
Ospriocerus nitens: Polished Rusty Robber
Ospriocerus parksi: Spot-backed Rusty Robber
Ospriocerus pumilus: Dwarf Rusty Robber
Ospriocerus rhadamanthus: Rhadamanthus Rusty Robber
Ospriocerus tenebrosus: Shadowy Rusty Robber
Ospriocerus tequilae: Tequila Rusty Robber
Ospriocerus vallensis: Idaho Rusty Robber
Ospriocerus villus: Shiny-faced Rusty Robber
Parataracticus cuyamus: Cuyama Spot-tailed Assassin
Parataracticus melanderi: Melander Spot-tailed Assassin
Parataracticus niger: Black Spot-tailed Assassin
Parataracticus rubens: Washington Spot-tailed Assassin
Parataracticus rubidus: Red Spot-tailed Assassin
Parataracticus wyliei: California Spot-tailed Assassin
Philonicus fuscatus: River Ruffian
Philonicus limpidipennis: Clear-winged Ruffian
Philonicus plebeius: Southwestern Ruffian
Philonicus rufipennis: Great Plains Ruffian
Plesiomma unicolor: Northern Wasp Robber
Pogonosoma dorsatum: Eastern Black Chiselmouth
Pogonosoma ridingsi: Black Chiselmouth
Polacantha arcuata: Arizona Twilight Robber
Polacantha composita: Western Twilight Robber
Polacantha gracilis: Southeastern Twilight Robber
Polacantha grossa: Texas Twilight Robber

Polacantha pegma: Black Twilight Robber
Polacantha sinuosa: Chisos Twilight Robber
Pritchardomyia vespoides: Hornet Robber
Proctacanthella cacopiloga: White-tipped Marauder
Proctacanthella exquisita: Exquisite Marauder
Proctacanthella leucopogon: White-faced Marauder
Proctacanthella robusta: Mexico Marauder
Proctacanthella tolandi: California Marauder
Proctacanthella wilcoxi: Wilcox Marauder
Proctacanthus brevipennis: Short-winged Marauder
Proctacanthus coquillettii: Kelso Dunes Marauder
Proctacanthus duryi: Ohio Marauder
Proctacanthus fulviventris: White-sand Marauder
Proctacanthus gracilis: Violin Marauder
Proctacanthus heros: Giant Marauder
Proctacanthus hinei: Western Red-tailed Marauder
Proctacanthus longus: Long-winged Marauder
Proctacanthus micans: Mottled Marauder
Proctacanthus milbertii: Common Marauder
Proctacanthus nearno: Desert Marauder
Proctacanthus nigriventris: Black-bellied Marauder
Proctacanthus nigrofemoratus: Black-thighed Marauder
Proctacanthus occidentalis: Western Marauder
Proctacanthus philadelphicus: Northeastern Marauder
Proctacanthus rodecki: Great Plains Marauder
Proctacanthus rufus: Eastern Red-tailed Marauder
Prolatiforceps fulviventris: Huachuca Bladetail
Prolatiforceps thulia: Grand Canyon Bladetail
Prolepsis tristis: Northern Tyrant
Promachella pilosa: Sonoran Lion Fly
Promachus albifacies: White-faced Lion Fly
Promachus aldrichii: Aldrich Lion Fly
Promachus atrox: Chocolate Lion Fly
Promachus bastardii: Northeastern Lion Fly
Promachus dimidiatus: Great Plains Lion Fly
Promachus fitchii: Prairie Lion Fly
Promachus giganteus: Giant Lion Fly
Promachus hinei: Maroon-legged Lion Fly
Promachus magnus: Mexico Lion Fly
Promachus minusculus: Little Lion Fly
Promachus nigrialbus: Southwestern Lion Fly
Promachus nigropilosus: Black-haired Lion Fly
Promachus oklahomensis: Oklahoma Lion Fly
Promachus painteri: Black Lion Fly
Promachus princeps: Gray Lion Fly
Promachus quadratus: Obscure Lion Fly
Promachus rufipes: Eastern Lion Fly
Promachus sackeni: Sacken Lion Fly
Promachus texanus: Texas Lion Fly
Promachus truquii: Arizona Lion Fly
Promachus vertebratus: Spot-tailed Lion Fly
Pseudorus distendens complex: Mexican Fancyfoot
Psilocurus birdi: Southeastern Shorehunter
Psilocurus modestus: Great Plains Shorehunter
Psilocurus nudiusculus: Golden Shorehunter
Psilocurus puellus: Desert Shorehunter
Psilocurus pygmaeus: Little Shorehunter
Psilocurus reinhardi: Red-legged Shorehunter
Psilocurus tibialis: Texas Shorehunter
Psilonyx annulatus: Ringed Pixie
Rhadiurgus variabilis: Boreal Assassin
Saropogon abbreviatus: Short-tailed Raider
Saropogon albifrons: White-faced Raider
Saropogon birdi: Oklahoma Raider
Saropogon bryanti: Desert Raider
Saropogon combustus: Great Plains Raider
Saropogon coquillettii: New Mexico Raider
Saropogon dispar: Dark Raider
Saropogon fletcheri: Pale Red Raider
Saropogon hyalinus: Clear-winged Raider
Saropogon hypomelas: Red-tailed Raider
Saropogon laparoides: Texas Raider
Saropogon luteus: Gold-faced Raider
Saropogon mohawki: Mohawk Raider
Saropogon nitidus: Shiny-sided Raider
Saropogon pritchardi: Red-legged Raider
Saropogon purus: Broad-winged Raider
Saropogon pyrodes: Fiery Raider
Saropogon semiustus: Gray-backed Raider
Saropogon senex: Black Raider
Saropogon solus: Red Raider
Scarbroughia delicatula: Delicate Bladetail
Scleropogon bradleyi: Red Rusty Robber
Scleropogon cinerascens: Ashy Rusty Robber
Scleropogon coyote: Coyote Rusty Robber
Scleropogon dispar: Patagonia Rusty Robber
Scleropogon duncani: New Mexican Rusty Robber
Scleropogon floridensis: Florida Rusty Robber
Scleropogon haigi: Arizona Rusty Robber
Scleropogon helvolus: Tawny Rusty Robber
Scleropogon huachucanus: Huachuca Rusty Robber
Scleropogon indistinctus: Southwestern Rusty Robber
Scleropogon kellogi: Golden Rusty Robber
Scleropogon neglectus: Gray Rusty Robber
Scleropogon picticornis: Spot-sided Rusty Robber
Scleropogon similis: Nebraska Rusty Robber
Scleropogon subulatus: Southeastern Rusty Robber
Scleropogon texanus: Texas Rusty Robber
Sintoria cazieri: Cazier Sintoria
Sintoria cyanea: Blue Sintoria
Sintoria mojavae: Mojave Sintoria
Sintoria pappi: Texas Sintoria
Stackelberginia cerberus: Hell-hound Sandpirate
Stenopogon adelantae: Adelanto Rusty Robber
Stenopogon albibasis: Little Rusty Robber
Stenopogon antoniae: San Bernardino Rusty Robber
Stenopogon bakeri: Claremont Rusty Robber
Stenopogon bartonae: Barton Rusty Robber
Stenopogon blaisdalli: Coronado Rusty Robber
Stenopogon boharti: Yuma Rusty Robber
Stenopogon breviusculoides: Monterey Rusty Robber
Stenopogon breviusculus: San Diego Rusty Robber

Stenopogon bromleyi: Bromley Rusty Robber
Stenopogon brookmani: Brookman Rusty Robber
Stenopogon californiae: California Rusty Robber
Stenopogon californioides: Slender Rusty Robber
Stenopogon cazieri: Black Rusty Robber
Stenopogon diablae: Diablo Rusty Robber
Stenopogon engelhardti: Crested Rusty Robber
Stenopogon englandi: Silverado Rusty Robber
Stenopogon felis: Feline Rusty Robber
Stenopogon figueroae: Figueroa Rusty Robber
Stenopogon gratus: Alameda Rusty Robber
Stenopogon inquinatus: Common Rusty Robber
Stenopogon inyae: Inyo Rusty Robber
Stenopogon jubatoides: Contra Costa Rusty Robber
Stenopogon jubatus: Crested Rusty Robber
Stenopogon jurupae: Jurupa Rusty Robber
Stenopogon kirkwoodi: Santa Barbara Rusty Robber
Stenopogon linsleyi: Linsley Rusty Robber
Stenopogon lomae: Riverside Rusty Robber
Stenopogon macswaini: Tanbark Rusty Robber
Stenopogon martini: Parma Rusty Robber
Stenopogon melanderi: Melander Rusty Robber
Stenopogon mojavae: Mojave Rusty Robber
Stenopogon neojubatus: Santa Rosa Rusty Robber
Stenopogon nigritulus: Los Angeles Rusty Robber
Stenopogon obispae: Obispo Rusty Robber
Stenopogon obscuriventris: Tan-tailed Rusty Robber
Stenopogon ozenae: Ozena Rusty Robber
Stenopogon pinyonae: Pinyon Rusty Robber
Stenopogon powelli: Pozo Rusty Robber
Stenopogon propinquus: Red-haired Rusty Robber
Stenopogon rafaelae: La Mesa Rusty Robber
Stenopogon rufibarbis: Orange-bearded Rusty Robber
Stenopogon rufibarboides: Sequoia Rusty Robber
Stenopogon tolandi: Lone Pine Rusty Robber
Stenopogon utahensis: Utah Rusty Robber
Stenopogon wilcoxi: Wilcox Rusty Robber
Stenopogon williamsi: San Diego Rusty Robber
Stichopogon abdominalis: Florida Pirate
Stichopogon arenicola: Golden Pirate
Stichopogon argenteus: Silvery Pirate
Stichopogon californica: California Pirate
Stichopogon catulus: Madrean Pirate
Stichopogon colei: Great Plains Pirate
Stichopogon coquillettii: Silver-faced Pirate
Stichopogon fragilis: Tiny Pirate
Stichopogon trifasciatus: Three-banded Pirate
Stichopogon venturiensis: Ventura Pirate
Taracticus octopunctatus: Eight-spotted Rainbow Robber
Taracticus paulus: California Rainbow Robber
Taracticus ruficaudus: Red-tailed Rainbow Robber
Tipulogaster glabrata: Shellac-backed Pixie
Townsendia albomacula: Spot-tailed Micropirate
Townsendia arenicola: Scrub Micropirate
Townsendia dilata: Mexico Micropirate
Townsendia nigra: Black-tailed Micropirate
Townsendia pulcherrima: Texas Micropirate
Triorla interrupta: Northern Triorla
Wilcoxia apache: Apache Bandit
Wilcoxia cinerea: Black-tailed Bandit
Wilcoxia flavipennis: Yellow-winged Bandit
Wilcoxia forbesi: Forbes Bandit
Wilcoxia martinorum: Martin Bandit
Wilcoxia monae: Brown-winged Bandit
Wilcoxia painteri: Painter Bandit
Wilcoxia pollinosa: Thin-tailed Bandit
Willistonina bilineata: Williston Assassin
Wyliea mydas: Mydas Bronzewing
Zabrops flavipilis: Yellow-haired Zabrops
Zabrops tagax: Thieving Zabrops
Zabrops wilcoxi: Wilcox Zabrops

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HISTORICAL DIPTEROLOGY

1467 papers and counting: Dalcy de Oliveira Albuquerque's 40-year legacy in Dipterology

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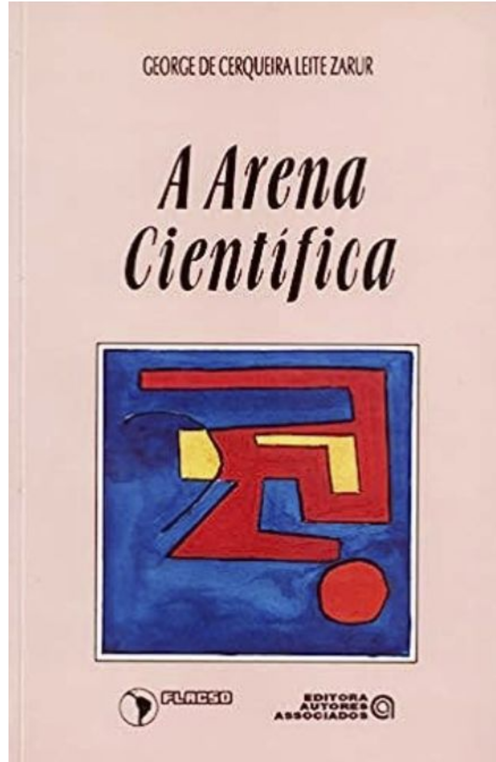
A few months ago, we realized that there were limited historical data published on the significance of the most prominent professors of Entomology in Brazil. We believe that this information is crucial for the new generations of students, as it may help answer a simple question students may ask themselves: "What was the academic foundation of my training as a Diptera researcher and teacher?" This is the focus of this study.

Fig. 1. Dalcy de Oliveira Albuquerque – Museu Nacional, Rio de Janeiro.

Dalcy de Oliveira Albuquerque (Fig. 1) was born in Cuiabá, the capital of Mato Grosso, Brazil. He graduated from Veterinary Medicine and soon engaged at the Museu Nacional, Universidade Federal do Rio de Janeiro, in 1944 to study South American Diptera (Lopes et al. 1997). He was supervised by Professor Hugo de Souza Lopes (Fig. 2), a specialist in Diptera of the family Sarcophagidae, who indicated that Albuquerque was studying Muscidae flies because, at that time, no Brazilian taxonomist was studying this rich family. Professor Hugo, along with many other professionals and their students, was part of the so-called Escola Travassos (Fig. 3), an informal school of scientific thought developed by Professor Lauro Pereira Travassos in the early 1930s (de Carvalho 2016). Lauro Travassos was a researcher in Manguinhos and an enthusiastic Professor of Zoology in the Escola Nacional de Veterinária in Rio de Janeiro (Zarur 1994). He attracted many students to his laboratory, many of whom became renowned scientists who inspired countless others, creating a multiplier effect. These scientists transmitted the principles of dedication to science, competence, honesty, and companionship learned throughout their academic and scientific lives to the next generation of students (Zarur 1994; de Carvalho 2016).



Figs. 2–3. 2 (left). Right to left: Hugo de Souza Lopes, Claudio J. B. de Carvalho, and Regina C. Z. de Carvalho (Brazilian Congress of Zoology, Londrina, 1990). 3 (right). The Arena científica book (1994).

Professor Dalcy described 126 new species throughout his scientific life—7 Anthomyiidae, 15 Fanniidae, 86 Muscidae, 2 Piophilidae, 2 Psilidae, 1 Sapromyzidae (=Lauxaniidae), and 1 Scathophagidae species—in 88 papers (Lopes et al. 1997). During his scientific career Professor Dalcy spent two years in the National Museum of Natural History in Paris under a Guggenheim Fellowship, working together with Eugène Sèguy. Some years later he also spent a couple of years in the Smithsonian's National Museum of Natural History in Washington collaborating with Curtis Sabrosky, both notable authorities in the field of Dipterology (de Carvalho 2000). After he returned to Brazil, in 1962, he was invited to be the Director of the Museu Paraense Emílio Goeldi in Belem, in the northern region of Brazil, where he remained in this position for six years. One of his main proposals was to encourage students from the Amazon region to conduct research in entomology. In that time, Therezinha de Jesus Pimentel Chaves was supervised by him in studying the taxonomy of flies (Overall & Gorayeb 1981). After his return to Rio de Janeiro (Fig. 4) in the early 1970s he acted as the Director of Museu Nacional for four years. After this period, he felt very motivated to create a new laboratory for the study of the Neotropical Diptera and to increase their scientific collection. He initiated a

Fig. 4. The new front of the Museu Nacional rebuilt after the 2018 fire (2023).

new selection process for students in various universities in Rio de Janeiro. This group of students included Claudio José Barros de Carvalho (CJBdeC), Denise Medeiros Pamplona, Márcia Souto Couri (MSC), Kátia Medeiros, and Sonia Maria Lopes. Those students were considered as the Dalcy F1 students and they initiated the Dalcy legacy. Professor Dalcy passed away in October 1982 due to natural causes in Rio de Janeiro, aged 64.

Organization and analysis of the data

We used a social network approach to understand how Prof. Dalcy's scientific legacy developed over time. Using this approach, we could map all connections, based on mentoring and publication, among all scientific descendants of Professor Dalcy's first-graduate students (F1). The data presented here were extracted from the Lattes Curriculum Vitae platform (CNPq 2023), a comprehensive Brazilian information system that integrates CV, research groups, and institutional databases into a unified information repository. The information provided was updated in December 2022. The dataset contains information on the year of paper publication, author names, paper titles, families studied, and the main areas of the respective papers.

Dalcy's legacy

Four Dalcy F1 students (de Carvalho, Couri, Pamplona, and Lopes) supervised several students, who are considered here as Dalcy's F2 students. Among them, the de Carvalho and Couri students further supervised the Dalcy F3 students, and those students in turn supervised the Dalcy F4 students. They were undergraduate, master, PhD, and postdoctoral students (Fig. 5).

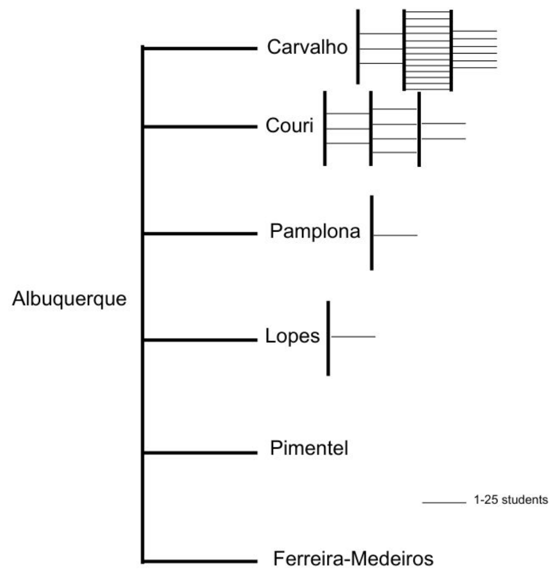
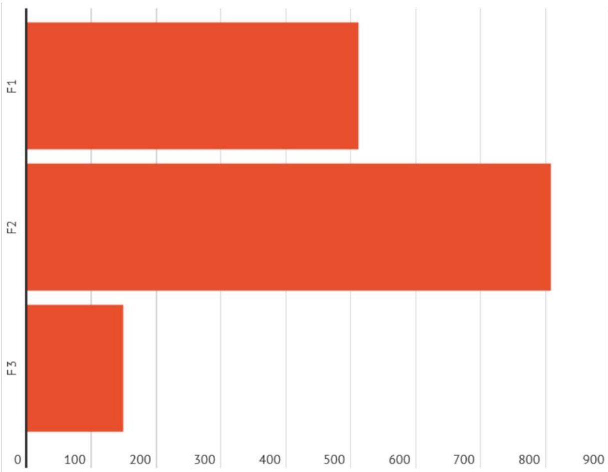
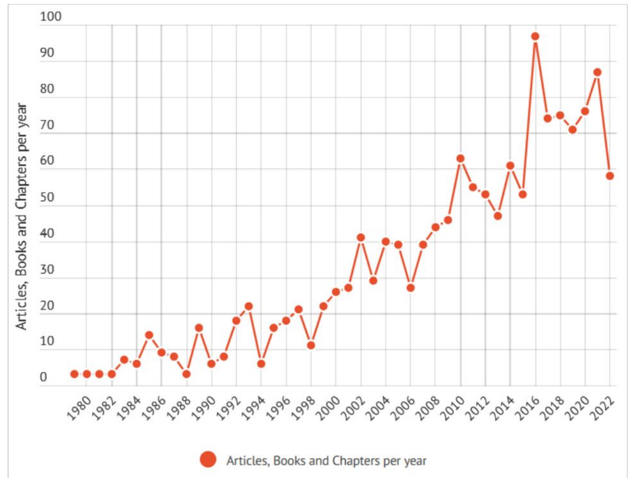


Fig. 5. Dalcy's F1, F2, F3, and F4 students.

A total of 1,467 Diptera papers were published, most of which were research on Calyptratae. Dalcy's F1 students published over 500 papers, Dalcy's F2 students published more than 800 papers, and Dalcy's F3 students made significant contributions with over 150 papers (Fig. 6). As expected, the number of papers on Diptera has increased since the late 1970s, coinciding with the growing number of students supervised by the Dalcy F1 students, and the creation of hubs of scientific connections (Fig. 7). This demonstrates that mentoring has played a dual role in fostering the rise of publications related to Diptera and catalyzing the expansion of research interests.

Dalcy students have made significant contributions across a wide range of Diptera families, with over 100 families being studied. Among these, the families with the highest number of publications were Muscidae, Cecidomyiidae, Tabanidae, Calliphoridae, and Tachinidae (Fig. 8). These families have received considerable attention from Dalcy students, demonstrating their expertise and research impact in these specific areas of knowledge. The research conducted by Dalcy's students covered various subjects, including biogeography, biodiversity, ecology, forensics, agriculture, and related fields. However, most of these investigations have been predominantly centered on taxonomy and systematics, highlighting their dedication to understanding and classifying insect species (Fig. 9).





Figs. 6–7. 6 (left). The total number of papers published on Diptera. 7 (right). The number of papers on Diptera published since the late 1970s by Dalcy's students.

Fig. 8. The number of families studied by Dalcy's students.

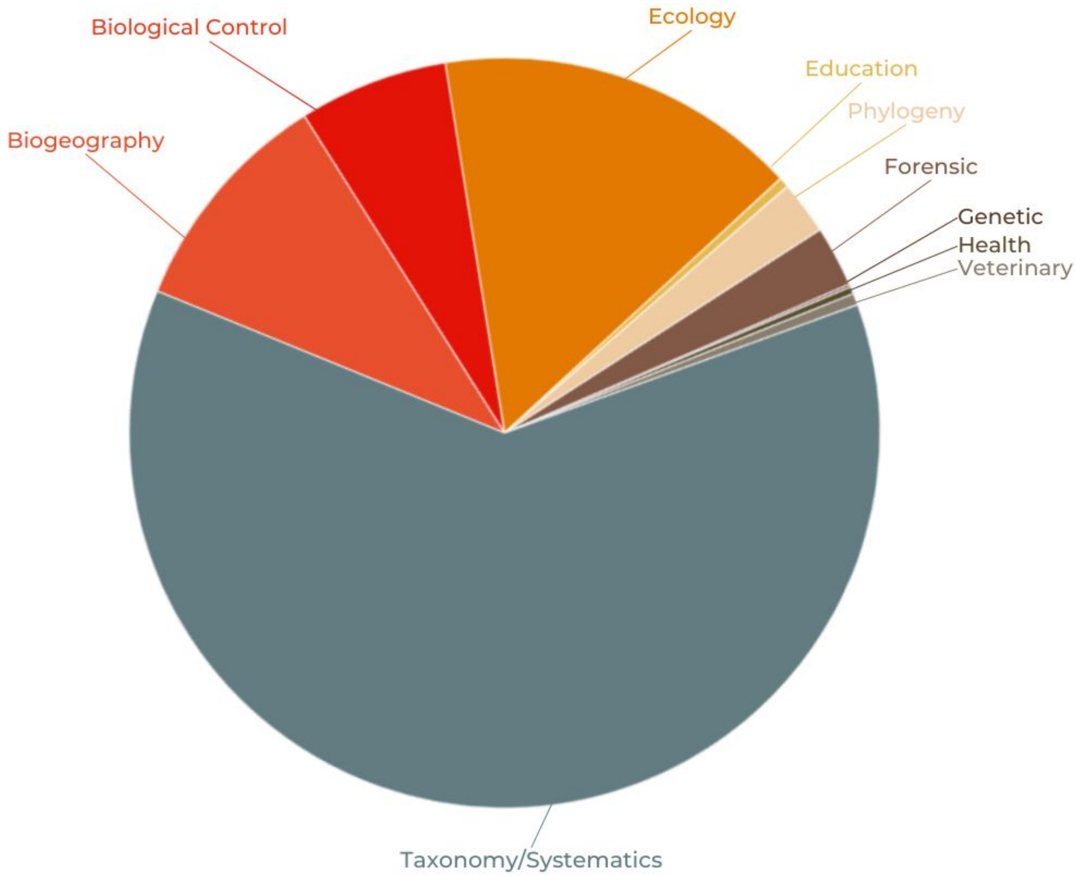


Figure 9. Distribution of topics of papers on Diptera from the late 1970s.

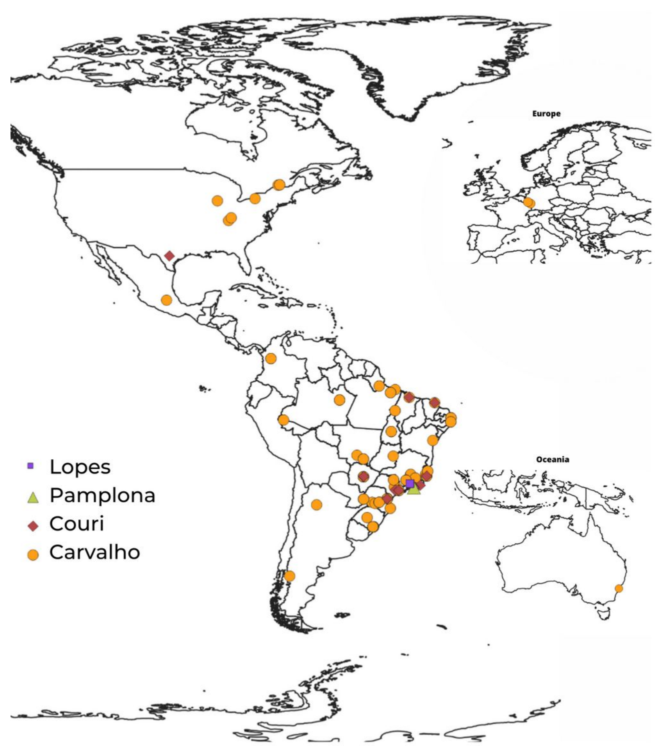


Fig. 10. Distribution of Dalcy' students and their students today.

Based on this comprehensive overview of the temporal dynamics of Diptera knowledge, the next question was: "Where are the students of Professor Dalcy's lineage and what are they doing today?" In response to this question, we mapped the geographical locations of all 714 Brazilian students specializing in Diptera and other taxa, providing a visual representation of Professor Dalcy's academic lineage on a global map. Most were from the Americas, and a few were from Europe and Australia (Fig. 10).

Several influential researchers and professors from both Brazil and abroad have had significant impacts on the development of many of Professor Dalcy's students, and we highlight a few notable names here. One such influential figure is Adrian Pont (Fig. 11). MSC and CJBdeC have been in contact since the early 1980s. At that time, the

CJBdeC sent him a letter requesting a loan from The Natural History Museum, London. Adrian promptly replied to the CJBdeC letter, and collaboration began. Subsequently, under Adrian's supervision, both the MSC and CJBdeCs had the opportunity to spend different periods in London and Oxford. MSC also had other special opportunities to benefit from Adrian's supervision and collaboration in postdoctoral programs conducted at the California Academy of Sciences, United States; the Muséum National d'Histoire Naturelle, France; and, more recently, the Museum für Naturkunde der Humboldt Universität zu Berlin, Germany.

Fig. 11. Left to right: Denise M. Pamplona, Claudio J. B. de Carvalho, Valéria Cid Maia, Adrian C. Pont, Márcia S. Couri (Museu Nacional, Rio de Janeiro, 1996).

Several colleagues and foreign researchers have collaborated on the scientific development of various generations of these students. These collaborations have opened up different avenues not only in the study of new dipteran families, in which MSC and CJBdeC are not specialists, but also in various areas of knowledge, new methodologies, and access to scientific material. In Brazil, the study of dipterans has benefited from a vast network of interactions with friends and colleagues who have made significant contributions to this legacy. There are many names to register here, and we chose to mention only Nelson Papavero from the Universidade de São Paulo, who was our teacher. In addition to Adrian Pont, other foreign colleagues have expanded our horizons. Remembering some of these names, Brian Wiegmann from North Carolina State University, USA, was important in accepting four CJBdeC students to initiate and perform molecular analysis of Muscidae, starting in the beginning of 2000. Brian also accepted an invitation to come to Curitiba to participate in the 2012 Brazilian Congress of Entomology (Fig. 12). To better understand the Diptera fauna of the Andes, Marta Wolff (Fig. 13) invited CJBdeC several times to teach courses on Diptera and Biogeography to students of the Universidad de Antioquia, Colombia. During these courses, we also had time collected flies from areas of stunning natural beauty. MSC and CJBdeC first met Marc Pollet (Fig. 14) from the Research Institute for Nature and Forest, Belgium, in 1998, at the ICD in Oxford. Subsequently, we met him several times in other ICDs, and over the years, he has been sending us valuable material from Ecuador, Chile, and French Guyana, which has significantly expanded our knowledge of South American flies.

Figs. 12–15. 12 (upper left). Brian Wiegmann, Claudio J. B. de Carvalho (Curitiba, 2012). 13 (upper right). Márcia S. Couri, Claudio J. B. de Carvalho, Marta Wolff (ICD, Potsdam, 2014). 14. (lower left). Left to right: Maurício Moura, Gustavo Graciolli, Marc Pollet, Claudio J. B. de Carvalho, Anja De Braekeleer (ICD, Brisbane, 2002). 15 (lower right). Left to right: Silvio S. Nihei, Márcia S. Couri, Guilherme Schnell e Schühli, Claudio J. B. de Carvalho, Carlos J. E. Lamas (UFPR, Curitiba, 2002).

Concluding remarks

In conclusion, we believe that Professor Dalcy's first students, along with their subsequent students, are legitimate descendants of the Travassos School, the informal school of scientific thought, influenced by many other distinguished researchers with different backgrounds from Brazil and abroad. We understand that the main contribution to society regarding the scientific education of students in Diptera is training them from the basic level of scientific initiation to the graduate level.

To date, several Dalcy students are still studying; most are working in universities, colleges, schools, and scientific or technical institutions in Brazil (Figs. 11, 14, and 15), whereas a few are working in other countries. Today, we are delighted to see students continuing to publish high-level research, with a significant portion of them in basic science and in applied science (de Carvalho 2016). Those who are already trained to continue to teach, and guide pass on to the future a philosophy of dedication, competence, honesty, and companionship that started at the beginning of the last century by Lauro Travassos in Rio de Janeiro. The greatest legacy of any researcher's career is to see their students multiplying the transmitted knowledge. And Prof. Dalcy fulfilled this mission beautifully!

Acknowledgments
We are grateful to Conselho Nacional de Desenvolvimento Científico e Tecnológico, CNPq, an agency of the Brazilian Government that fosters scientific and technological development, for the Productivity Grants to CJBdeC (process # 307959/2021–9) and MSC (process # 303414/2018–9). Many thanks to Dr. Maurício O. Moura (UFPR) for revising and greatly improving the text, and to undergraduate students Leonardo Leonardo Ricardo Nunes (UFPR) for the electronic preparation of some images and Nicole Isabelle Stocco (UFPR), who made the hard work of planning and analyzing the data and produced images from 5 to 10.

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The dating of Phillippi's *Aufzählung der chilenischen Dipteren*: 1865 or 1866?

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Rodolfo Amando [né Rudolf Amandus] Philippi (1808–1904) (Fig. 1) was a Prussian-born paleontologist and zoologist who was most famous for his contributions to malacology. However, we fly workers know him for his venture into dipterology, almost fifteen years after his 1851 arrival in Chile, where he penned his major dipterological accomplishment: a monograph on Chilean Diptera humbly entitled *Aufzählung der chilenischen Dipteren* (Fig. 2), published in the *Abhandlungen* of volume 15 of the *Verhandlungen der K.K. Zoologisch-Botanischen Gesellschaft in Wien* (hereafter *Verhandlungen*). Some 426 new nominal species and 53 new nominal genera were proposed in this work, making it one of the most significant single works on Chilean Diptera.

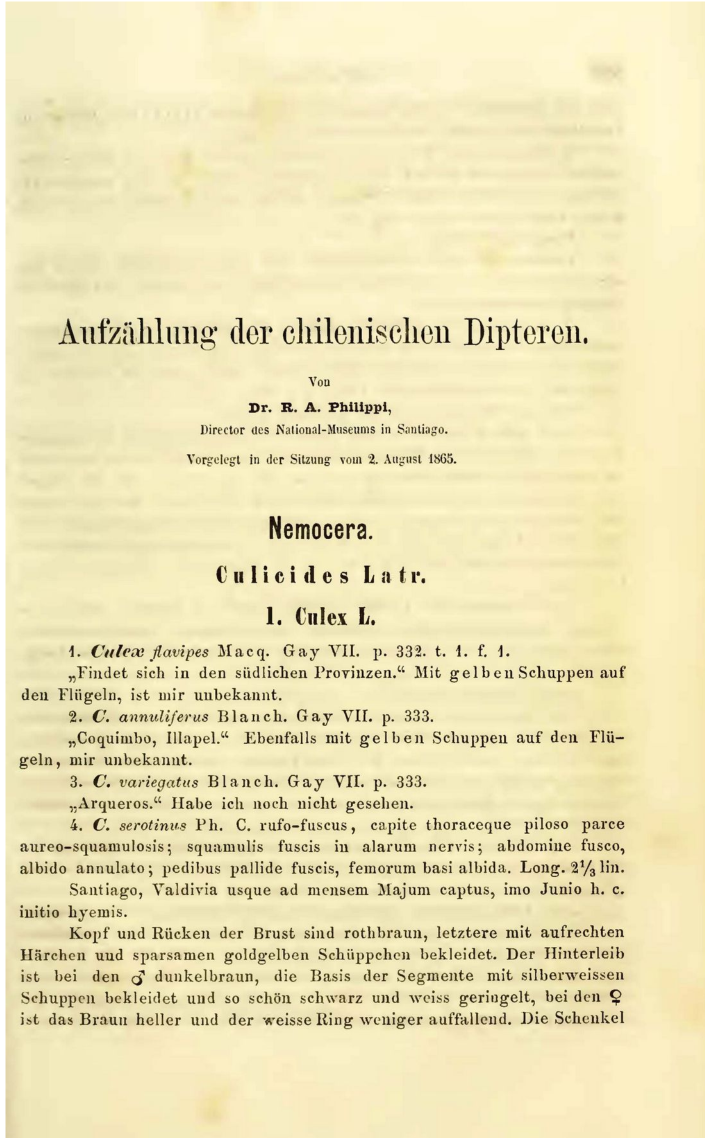

Fig. 1. R.A. Philippi. Source: Fundación R.A. Philippi, Santiago.

Philippi had submitted his paper to the *Zoologisch-Botanischen Gesellschaft in Wien* (hereafter Society) in two parts. The first was presented to the Society on 5 January 1865 (presented by Secretary G.R. Frauenfeld) and the second on 2 August 1865 (presented by J.R. Schiner).

The work has always been treated as published in 1865 based on the printed year of the journal. However, when I began working on the publication and dating of the *Verhandlungen* many years ago, I found that it was published in "Quartalen" (issues); usually 4 issues per volume but often with double issues (e.g., 1, 2–3, 4 or 1–2, 3–4). In many cases, the last issue was delayed in publication and came out early the following year. Preliminary dating research implied that was the case with the last issue (4) of volume 15 of the *Verhandlungen*, which contained Philippi's article. Unfortunately, any archival records of the Society that might have given dates of printing, receipts of issues from the printer, and/or when the issues were issued was lost in a fire in 1945 during World War II (P. Hudler, pers. comm.). Its publisher, W. Braumüller in Vienna was also to find any records of issuance. This meant relying on dates of receipt or notices in publishers' periodical literature and any information that could be obtained from the Society's own published meeting minutes. Receipts by zoological and entomological societies in European countries for this volume of the journal all were in 1866, most after June 1866. However, this was not conclusive evidence of a delay in publication, as there can be many reasons for delays in published receipts of periodicals, including those related to shipping as well as delays in recording receipt.

Fortunately, there are records within the *Verhandlungen* itself with many dates pertaining to the progress of various issues, and that year was no exception. Additionally, there are fairly consistent records of receipt of the journal by the Akademie der Wissenschaften in Wien (in its *Denkschriften*, *Sitzungsberichten* and *Anzeigen*) and the city's newspaper, the *Wiener Zeitung*. Based on information from these sources I was able to conclude that the earliest date of publication of issue 4 of volume 15 of the *Verhandlungen* was 8 March 1866 (via a notice of receipt by the Akademie der Wissenschaften in Wien). Notices or receipts within 1865 have never been found. This new dating was used by me in Evenhuis (2002).

But this 1866 date was soon confounded by the discovery by Chris Thompson of a separate of Philippi's work held in the U.S. National Agricultural Library in Beltsville, Maryland. It has a printed date of 1865 on it and Chris claimed that, despite the fact it has the same pagination as the journal, until that date could be refuted, 1865 had to be considered as the date of publication.



Aufzählung der chilenischen Dipteren.

Von

Dr. R. A. Philippi,

Director des National-Museums in Santiago.

Vorgelegt in der Sitzung vom 2. August 1865.

Nemocera.

Culicides Latr.

1. Culex L.

1. ***Culex** flavipes* Macq. Gay VII. p. 332. t. 1. f. 1.

„Findet sich in den südlichen Provinzen.“ Mit gelben Schuppen auf den Flügeln, ist mir unbekannt.

2. ***C.** annuliferus* Blanch. Gay VII. p. 333.

„Coquimbo, Illapel.“ Ebenfalls mit gelben Schuppen auf den Flügeln, mir unbekannt.

3. ***C.** variegatus* Blanch. Gay VII. p. 333.

„Arqueros.“ Habe ich noch nicht gesehen.

4. ***C.** serotinus* Ph. C. rufo-fuscus, capite thoraceque piloso parce aureo-squamulosis; squamulis fuscis in alarum nervis; abdomine fusco, albido annulato; pedibus pallide fuscis, femorum basi albida. Long. 2⅓ lin.

Santiago, Valdivia usque ad mensem Majum captus, imo Junio h. c. initio hyemis.

Kopf und Rücken der Brust sind rothbraun, letztere mit aufrechten Härchen und sparsamen goldgelben Schüppchen bekleidet. Der Hinterleib ist bei den ♂ dunkelbraun, die Basis der Segmente mit silberweissen Schuppen bekleidet und so schön schwarz und weiss geringelt, bei den ♀ ist das Braun heller und der weisse Ring weniger auffallend. Die Schenkel

Fig. 2. First page of Philippi's *Aufzahlung der chilenischen Dipteren*. Source: Biodiversity Heritage Library.

I was reluctant to accept that. The date on the separate could well have been printed as 1865 the same as the printed "1865" on the title page of the journal itself (which we know was not completed and published in full until 1866). Further research helped find a clue: I found that the minutes of the Society announced at their 3 January 1866 meeting: "Das 4. Heft der *Verhandlungen* des Jahres 1865 ist geschlossen und seine Ausgabe und Versendung wird in der 2. Hälfte des Monate Jänner beginnen können" [The 4th issue of the *Verhandlungen* of the year 1865 is closed and its publication and distribution will begin in the second half of January]. This to me meant that there could not have been an 1865 publication date for the last issue with Philippi's article, but Chris was insistent we had to use the 1865 date, saying separates could have been issued prior to the journal. I tried to find information from the volumes themselves on how separates were handled by the Society and could not find anything conclusive about issuance prior to issuance of the parts or of the completed volume. Most were issued the same year as the completed journal volume and sometimes (but not always) showed up in the society's financial reports for that year further supporting publication of separates and journal in the same year.

Then recently, I found an article by Bob Carlson [ironically, a co-worker of Chris] concerning the dating of one of the volumes of the *Verhandlungen*. And in it, there was finally an explanation. It was a letter sent to Carlson by the general and editorial secretary of the Society in 1978, Dr. Karl Burian, who stated regarding the practice of publishing separates of the *Verhandlungen* in the 19th century that: “it was not until after the complete volume had been distributed that separate prints of papers were sent to authors” (Carlson, 1980: 123). This meant that Philippi’s separate could not have been issued in 1865, but only in 1866 when the completed volume had been published. With that final piece to the puzzle, and all the other the evidence at hand, I here conclude that 1866 is the year of publication for Philippi’s work in that volume of the journal.

The following is a suggested citation for Philippi’s work:

Philippi, R.A. 1866. Aufzählung der chilenischen Dipteren. *Verhandlungen der K.K. Zoologisch-Botanischen Gesellschaft in Wien* (*Abhandlungen*) 15[1865]: 595–782. [8 March 1866*]
[*date of receipt by the Akademie der Wissenschaften in Wien]

Acknowledgments
A number of people assisted me over the years in obtaining dates of publication of the *Verhandlungen* through library or society receipts and also helped with researching publication practices of the Society: Jason Weintraub and Eileen Mathias, Academy of Natural Sciences, Philadelphia; Susanne Gföller, Braumüller Verlagsbuchhandlung, Vienna; Michael Ohl, Museum für Naturkunde, Berlin; Reinhard Gaedicke, Senckenberg Deutsches Entomologisches Institut; Gerardo Lamas, Peru, Alan Peterson, Walla Walla, Washington; Petra Hudler, Zoologisch-Botanischen Gesellschaft in Österreich. Thomas Pape kindly reviewed the manuscript and is thanked for suggestions and corrections that helped improve the paper.

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Welcome to Old Florida, Yankee!

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It's probably safe to assume that just about anyone familiar with mosquito control in the State of Florida is familiar with the words of Virginia Congressman John Randolph, who, addressing the US House of Representatives said, "Florida, sir, is not worth buying. It is a land of swamps, of quagmires, of frogs and alligators and mosquitoes! A man, sir, would not immigrate into Florida – no, not from hell itself!" (Brown 1991).

Back in 2013 I wrote a short paper describing the historical impact that mosquitoes had on the State of Florida (Hribar 2013). In the succeeding years I became curious about just how bad mosquitoes were in days of yore. After all, as Harden (1981) wrote, "Only a miniscule number … have the remotest idea of what a mosquito problem can be, or was at one time, in this state." Harden (1981) also wrote, "…they weren't around when you could not go outside after dark and most of the coastal communities closed down during the summer." Viele (1996) repeated a quote from a resident of Lower Sugarloaf Key, "…you could rake the mosquitoes off your arms in piles."

The annual publication *Tequesta*, the journal of the Historical Association of Southern Florida, contains many references to and mentions of mosquitoes within its pages. The journal mainly contains information pertinent to the Miami area, the Everglades, and the Florida Keys. There are reprinted letters that present chronologically anomalous citations, and many articles are reminiscences so they tend to ramble and statements therein are not supported in the manner that those of us who are scientists are accustomed to seeing. Nevertheless, the contents of this journal are interesting and eye-opening, and let us see southern Florida as it used to be, prior to its becoming a tourist and retirement destination. Life was difficult and a lot of that difficulty was due to mosquitoes. According to Straight (1998), heavy clothing was worn all year long regardless of temperature and fastened tightly at neck, wrists, and ankles. Smudge pots were kept burning, and mosquito nets and gauze were used liberally to avoid mosquitoes – even the Seminoles slept under "mosquito bars when they could acquire them." Later, Straight (2003) described how terrible the mosquitoes were in Miami and wrote that people sat in the smoke of smudges that were made of coconut hulls, rags, and "mosquito powder" that was bought from a local drug store. Gillis (2009) described smudges being made of palmetto roots with wet moss or wet grass. Gillis (2009) also wrote that everyone, Native Americans, European Americans, and African Americans, wore long clothing even on the hottest days. Gentry (1974) wrote that veils were worn by residents to avoid mosquito bites. In relatively modern times, Seminoles were still using mosquito netting to sleep (Covington 1976).

The following notes and citations were made by examining the online archives of the journal at Florida International University in Miami (http://digitalcollections.fiu.edu/tequesta/). A few other references are mentioned where I felt it appropriate. Volumes published from 1941 to 2003 are available online. There may be more references to mosquitoes in the journal, but volumes not accessible online have not been seen. Citations are grouped according to my idea of whether they pertain to religion, agriculture, science, railroad, medical, Bamboo Key, early exploration, news media, military, tourism, and other mentions of mosquitoes. I have also included a few other interesting facts that I found during the research for this article. These are set off with brackets.

Religion
As is well-known among mosquito control personnel, it was common in South Florida prior to mosquito control to see people carrying smudge pots to ward off mosquitoes and other biting flies (Dorn 1949, Voss 1968, Gentry 1974, Viele 1996, Gillis 2009). Mosquitoes impacted churches and church services in Miami. People carried smudge pots to church (Dorn 1949). Pennington (1992) reports that one early church in Miami had no glass in the windows; the congregation used cheesecloth to exclude mosquitoes. Parishioners called it the Church of the Holy Cheesecloth. McNicholl (1941) reproduced a letter to a Spanish priest from a Spanish religious brother. [A brother is a member of a religious institute or order who has taken monastic vows but is not ordained a deacon or priest.] In that letter, the brother mentioned mosquitoes being so bad that he and his fellows could not sleep even an hour undisturbed. The Barry family worked to establish a college for Catholic women in the southern United States. The land where Barry University now stands in Miami Shores was remarked at its purchase to be, "inhabited by mosquitoes and snakes". One of the founding faculty members, Sr. Regina Marie LaLonde, mentioned being bitten by mosquitoes upon her arrival (Rice 1989). Mosquitoes are nonsectarian in their hematophagy; Pennington (1941) writes that an early Episcopal bishop was warned about mosquitoes in the area of present-day Miami. Later, another Episcopal bishop wrote about the terrible mosquitoes (Pennington 1992). Patton (1964) also writes about mosquitoes plaguing early Episcopal missions in the area.

Agriculture
Agricultural workers in Florida often wore veils to avoid the swarms of mosquitoes (Niemiec 1996, Viele 1996). Both settlers and Seminoles in present-day Broward County had problems keeping livestock alive due to attacks by mosquitoes and horse flies (Gillis 2009). Straight (1998) reproduces comments from early settlers in the Miami River area; mosquitoes, sand flies, and "blue flies" were pestiferous and even killed chickens and young pigs. Bishopp (1933) reported an event in the Miami area in which no less than 173 animals were killed during a severe outbreak of *Psorophora columbiae*. The dead animals included 80 cattle, 67 hogs, 3 horses, 1 mule, 20 chickens, and 2 dogs. There were unverified reports of even more dead animals closer to the Everglades. Milk production in the area was reduced by 1000 gallons per day for five days and even two weeks later had not returned to normal. Even in the early days of free-ranging cattle, mosquitoes were among the problems that the old cattlemen faced (Will 1966). Pioneering families on Upper Matecumbe Key were unable to keep chickens because the birds were killed by mosquitoes (Gentry 1974). Mosquitoes were among the difficulties faced by the Japanese agricultural community at Yamato in Palm Beach County (Pozzetta and Kersey 1976).

Science
An early botanist to visit the Florida Keys, John Henry Blodgett, remarked on how terrible the "Mosketoes" were (Ledin 1953). [One interesting fact about his travels is that on an island near Key West he collected the plant *Torrubia floridana* which was said to never have been collected again. A little Internet research revealed that the name *Torrubia floridana* is a synonym of *Guapira discolor* (Nyctaginaceae) (Wunderlin et al. 2022).]

Gifford (1944) writes that "Florida quinine", *Pinckneya pubens* Michaux (Rubiaceae), was used as an antimalarial treatment. [Gifford (1944) also mentions that David Fairchild, for whom Fairchild Tropical Garden in Miami is named and father of Alexander Graham Bell ("Sandy") Fairchild, expert on Phlebotominae and Tabanidae (yes, Sandy's mother was related to Alexander Graham Bell), had a guava jelly factory on his property in Coconut Grove; it was destroyed by a hurricane in 1926.] Goggin (1944) wrote about an archaeological expedition to Key Largo on which mosquitoes interfered with the excavation of a historical site. (Smith (1933) also points out that mosquitoes and other biting flies hampered the progress of archaeological research in Florida.) Smiley (1990) speculated that the fruit of the paradise tree (*Simarouba glauca*) (Simaroubaceae) might have been used by natives to prepare an insect repellent. Smiley (1991) stocked a pond with *Gambusia* fish to prevent development of mosquitoes. He also stated that development of south Dade County was impeded by mosquitoes. An interesting (to me, at least) account of travel in south Florida is given in an article reprinted from a book written by Willis S. Blatchley, a name familiar to all students of entomology (Blatchley 1932, 1974). Blatchley rather unscientifically refers to "millions" of mosquitoes vexing his collecting trip.

Railroad

Potential railroad construction laborers were recruited partly with tales of a workplace with, "no swamps, no malaria, and no mosquitoes" (Knetsch 1999). Corliss (1953) wrote that mosquitoes and sand flies were the reason that so many railroad construction workers quit. Krome (1979) wrote about the horrible problem with mosquitoes during the surveys for the railroad.

Medical

A Charleston doctor, Benjamin Beard Strobel, arrived in Key West in 1829 to practice "medicine, surgery, and midwifery"; he arrived at an opportune time because Key West was in the midst of an outbreak of yellow fever (Hammond 1969). [Hammond also mentions a dinner of turtle, fish, and young flamingo, "cooked in a style peculiar to Key West."]

Bamboo Key

Buck (1979) wrote about mosquitoes, sand flies, fleas, and ants in the so-called "paradise" of South Florida. He also mentions that Bamboo Key is free of mosquitoes. Moznette (1924) and companions explored and did find mosquitoes on Bamboo Key, although not as many as on other islands. While on the island, the group found the remains of the foundation of a house; Brigham (1958) states that the Pent family had moved from Key Vaca to Bamboo Key sometime around 1866. The reason they moved was because Key Vaca (often erroneously called "Marathon Key") was infested with mosquitoes (Brigham 1957). [The Pent family name appears in more than one article, both in the Florida Keys and in the Miami area.] Johnson (1991) also wrote that Key Vaca settlers were attacked by mosquitoes. Brigham (1958) stated that mosquito control was one of the main reasons that Key Vaca developed.

Early Exploration

Preble (1945) recounted that early explorers were unable to sleep due to mosquitoes. Shafer (1984) writes about Frederick George Mulcaster, who came to survey after Spain ceded La Florida to Britain. In his attempt to survey the Province of Biscayne Bay he encountered "only a few Musquitos"; the low numbers, he wrote, were "very remarkable". Wintringham's (1964) account of an expedition through the Everglades contains a long passage about dealing with mosquitoes on the trip. Leonard's (1968) reportage on Kurt Munroe's canoe trip also contains mention of terrible hordes of mosquitoes, as does the report of Juan Baptista Franco (Holmes and Ware 1968). Reiger (1971) reproduces the memoirs of James Alexander Henshaw, a Maryland physician who, with several companions, traveled through central and southern Florida in the early 1880s. Henshaw encountered mosquitoes at a Seminole village and noted that the Seminoles slept under "mosquito bars." Early surveyors were warned to bring mosquito netting with them (Parks 1973). Chardon (1975) also mentions lots of mosquitoes.

News Media

An anonymous reprint of an old newspaper article (Anonymous 1960) mentions mosquitoes and deer flies in the Everglades. Very interestingly, Fleischmann (1987) reports that the local newspaper, The Miami Metropolis, tried to entice the US Government to commit more men and materiel to Miami for the Spanish-American War. The paper boasted, among other things, that Miami offered, "No malaria. No fevers. No mosquitoes." That wasn't exactly the case and things didn't stay that way for long (Hribar 2013). A similar set of illusions was used to recruit workers for the railroad – potential laborers were told of a place where there were "no swamps, no malaria, and no mosquitoes" (Knetsch 1999).

Military

Fort Dallas was established in the area of present day Miami and prospective commanders were warned about the terrible mosquito problem in the area (Shappe 1961). In 1849 the US Army reoccupied Ft. Dallas and sent a detachment of men to guard a single worker at a starch-making facility in the Everglades. The leader of the detachment, Auson J. Cooke, referred to this post as "Fort Desolation" and complained about the mosquitoes (Gaby 1988). Staubach (1993) wrote about "huge swarms of mosquitoes" in Miami during Civil War times. Mariotti (1994) also wrote that mosquitoes were so bad that soldiers could not sleep in the area that is now present-day Miami. Florida's Everglades were seen as a danger to health, producing malaria-carrying mosquitoes (Meindl 2003). This is a change in thinking from earlier times, when George Gauld, visiting what was then West Florida (present day Alabama) attributed "summer fevers and agues" to lagoons and marshland, rather than to the mosquitoes produced therein [this account appears to be written by a descendant] (Gauld 1969). This kind of thinking was still accepted medical opinion in the late 1890s (Straight 1972). Tebeau (1960) also mentions troops being bothered by mosquitoes in Key West. Covington (1968) wrote about the impact mosquitoes had on the Air Force's base at Cape Canaveral. [When the Navy and Marine Corps stationed personnel in Key West to combat piracy in the Caribbean, among their craft were the first steam vessel in the US Navy, the Sea Gull, that towed five "rowing barges" for close-in combat. The barges were named Mosquito, Gnat, Midge, Gallinipper, and Sandfly (Roth 1970).] Even as late as the World War Two years, mosquitoes were a problem in Key West (Roth 1970). Straight (1988) mentioned the possibility of old Navy sailing ships providing larval habitat for mosquitoes and transporting mosquito-borne disease.

Tourism

I wrote previously about the impact of mosquito control on tourism to Florida (Hribar 2013). It turns out that when President Chester A. Arthur was planning a public relations trip to the southern states, disguised as a fishing trip, plenty of silk fabric was purchased to serve as mosquito screening (Richardson 1964). This comment that I found, a memory of someone who grew up in old Coral Gables in old Florida, seems appropriate to close out, especially for those of us who have listened to visitors complain about mosquitoes. "Occasionally, in the rainy season, we could expect mosquitoes from the Everglades. They came in swarms on hot, humid evenings, blackening screen doors in their attempts to enter. Tourists in the winter often complained about the many bugs in the area; I would think to myself, 'They should see them in the summer'" (Kuhn 2000).

Other Mentions of Mosquitoes

Will (1959) recounts that a contractor friend of his refused to bid on the job of digging a canal at Cape Sable; among his reasons were mosquitoes and deer flies. Will (1959) makes repeated mention of mosquitoes bothering the crew. Du Bois (1960, 1968, 1973) writes about mosquitoes and "sand flies" attacking lighthouse keepers and other people in Jupiter. [Du Bois (1973) reports that one future lighthouse keeper, while in military service, had to go looking for critical parts of the lighthouse, which were stolen by Confederate sympathizers during the Civil War. He found them and after the war the Jupiter lighthouse was relit.] Pierce (1962) met with large swarms of mosquitoes on his trip from Miami to the Thousand Islands. Peters (1965) remarks on the mosquito problem in Key West. Darrow (1967) wrote about the mosquitoes around her home near Lake Okeechobee. Darrow (1967) also mentions that Ft. Pierce had the worst mosquitoes of anyplace in the area, corroborating Zora Neale Hurston's assertion that the eastern coast of Florida was the devil's country (Hurston 1935). Williams (1979) wrote about mosquitoes and horse flies in West Palm Beach. True (1946) mentions mosquitoes on Boca Chica Key. Gilpin (1947), Dovell (1948), Marchman (1957), Voss (1968), and Davenport (1980) all write about the large numbers of mosquitoes in various parts of South Florida. La Plante (1995) recounts Charles Torrey Simpson's mention of mosquitoes on Long Key in his book (Simpson 1920). Diddle (1946) even mentioned a nautical feature, Mosquito Shoal, near Tavernier in present-day Monroe County. Humes (1965) counts mosquitoes among the man hazards afflicting snail collectors in the Florida Keys. [This article tells the tale of the people who hunted tree snails for profit.] Long (1968) reports that mosquitoes were among the reasons for a Works Progress Administration (WPA) worker's strike in Key West in the 1930s. Niedhauk (1969) wrote about problem mosquitoes on Elliott Key. Ball (1970) tells the sad tale of Samuel Hodgman of Haines City, who found mosquitoes no worse than in his native Michigan. [Reading the tale of continuous misfortune, constant bad luck, chronic disease, and ever-declining health, it becomes obvious that mosquitoes were the least of Hodgman's problems.] Kent (1971) briefly mentions mosquitoes in Coconut Grove. Lundstrom (1971) encountered mosquitoes and no-see-ums on Marco Island. Parks (1975) mentioned that a construction project in Miami was completed in a substandard fashion and the builder stated that it was impossible to do good work in the presence of so many mosquitoes. Peters (1978) reproduces the log of the Biscayne House of Refuge (a shelter for shipwreck survivors) in which problem mosquitoes are mentioned. Peters (1986) later published a second article with the exact title of the first; this second paper does not mention mosquitoes. Hancock (1978) wrote of mosquito problems in the Kissimmee Valley.

Conclusion

It is obvious that mosquitoes have had a great impact on the history of Florida. Much of the state's story is intertwined with the effort to control these insects. Early inhabitants of Florida struggled against mosquitoes and the diseases they transmitted. Life today is easier, healthier, and in many cases longer than it was in times gone by. Medical care is much easier to find than it was years ago, for example, before James Jackson arrived in Miami (Straight 1972).

It is a mistake to romanticize the past, to think that people in times gone by lived in some lost paradise. Things were not necessarily better long ago, and they are not necessarily worse now; the past is just the past, and it was different (Anonymous 1976). All we know of the past is what we have learned from others who lived then, and we don't know what we don't know. We have no way of knowing if something did not happen in the past (Stokes and Keegan 1996). The intersection of entomology and history is a fascinating place, filled with such questions as whether there was malaria or yellow fever in the Americas prior to European arrival (Jarcho 1964, Dunn 1965, Steverding 2020) and if there were honey bees in North America prior to the arrival of Europeans (Weber 2012). The arrival of the British, French, and Spanish, and to a lesser extent the Dutch and the Swedes, irrevocably changed the Americas (Bianchine and Russo 1992). But then again, so did the arrival of the first human populations in the Americas (Zettler 2015). Probably a good many things happened or were seen that were never recorded. Perhaps all that can be said is, "Don't go back to the 'Good Old Days,' but remember, you wouldn't be here except for them" (Tebeau 1993).

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Diptera at the Carnegie Museum of Natural History, and an interesting encounter

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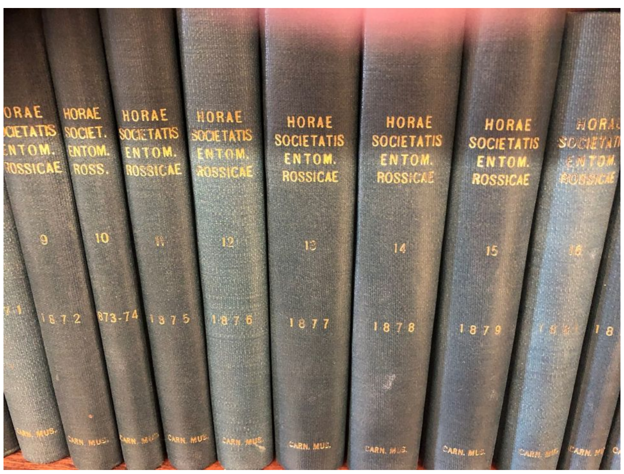
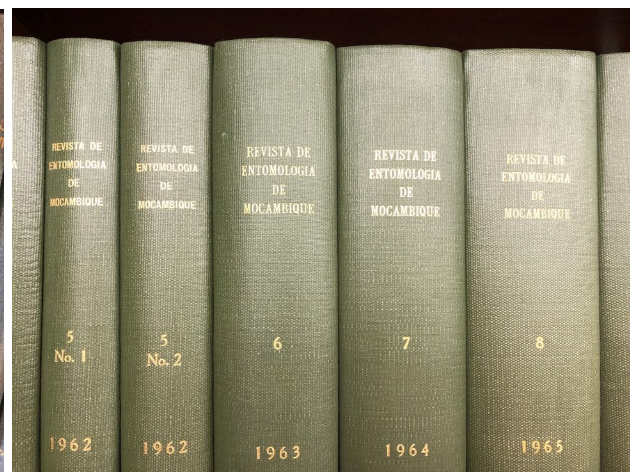
In August 2023 I spent some time visiting relatives in the Pittsburgh area. During that time I visited the entomology collection in the Carnegie Museum of Natural History. The last time I visited the collection was in the early 1980s while I was an undergraduate student. Upon my arrival I met Dr. Ainsley Seago, Associate Curator of Invertebrate Zoology, at the door and she led me to the collection. During my visit I also met the collection manager, Dr. Kevin Keegan.

The Diptera collection consists of 1,338 drawers of specimens (I think that number is correct). I counted 82 families of Diptera in the collection. Chen Young's immaculately prepared Tipulidae are definitely worth the trip in and of themselves. His specimens are absolutely perfect. The room where the Diptera are stored also contains a number of entomology books and journals, some very hard to find (Figs. 1–2).





Figs. 1–2. Some interesting journals in the Carnegie Museum.

I was somewhat surprised at how few mosquitoes were in the collection. Most of them were old specimens. There haven't been many recent additions to the mosquito collection. I counted 20 species of *Aedes*, five species of *Anopheles*, eight species of *Culex*, three of *Culiseta*, one each of *Mansonia* and *Orthopodomyia*, three *Psorophora*, and one *Toxorhynchites*. There are specimens from Bolivia, Cameroon, Costa Rica, Cuba, Dominican Republic, Alabama, Alaska, Georgia, Kansas, Mississippi, Missouri, New York, Oregon, and Pennsylvania. I did find it a little funny that the second drawer that I looked into contained mosquitoes collected in Florida. There were not too many of those. But I did see something interesting in that drawer. There was a label in that drawer that indicated some specimens were collected by E.L. Seabrook and identified by W.D Sudia (Fig. 6). E.L. Seabrook was an entomologist who worked for the Palm Beach County Anti-Mosquito Control District (according to Pritchard et al. 1947). W.D. Sudia worked for the CDC for many years, mostly in the Atlanta facility with a few years in Ft. Collins.

W. Daniel Sudia seems to have been an interesting character. Details of his life were taken from obituaries (Anonymous 2010, Comer and Calisher 2011). He was born near Pittsburgh, in Ambridge, PA, the town where I went to high school. He was one of eight children born to Feydor J. Sudia (who became Frank in the USA) & Paraskeva (Paraska) Staroska Sudia (who may have also been known as Mary). Both of Dan Sudia's parents were ethnic Ukrainians born in Galicia, then part of the Austro-Hungarian Empire but now part of Poland. Their birth villages were Tilawa and Chestohorb, respectively. They did not know each other in the "old country" but after immigrating to the USA via Ellis Island, they met married while working at a lumber camp in Cross Fork, PA, located in Potter County. The family moved to Ambridge at some point, where Dan was born. His parents are buried in Saints Peter and Paul Ukrainian Catholic cemetery just outside of Ambridge, PA (Anonymous undated).

Fig. 3. Mosquitoes identified by W.D Sudia

Dr. Sudia received his B.S. degree from the University of Florida and his M.S. and Ph.D. degrees from the Ohio State University. Dan was an entomologist and epidemiologist who worked for the CDC and its predecessor agencies, mostly in Atlanta, GA, but for a few years in Montgomery, AL, and Ft. Collins, CO. He isolated many arboviruses from mosquitoes and discovered a new species of mosquito during his work, *Culex cedecei*. He is best remembered for his invention of the CDC miniature light trap, a device that was small, lightweight, portable, and powered by flashlight batteries (Sudia and Chamberlain 1962). This trap revolutionized arbovirus research because it could be carried into remote areas and mosquitoes (and other insects) could be trapped from habitats that were previously considered inaccessible. Dan also invented the CDC entomological chill table, that allowed maintenance of a cold chain when sorting specimens and greatly facilitated isolation of viruses (Sudia et al. 1965). During his tenure with the CDC Dan Sudia received many awards and honors during his professional life, one of them being the United States Public Health Service

Meritorious Service Medal. He published 85 scientific documents and was a consultant to several foreign governments.

Dan was also known for his hobbies. He worked with stained glass, he made the furniture in the home with his woodworking tools, assembled a renowned collection of barbed wire, and became famous for his photographs of birds. His photographs are housed in the Georgia and Florida Museums of Natural History. He was also known for his ability to grow many varieties of ornamental plants. Daniel Sudia died in 2010 due to lung cancer. He is buried in Floral Hills Memory Gardens in Tucker, GA (Anonymous undated).

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Edward Irving Coher,
November 22, 1920–July 26, 2023

Lawrence J. Hribar

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Obituary – Edward Irving Coher, 102, passed away on July 26, 2023, in Delray Beach, Florida, after a brief illness. He was born in Chicago, Illinois, USA on November 22, 1920, to Samuel Nathan and Esther Weisman Coher and spent his youth in Dorchester, Massachusetts. Esther appears to have been born in Russia. An elder brother, Shepherd Milton, was born in Minneapolis, so the family moved around until finally settling in Massachusetts. Ed was a graduate of the University of Connecticut and the University of Massachusetts, from which he received both his Master of Science and Doctor of Philosophy degrees. He also received a certificate in public health in São Paulo, Brazil, presumably at the Universidade de São Paulo. There is a Brazilian immigration record from the Port of São Paulo that gives his address as the Faculdade de Higiene e Salud de S. Paolo, where John Lane worked. Ed was also an Army veteran.

Figs. 1–3. Photographs of Edward Coher. 1 (left). Brazilian immigration documents, Rio de Janeiro. 2 (middle). Brazilian immigration documents, São Paulo. 3 (right). Unknown source.

Ed was an entomologist with the World Health Organization and he worked in Afghanistan, Nepal, and Thailand. While with WHO he started the original Asian Biting Fly Study. He studied mosquitoes, horse flies, winter crane flies, and fungus gnats. He provided Tipulidae material to C.P. Alexander for study; several undescribed species were found among his specimens, one of which, *Hexatoma* (*Eriocera*) *coheri*, was named for Ed by Dr. Alexander (Alexander 1956). Ed also provided Ephemeroptera to G.F. Edmunds and his coauthors for their magnum opus on the nymphs of mayflies (Edmunds et al. 1963). The Sciaridae that Ed collected were studied by Frank Menzel and his coauthors and new taxa described therefrom, including *Prosciara coheri* (Mohrig & Menzel 1994, Menzel & Martens 1995). Mecoptera that Ed collected in Nepal were described later as a new species (Bicha 2011). While he was working in Nepal, Ed was attacked by a leopard (he survived; the leopard did not).

Ed eventually relocated to New York's Long Island and joined the faculty of Long Island University's Southampton College. Ed taught biology classes at the university while his wife, Cynthia Varrell Coher, taught in the elementary school and acted, painted, and sang. Over 100 of her paintings were purchased by collectors. Among his friends Ed was known as an adept bridge and poker player. He retired to Boynton Beach, Florida, sometime around 2006; he had been an emeritus professor at LIU since 1985. After the death of his wife he moved into an assisted living community in Delray Beach. He is survived by one niece and four nephews. Services were private. I have no information regarding disposition of his remains. I contacted the cemetery in Rye, New Hampshire, his wife's hometown, where she was interred and where Ed liked to vacation. I was informed that he is not interred there.

Ed Coher was a research associate with the Florida State Collection of Arthropods (FSCA). He maintained a large personal collection of Diptera, much of which was donated to the FSCA prior to his passing. One donation consisted of over 3,000 pinned specimens and hundreds of microscope slides, including numerous holotypes and paratypes. A second donation of nearly 4,000 specimens was made about 13 years later. He also donated over 100 books and a number of reprints to the FSCA. Ed was formerly a member of North American Dipterists Society. I met him through the old membership directory when I was looking for someone to help me identify a strange fungus gnat that I found. It turned out to be a species new to science and he named it after me, my first patronym. We met in person a couple of times and spoke on the phone occasionally. He was an interesting individual and I always enjoyed speaking with him. When we last spoke he was in the medical center of his assisted living facility. He had spent some time in the hospital and was recovering in the medical center. He was in good spirits when we talked and I thought that he would fully recover. I have no recent photographs of Ed Coher. I intended to take at least one during one of our visits but I never did. Two of the photographs that accompany this obituary (Figs. 1–2) were taken from Ed's Brazilian immigration documents. The third (Fig. 3) was taken from his online obituary notice and I do not know the source. Following the literature citations for this note is a list of Ed Coher's publications. I thank Gary Steck, FSCA, and Frank Menzel, Senckenberg Deutsches Entomologisches Institut, for their assistance in compiling the information for this obituary.

Names proposed by Edward I. Coher

<u>Tribe – 1</u>
Chiasmoneurini (Keroplatidae)

<u>Genera – 5</u>
Aphrastomyia Coher & Lane (Mycetophilidae)
Calusamyia (Keroplatidae)
Jugazana (Mycetophilidae)
Laneocera (Mycetophilidae)
Neoepicypta (Mycetophilide)

<u>Species – 112</u>
Keroplatidae – 13
Mycetophilidae – 79
Tabanidae – 13

Patronyms

<u>Tipulidae</u>
Hexatoma (*Eriocera*) *coheri* Alexander

<u>Mycetophilidae</u>
Subgenus (*Coheromyia*) Väisänen

Dziedzickia coheri Lane
Echinopodium coheri Duret
Epicypta coheri Lane
Rymosia coheri Shaw

Sciaridae
Prosciara coheri (Mohrig & Menzel)

Tabanidae
Hybomitra coheri Xu et al.

Ephemeroptera
Crinitella coheri Allen & Edmunds

Mecoptera
Bittacus coheri Bicha

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PHILAMYIANY

Diptera on stamps (6): Tabanoidea

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The sixth contribution to "Diptera on stamps" deals with the superfamily Tabanoidea. Because none of the families of Tabanoidea include any species of outstanding medical importance, there are hardly any stamps known depicting these flies. The first example is from Belgium (1971) and highlights the obvious colouration of eyes that are typical for several Tabanidae. This is the very first stamp showing a Diptera due to morphological features. Another very special stamp showing a Tabanidae is the Austrian (1999) stamp that illustrates a Diptera as part of the ecosystem Danube floodplains. This is the earliest stamp showing a Biotop with a fly. In the stamp from Japan (1999) it is hardly possible to identify the small insect as a fly. However, the stamp shows a reproduction of the famous woodblock print "Chyrsanthemus and Horsefly" (1833–1834) from Katsushika Hokusai (1760–1849) and looking at the original it is clearly visible that an unidentified Tabanidae is depicted. The last Tabanidae that is found on a stamp is from Mongolia (2004). All other objects illustrate exemples that look like stamps bur are none: Mozambique (2018) and São Tomé and Príncipe (2015) are issues that are very likely to never have been sold nor used in Mozambique nor São Tomé and Príncipe but were only printed for stamp collectors. While these stamps were published with the formal authorisation of the national post offices the print from Central African Republic (2012) with a Rhagionidae is an illegal stamp without any potential to be ever used as a regular stamp. The Eynhallow (1982) is a cinderella, meaning a fantasy stamp. The Scottish Island Eynhallow is only 75 hectars in size and not populated. The stamp from "Cartonia" is another cinderella that doesn’t even have any regional reference. Although it is not a stamp, the unidentified Tabanidae on the Poland (1983) issue at least has an exciting history as a propaganda stamp issued by the Polish trade Union Solidarność. The New York Times called these stamps "Cinderellas for Solidarity".

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/).

Acknowledgement
Theo Zeegers (Leiden) helped with the identification of the illustrated Tabanidae and Rhagionidae.

Tabanidae indet – Belgium 1971: *Tabanus bromius* ♀ [Zoo – Antwerpen – Anvers], 3.50 + 1.50 Belgian franc. – Michel number: BE 1664; stamp number: BE B880.

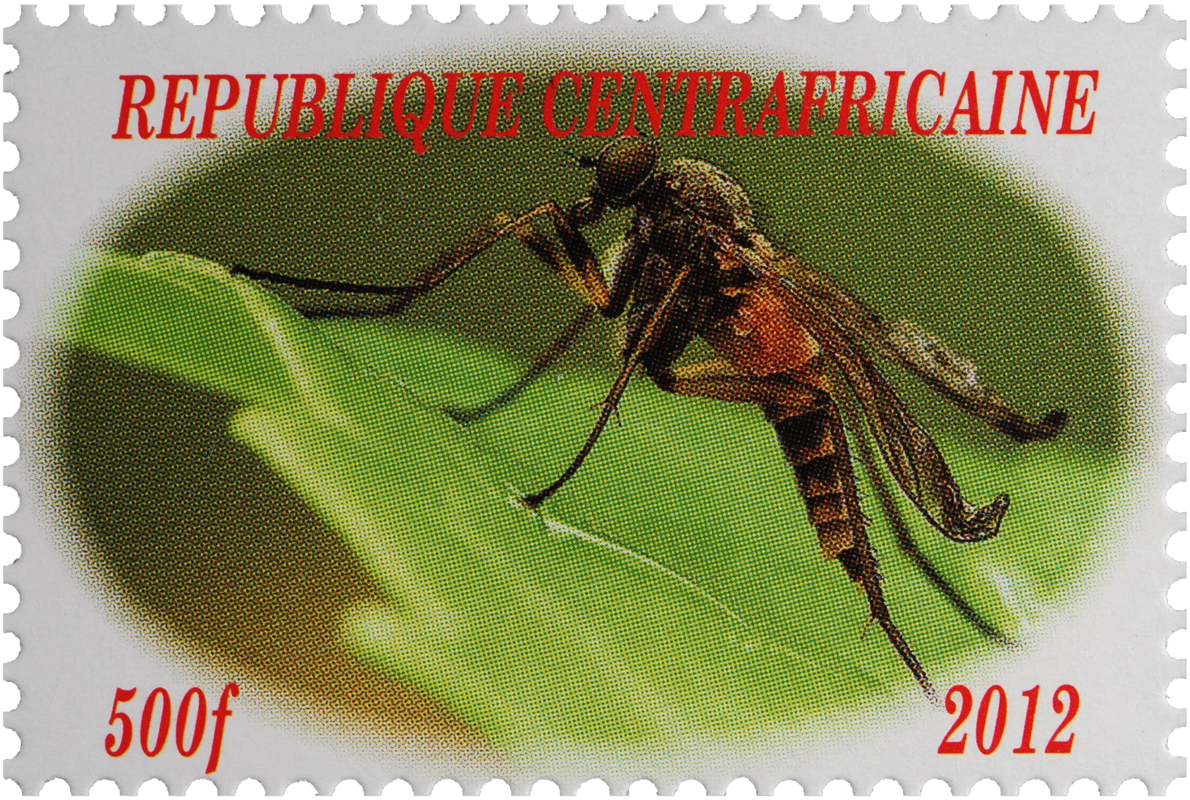
***Tabanus* spec. or *Hybomitra* spec. – Austria 1999:** Nationalpark Donau-Auen, 7 Austrian schilling. – Michel number: AT 2288; stamp number: AT 1792.

Tabanidae indet – Japan 1999: Chrysanthemums and Horsefly, 110 Japanese yen. – Michel number: JP 2786; stamp number: JP 2712.

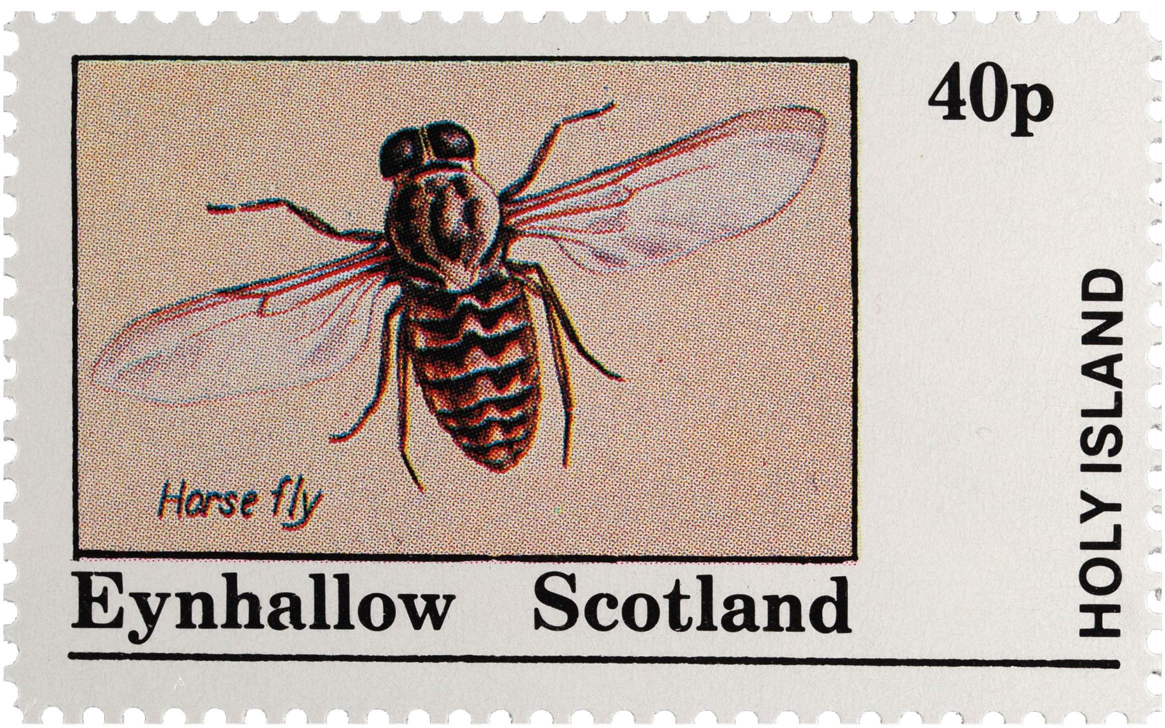
***Tabanus* spec. or *Hybomitra* spec. – Mongolia 2004:** *Tabanus bovinus* [Mongolian insects and flowers], 200 Mongolian tögrög. – Michel number: MN 3544; stamp number: MN 2589d.

Chrysopilus thoracicus **(FABRICIUS, 1805) – São Tomé and Príncipe 2015:** *Chrysopilus thoracicus, Heliamorpha ionasi* [Plantas carnivoras e suas vitimas em floresta tropical], 19000 São Tomé and Príncipe dobra. – Michel number: ST 6119; stamp number: –; issue was not placed on sale in São Tomé and Príncipe.

***Haematopota* spec. – Mozambique 2018:** *Haematopota pluvialis* [Micro-Monstros], 116 Mozambican metical. – Michel number: MZ 9191; stamp number: –;issue was not placed on sale in Mozambique.



***Rhagio* spec. – Central African Republic 2012:** [les insectes du monde]. private printed, illegal stamp.



***Tabanus sudeticus* ZELLER, 1842 – Britain [Eynhallow] 1982:** Horse fly, 40 British penny. – Cinderella.

***Tabanus* spec. – Chartonia without date:** Brown Horsefly (*Tabanus bromius*). Cinderella.

Tabanidae indet – Poland [Poczta "Solidarność" Warszawa] 1983: Ślepak (tabanida masc.), 10 Polish złoty. – propaganda stamp.

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**Diptera Trading Cards and Trade Cards (IV),
Cigarette Cards, second half of the 20th century**

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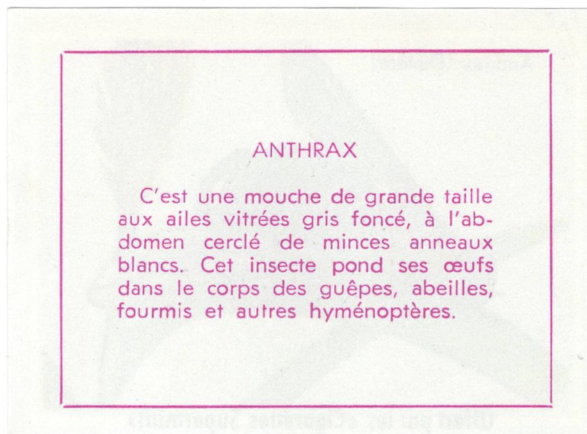
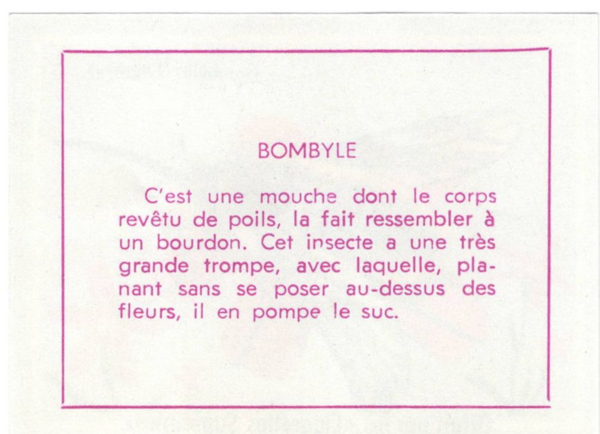
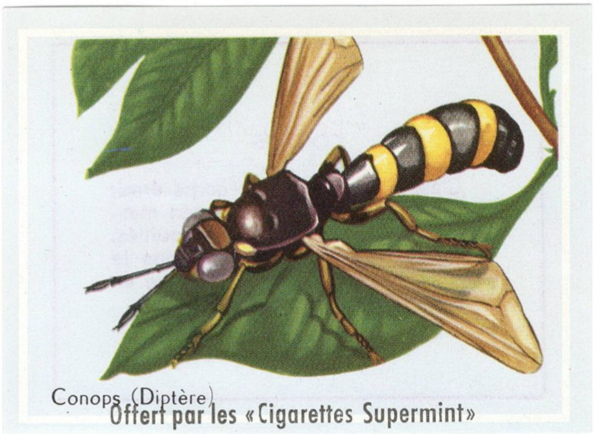
In the last issue of *Fly Times*, I focused on cigarette cards from the early 20th century, before World War II. Although that kind of cigarette card (the type used to stiffen the cigarette packs) continued for a while after, it later morphed into being strictly a collectible. That is, there was no longer a practical reason for including them (i.e., to stiffen the cigarette pack) except for interest and advertising. One company that produced such cards was Cigarettes Supermint, a French manufacturer of menthol cigarettes. The following cards are from their insect series in the 1960s, and are notably much more flimsy than the previous cards, being printed on very thin glossy paper. You can see in some of the cards that the cuts are sometimes rather sloppy, but they are interesting cards. They are also considerably larger, with the following cards depicted at actual size.

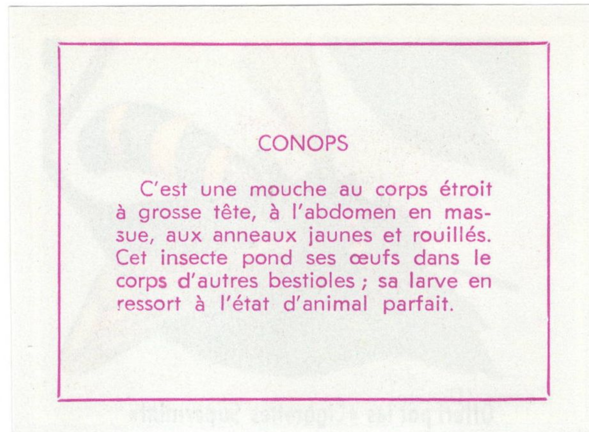


This card depicts a species of the bombyliid genus *Anthrax*, with the text saying it is large, with dark gray, glassy wings, and an abdomen with thin white rings. The biology is given as laying its eggs in the bodies of wasps, bees, ants and other hymenopterans.

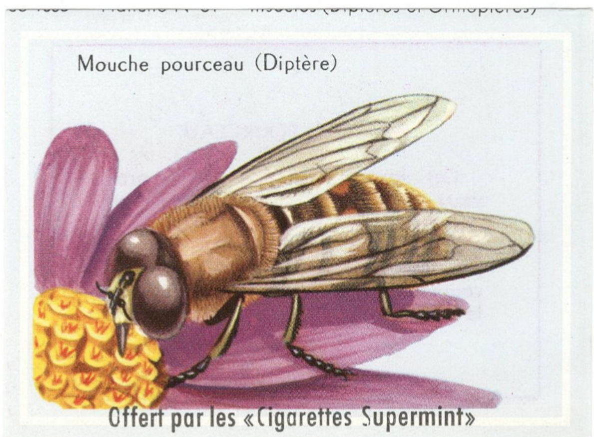


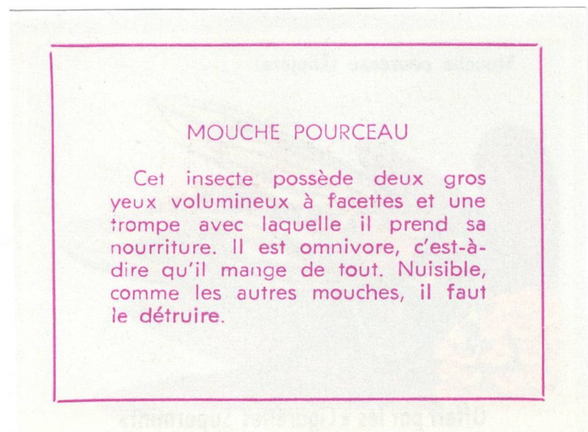
Another bombyliid, this card depicts *Bombylius major*, which they describe as being hairy and looking like a bumblebee, and that they have a very large proboscis with which they hover above flowers without landing to such our their juice.



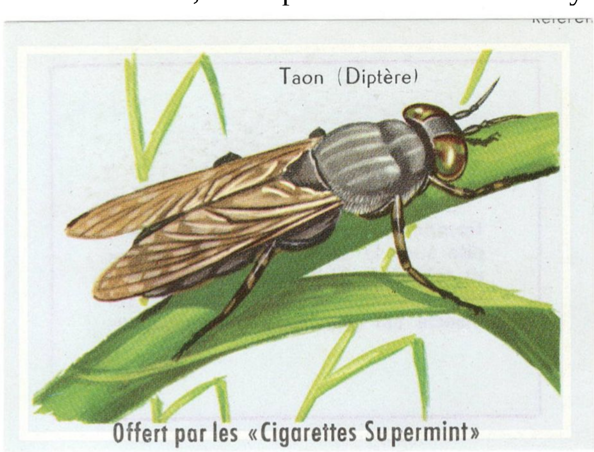


This card depicts a species of *Conops*, which they describe as being narrow bodied with a large head and a club-shaped abdomen with yellow and dark rings, and that they lay eggs in the bodies of other insects with larvae emerging as a "perfect animal".

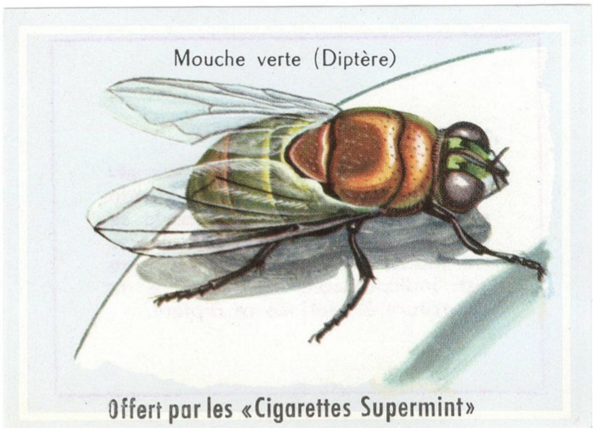




This card depicts *Eristalis tenax*, describing it as having two large, faceted yes and a proboscis for taking food. They further describe it as omnivorous, specifying that it east everything and stating the "Like other flies, it is a pest and must be destroyed".



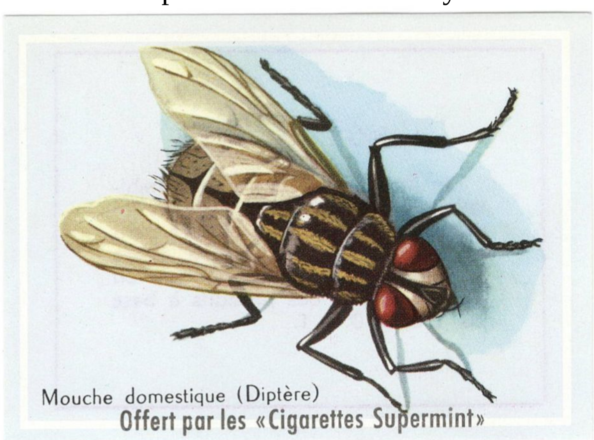
This card depicts *Haematopota pulvialis*, which they describe as being stocky, with a short proboscis to such the blood of large animals, and even humans, with a painful bit. They say it is very common, and gray to reddish in color.

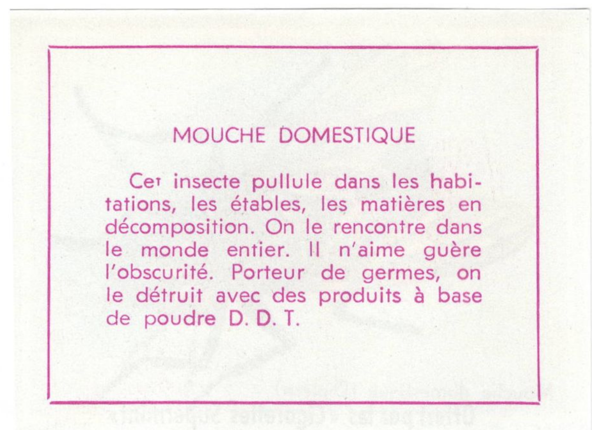



MOUCHE VERTE

D'aspect agréable, cet insecte affectionne particulièrement la viande dans laquelle il pond ses œufs se transformant rapidement en larves ou « asticots ». L'armoire frigorifique est indispensable pour mettre la nourriture à l'abri de ce diptère.

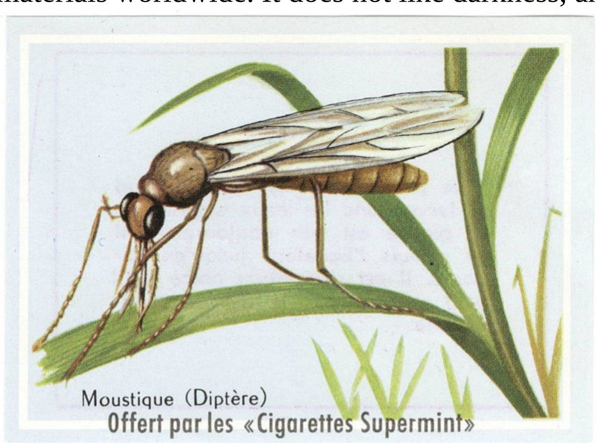
This card depicts a species of *Lucilia*, which they say is pleasant-looking, and is particularly fond of meat where it lays its eggs that transform into "maggots". They also stress that a refrigerator is essential to keep food safe from this fly.

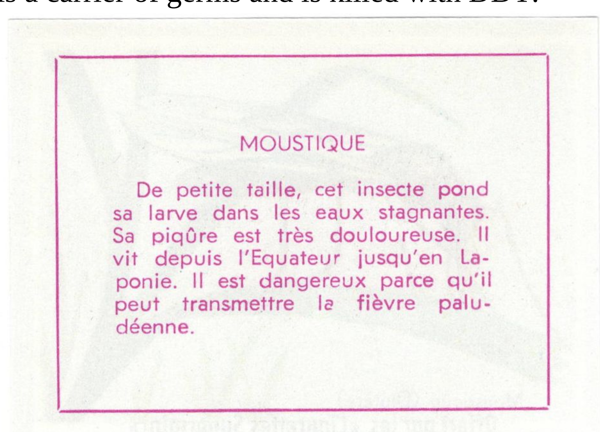



MOUCHE DOMESTIQUE

Cet insecte pullule dans les habitations, les étables, les matières en décomposition. On le rencontre dans le monde entier. Il n'aime guère l'obscurité. Porteur de germes, on le détruit avec des produits à base de poudre D. D. T.

This card depicts *Musca domestica*, stating that it swarms in stable housing and decomposing materials worldwide. It does not like darkness, and is a carrier of germs and is killed with DDT.




MOUSTIQUE

De petite taille, cet insecte pond sa larve dans les eaux stagnantes. Sa piqûre est très douloureuse. Il vit depuis l'Equateur jusqu'en Laponie. Il est dangereux parce qu'il peut transmettre la fièvre paludéenne.

This card depicts a species of *Anopheles* mosquito, which they describe as being small and laying eggs in stagnant water, and having a painful bite. They occur from the equator to Lapland (the far north of Finland) and are dangerous because they transmit malarial fever.

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Fly Poetry

Neal L. Evenhuis

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These poems by me are originally from the late Terry Wheeler's blog: "Three lines about six legs", which can still be found here: (https://3linesabout6legs.wordpress.com). There are a number of other fly-related poem there; as well as other insect-related poems by Terry and many of his friends. Go visit and remember Terry. You could be rewarded with a smile, a chuckle; or you might find yourself subconsciously responding with a slight nod and/or small sound of concurrence.

Flies that Cannot Swim
to the fly laying at the bottom of my pan trap
why were you attracted to yellow
when there is no yellow in the rainforest

Apology to A Fly
i originally named you *Phthiria relativitae*, but now you are a *Poecilognathus*
it was short-lived, and you were happy then;
i am very sorry, but you are no longer a funny fly

Genetic Plaything
today there is a leg sticking out of my head
tomorrow my wings are all wrinkled
i am: *Drosophila melanogaster*

Phorid Identity Crisis
if i were a coffin fly
i'd rather be called a scuttle fly
it sounds more like i have boundless energy

He Can't Wear Long Pants
Campsicnemus magius
has incredibly complex worm-like processes on his legs
yet he somehow does not trip over himself

The *Entomophthora* blues
fly with mouth wide open
mindless flight
stuck to the window, all fuzzy

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MEETING NEWS

ICDX – Reno: a personal reflection from a Musca-teer

Neal L. Evenhuis

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ICDX [International Congress of Dipterology-X], or the 10th iteration of getting fly folks from all over the world to give their precious money to airline companies just in order to be able get together for a really good time for less than a week, was held in Reno, Nevada from 16–21 July 2023. The venue was the Silver Legacy Hotel and Casino in beautiful downtown Reno. [Given that I was going to be immersed in every type of legal gambling known to humans, when I arrived, I thought that maybe I'd see dung fly workers at the crap tables, but it didn't happen.] After 9 previous Diptera congresses, this was the first time an ICD was held in the U.S. (although some thought the 1994 one in Guelph, Canada was—but you can easily tell the difference between Canada and the U.S. by the fact that in Canada, curling is a sport, but in the U.S. it used to be a hair style trend).

From a logistics standpoint, the venue was well-chosen as it offered most everything in one place (food, beverages, bars, entertainment, lodging, bars, snacks, meetings, bars, did I mention bars?), so heading outside was an option and not a necessity. And the record heat during that week across the western U.S. including around the Reno area (over 110 °F [43.3 °C] some days) pretty much solidified the notion that staying inside with nice cool air-conditioning wafting through was a good idea. Still, there were a few brave souls who ventured out to see if the rumors were true that there really were trees growing in Reno; and that you really could fry an egg on the sidewalk in that heat. And there were actually some good eateries and drinkeries nearby within a short walk, while meeting the local citizens relaxing on the sidewalk, some of whom seemed to talk to people we could not see. Back inside, I think most rooms in the hotel had pretty amazing views. It was particularly nice to wake up to a view of brilliant blue sky and the Sierra Nevada in the distance with patches of snow, even in the middle of a hot summer. However, it baffles me that there could be 110 °F heat yet still have patches of snow on hills and mountains not that far away (although over the week those patches did shrink a bit).

The location of the meeting rooms was a fer bit of a walk from one's hotel room (my Fitbit definitely spiked the week I was in Reno) and it turned out to be a good idea to find exactly where they were before the meetings started, as it was not entirely clear at first (the hotel was not great about maps of meeting rooms as their first concern was to be able to separate you from your money at the gambling tables, which were for some reason much easier to find). But, just like walking an old favorite trail in the woods to collect flies, it became natural and easy to get to the meeting venue once you recognized landmarks; as you took the same path down elevators, stairs and escalators, past boutiques, ignoring trapeze artists in glittering costumes passing out ticket information for their upcoming act someplace, and past bars, restaurants, gift shops, coffee shops and various slot machines, roulette wheels and card tables—to eventually gather in the lekking area [= the Royal Salon meeting lobby] to meet old friends and colleagues while sipping hot coffee and partaking of donuts and Danishes. (I did not weigh myself after that week as I was truly terrified of the result.)

After seeing a few colleagues the day prior and having a few beers (OK, maybe not a few) and doing a lot of catch-up (after all, it had been five years and a pandemic since the last Congress in Namibia), it was time to get down to the important business the next day. The Welcome Mixer! ... But first, we had to register and get our swag bag. And, thanks to the help of the pleasant and happy-go-lucky volunteers from the University of Reno, registration was easy and painless. So, with logo-emblazoned lanyards, logo-emblazoned tote bag, logo-emblazoned thumb-drive, logo-emblazoned notepad, and logo-emblazoned program in hand, we were all set for the week to come.

There are many good things to say about ICDX, and one of them was that the beer and wine was free-flowing at all of the evening get-togethers during the week. The Welcome mixer on Sunday evening and the receptions and dinners in the evenings were all accompanied by ample amounts of conversation-lubricating beverages (and Congress organizer Steve Gaimari learned that being a wine club member has its benefits!). The dinners were buffets and "themed" – i.e., one night was "Taqueria" night with delicious fixings for burritos and tacos — and desserts were plentiful and rather sinful (and should be prescribed at every conference).

The opening of the 10th Congress included all the normal speeches, thank-yous, and notices, and featured an elite group of just four surviving delegates who had attended every Congress since the first in 1986 in Budapest. Given the unique opportunity, the group was aptly nicknamed the 4 Musca-Teers and wore identical white T-shirts printed with that moniker plus two fly graphics. Thomas Pape, myself, Dan Bickel and Adrian Pont each briefly reminisced at the podium about some of the past Congresses and there were a few images of younger-looking delegates, that surely no one recognized, accompanying the talks.

The meeting site offered spacious rooms with ample seating and good audio and visual; and each meeting room was directly opposite the Royal Salon meeting lobby, which offered planned, spontaneous and serendipitous meetings with colleagues at any time, plus coffee, tea, and snacks. There were also small break-out rooms off the lobby area, which allowed for ad hoc meetings of smaller groups. The lobby area was also where the registration desk offered late registration as well as purchase-offerings of Congress merch including a variety of Diptera-themed T-shirts.

Although there were excellent plenary talks and meeting presentations at the various symposia, the highlight of most of the recent congresses was the banquet speech, and ICDX was no exception. On Wednesday evening Erica McAlister (photo right) entertained the crowd with her "Charismatic Diptera" talk, replete with stunning photographs of Diptera in various compromising positions. Erica, well-known author of two popular books on

Diptera and highly sought-after for speeches about how amazing Diptera are — given mostly to non-dipterists, not surprisingly wowed the crowd of dipterists with awesome Diptera facts, little-known dipterological tidbits, combined with her infectious humor. And in an amazing feat, her glass of white wine, precariously perched on the podium throughout her talk, did not spill over, and instead was used for a highly proper toast at the end. Well played Erica!

Wednesday saw the poster presentation room overtaken by the Diptera photo contest, with some remarkable and whimsical entries. And, ironically, it was also the day where the traditional group photo was attempted (read “herding cats”). Unfortunately, the group photo did not make the cut for the photo contest, but it joins the Namibia Congress group photo in that elite group of attempted Congress group photos.

We four Musca-Teers have been to 10 Diptera congresses over the past 5 decades and have seen plenty of good and bad during that time (Heck! I make lists of them!). Each Congress has some superlatives that can be said about it; as well as some disparaging things that might be uttered in private company. But one thing is true for each one. It takes a dedicated team and hard work to pull it off. ICDX was surely no cake-walk, but it came off. Kudos! Congress organizers Steve Gaimari, Shaun Winterton, Martin Hauser, Chris Borkent, Giar-Ann Kung, Brian Brown, Alessandra Rung, and Peter Kerr are to be thanked for all the work they did to make ICDX the success that it was.

We now look forward to Croatia for the 11th International Congress of Dipterology. I hope to see you all there! And I will be ready to touch the big toe of Grgur Ninski for good luck.

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ICDX USA 2023 10TH INTERNATIONAL CONGRESS OF DIPTEROLOGY | RENO, USA 16–21 JULY, 2023

ICDX wrap-up

Stephen D. Gaimari

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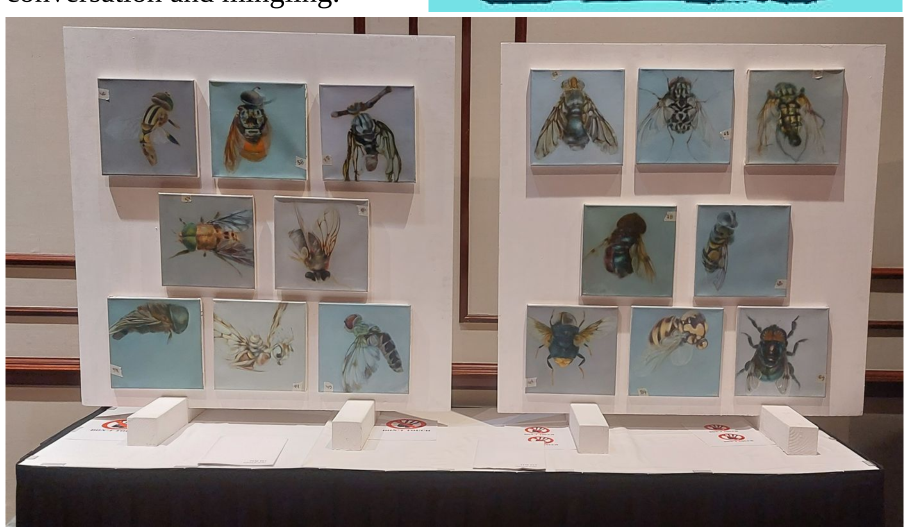
The 10th International Congress of Dipterology (ICDX) was held from 16–21 July 2023 at the Silver Legacy Resort in Reno, Nevada, USA. The full Abstract Volume (Fig. 1) was published as *Fly Times Supplement* 5, and is downloadable from https://dipterists.org/fly_times_supplement.html along with the Scientific Program. There is also a buy-it-now option if you want to purchase the same hard copy that delegates received for each of these. German artist Natalie Port provided the artwork for the front and back covers of both, and loaned us some examples from her series "Entomologische Sammlung der Labors Diptera" (Fig. 2).

The scientific program ran from the morning of Monday, 17 July, through noon Friday, 21 July. Sunday, 16 July, consisted of registration and hobnobbing among dipterists, with a very well attended welcome reception that evening. That reception set the pace for the rest of the Congress, with receptions each evening, with drinks freely flowing, plenty of hors d'oeuvres, and great conversation and mingling.



Figs. 1–2. 1 (top) Cover of the ICDX Abstract Volume. 2 (bottom). Paintings by German artist Natalie Port, loaned to us for display.

Monday morning saw the meeting kick off with an opening ceremony prior to the first plenary talk. After my general introduction and background, I turned the microphone over to our very special guests, the 4 Musca-teers, i.e., the four dipterists who have been to every ICD – Thomas Pape, Neal Evenhuis, Dan Bickel, and Adrian Pont, each wearing a t-shirt with their logo (see Fig. 3).

Each morning, a plenary presentation was given (except for one cancellation) to kick off the day's scientific program. Several of these are available on the Dipterists Society Youtube channel (https://youtube.com/@dipterists). In total, the scientific program consisted of 222 abstracts, with 190 oral presentations in 20 symposia, a general session, a book launch event, plenary talks, and a banquet address. The poster session consisted of 32 posters. On Monday and Tuesday of the meeting, we ran three concurrent symposia, while on Wednesday through Friday we ran two. Time slots were 15 minutes for each presentation including questions/discussion. Many symposia had keynote presentations, which could take two time slots. Lunch each day had a differently themed buffet, including Italian, New York Deli, Baja Peninsula (Mexican).

Figs. 3–4. 3 (top). Dan Bickel, one of the 4 Musca-teers, giving some reflections during the opening ceremony. 4 (bottom). Dipterists engrossed by the banquet address of Erica McAlister.

On Tuesday evening, the Congress hosted a book launch reception for Volume 3 of the Manual of Afrotropical Diptera. The event was kicked off with an excellent harp performance by Michał Majkowski, the son of one of our exhibitors. Following an introduction and background from Ashley Kirk-Spriggs, the official launch was given by Adrian Pont, followed by a talk on fly photography for the book by Steve Marshall. The Dipterists Society Youtube channel also has videos of this whole event, including the harp performance.

Wednesday was the Diptera photo competition and the start of the poster session. We also had the group photos, which you can see in the article prior to this one by Neal Evenhuis. In the evening was the Congress banquet, which was attended by almost 200 dipterists and their accompanying persons, and was enjoyed by all. The lead off for the banquet was a presentation by Erica McAlister entitled "Charismatic Diptera – who are we kidding?" (also available on our Youtube channel), as expected an excellent talk that kept everyone thoroughly entertained while their hunger increased. Fortunately, the drinks were already flowing, and the meal that followed was excellent, and accompanied by wines donated for the event by Berryessa Gap vineyard in California.

There were 201 total registered delegates (4 were no-shows) from 35 countries, along with 30 accompanying persons, and several exhibitors. Following are a few relevant demographics (percentages rounded off) about the makeup of our delegates. Of the registered delegates, 48 were students (24%), 68 were 35 years old or younger (35%), and 70 were women (35%). Regarding participation by women, they gave 31% of the oral presentations and 39% of the posters, and were 27% of the symposium organizers. Regarding participation by students, they gave 20% of the oral presentations and 30% of the posters, and were 12% of the symposium organizers.

Following is a list of the symposia (and their organizers) held during the Congress:

- Acalyptrates (Keith Bayless)
- Advances in Afrotropical dipterology (Ashley Kirk-Spriggs & Bradley Sinclair)
- Advances in Diptera palaeontology (Vladimir Blagoderov & Agnieszka Soszyńska)
- Advances in lower Brachycera systematics and taxonomy (Xuankun Li & Diego A. Fachin)
- Biodiversity surveys and conservation (Marc Pollet & Justin Runyon)
- Biology, ecology and development of management strategies for biting flies (Daniel Kline & Jerry Hogsette)
- Calyptrate systematics and diversity (Daniel Whitmore & Pierfilippo Cerrett)
- Culicomorpha (John Soghigian & Brian Wiedmann)
- Diptera morphology (Gregory Curler & André P. Amaral)
- Diptera phylogenomics (Jessica Gillung & Liping Yan)
- Diptera pollinators (Andrew Young)
- Dipterans as parasites and vectors (Tamara Szentiványi)
- Dipterology in forensic entomology (Robert Kimsey)
- Empidoidea (Marija Ivković)
- General session (Brian Brown)
- Multilevel solutions to large taxonomic problems in Diptera (Leshon Lee & Valerio Caruso)
- Syrphoidea (Ximo Mengual)
- Systematics and ecology of Bibionomorpha (Chris Borkent & Netta Dorchin)
- Taxonomy and phylogeny of Asilidae – honoring Eric Fisher and his impact on understanding the Nearctic and Neotropical fauna (Torsten Dikon)
- Tephritoidea of economic importance (Severyn Korneyev)
- Tipuloidea (Solange Akimana & Jon Gelhaus)

The Dipterists Society was very happy to provide student grants to aid their attending ICDX. In total we received 41 proposals from students in 19 countries, and we awarded grants to nine students from six countries for a total of $12,000. In addition, we awarded undergraduate grants totaling $1800 for two local students from the University of Nevada, Reno.

During the closing ceremony on the last day, the various awards were given out for the student presentation competition, the student poster competition, and the Diptera photography competition. First, I want to thank all the judges who spent their time going to the various student talks and posters, and assessing the photos, and making their difficult decisions. Despite their needing to decide from among an excellent pool of choices, the judges did come up with 1st, 2nd and 3rd place winners for each of the two student competitions. The same holds true for the photography competition, but with only 1st and 2nd places. Following are the winners:

Student Presentation competition:

1st place: **Kinga Walczak**, from Nicolaus Copernicus University in Toruń, Poland, for the talk titled “To see the unseen: on confocal microscopy in Diptera morphology studies”.

2nd place: **Ezra Bailey**, from North Carolina State University, in Raleigh, North Carolina, USA, for the talk titled “Using anchored hybrid enrichment to resolve the higher-level phylogeny of anthomyiid flies (Muscoidea: Anthomyiidae)”.

3rd place: **Tais Madeira Ott**, from University of Campinas in Brazil, for the talk titled “An integrated overview of *Paralucilia* species (Diptera, Oestroidea, Calliphoridae) in the Amazonian biome”.

Student Poster competition:

1st place: **Maxwell Arnold**, from University of California in Davis, California, USA, for the poster titled “Succession and richness of acalyptrate muscoid families with respect to vertebrate decomposition”.

2nd place: **Gabriela Antonieta Oyarce**, from Universidad de Concepción in Chillán, Chile, for the poster titled “Insecticidal activity of *Dysphania ambrosioides* (Amaranthaceae) essential oil against house fly (*Musca domestica*) (Muscidae)”.

3rd place: **Alice Dabrowski**, from University of Guelph in Guelph, Ontario, Canada, for the poster titled “The state of North American aphidophagous syrphid larvae (Syrphidae) descriptions and knowledge gaps”.

Photography competition:

1st place: **Zachary Dankowicz**, from Walter Johnson High School in Bethesda, Maryland, USA, for his photo of a hanging tipulid, *Nephrotoma eucera* (Loew) or near (Fig. 5).

2nd place: (tie for 2nd place between)
Santiago Jaume-Schinkel, from the Museum Koenig in Bonn, Germany, for his photo of the posterior end of an interesting tipulid larva (Fig. 6, right).
Chien-Yu Hunag, from the National Taiwan University in Taipei, for her photo of a mating pair of the tephritid species *Rhabdochaeta formosana* Shiraki (Fig. 6, left).

Fig. 5.. 1st prize in the ICDX photography competition, won by Zachary Dankowicz for this hanging tipulid, *Nephrotoma eucera* (or near).

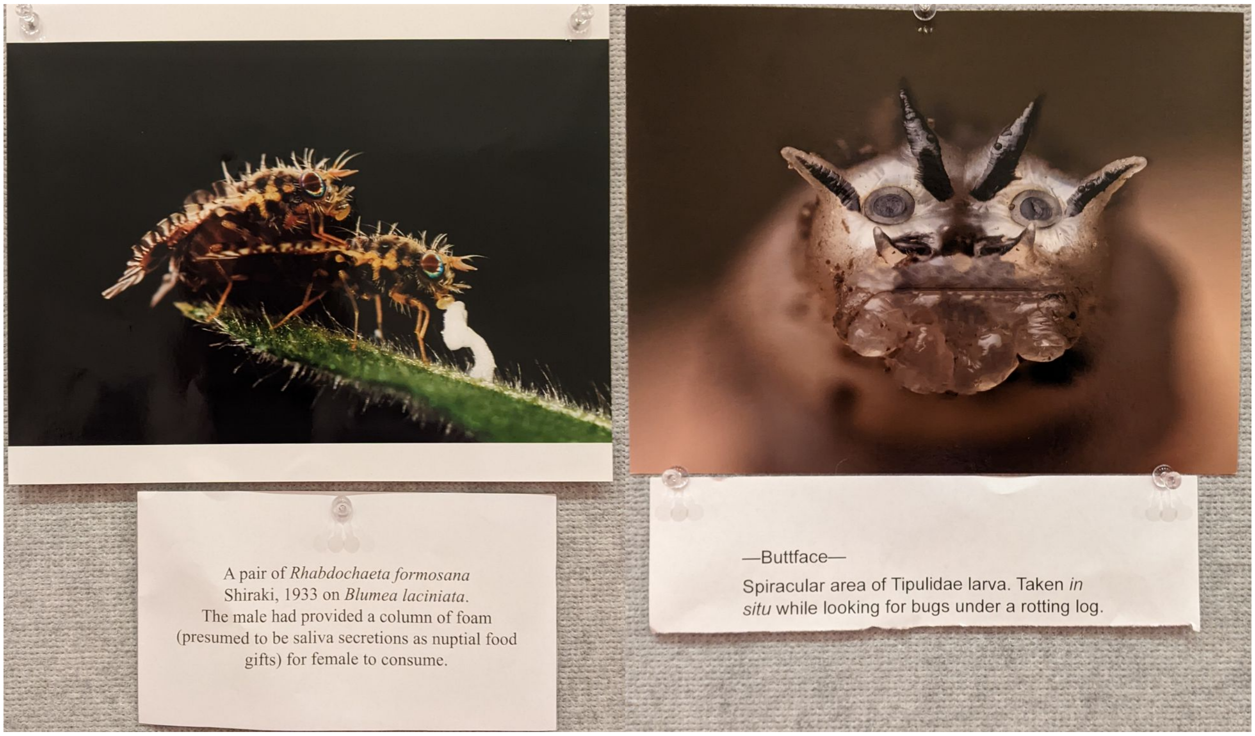


Fig. 6. The two photographs tied for 2nd prize in the ICDX photography competition. (left) Chien-Yu Hunag's photo of a mating pair of the tephritid species *Rhabdochaeta formosana* Shiraki. (right) Santiago Jaume-Schinkel's photo of the posterior end of an interesting tipulid larva,

The next part of the closing ceremony entailed comments from the outgoing chair, Rudolf Meier, announcing the changes to the council membership and making known that the next Congress will be held in Zagreb, Croatia. This was followed by a presentation by the ICD XI Chair, Marija Ivković, announcing that it will be held from 10–16 July 2027, and telling everyone about Zagreb and Croatia. This looks like it will be a fantastic Congress, and I am really looking forward to seeing everyone there! Then Makoto Tokuda told us about the upcoming International Congress of Entomology being held from 25–30 August 2024 in Kyoto, Japan, and showed a short video. Another meeting that will be great for dipterists! Lots of great meetings to look forward to.

As a parting thought, I wish to thank everyone who helped make this such a successful Congress! The folks at the venue were fantastic, Andrew Silva and the team at the Silver Legacy – they took care of all of the meeting rooms, audio-visuals, the food, the bar at the evening receptions, and were available at a moments notice to fix any issues, and remained entirely flexible through the whole meeting! Also thanks go out to the ICD Council, the organizing committee, all of the many symposium organizers, the plenary and banquet speakers, all of the presenters of both posters and talks, those who entered the photography contest, all of the accompanying persons, and everyone who participated in any way. In terms of organizing, Martin Hauser went above and beyond as the Symposium Chair, making sure the symposium organizers were on track, taking care of abstract submissions, etc., in addition to being a great help with virtually any task asked of him, both before and during the Congress. As for those who helped run the show on site, I thank Giar-Ann Kung and Alessandra Rung who really did everything asked of them, and were always available! And our student helpers from the University of Nevada, Reno – Hannah Prins and Khong Lunaria – were with us all week and ran the main ICDX table the whole time. We could not have asked for better, more reliable or more conscientious helpers! On day one, when we had the rush of registration, we also appreciate the help of Brittany Kohler and Maples.

And last, but by no means least, our deepest gratitude goes out to all of our sponsors and donors, without whom we could never have put on such a successful meeting. In organizing the Congress, I approached more than 200 organizations, companies, and institutions seeking sponsorship. I am very happy to recognize here the generosity of those who sponsored us. Besides these kinds of sponsorships, we also received many personal donations, led by the very generous donations of Mike & Bonnie Irwin[1] and Terry & Faye Whitworth[1], and also including Ashley Kirk-Spriggs, Bill Murphy, Casey Rush, David Grimaldi, Fenja Brodo, Fiona Hunter, Lance Jones, Melissa Espinoza, and Steve & Helen Gaimari.

Following is a list of our institutional sponsors (you may have seen their half-page ads in the back pages of the Scientific Program, and their logos on the back of the Abstract Volume and on our website), in alphabetical order:

- Africal Natural History Research Trust[1] (United Kingdom)
- Amber Inclusions (Lithuania)
- Berryessa Gap Winery (USA)
- Center for Biological Diversity (USA)
- Don't Pack a Pest[2] (USA)
- Entomological Society of Canada
- Entoquip (USA)
- International Union of Biological Sciences[2] (France)
- Linnean Society of London (United Kingdom)
- Magnolia Press (New Zealand)
- Majkowski Woodworking Company[2] (Poland)
- Natalie Port (Germany)
- NHBS Ltd (United Kingdom)
- North American Dipterists Society[1] (USA)
- Pensoft Publishers (Bulgaria)
- Reno-Sparks Convention & Visitors Authority[2] (USA)
- Royal Entomological Society (United Kingdom)
- Species File Group (USA)

And finally, very best wishes to Marija and the rest of the organizers of ICD XI – looking forward to seeing everyone in Zagreb!

We greatly appreciate donations and sponsorships at ALL levels, but want to give special recognition to:

[1]our donors of $10,000 or more, and

[2]our donors of $5,000 or more.

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North American Dipterists Society 18th biennial field meeting:
July 15–19, 2024 at the Evergreen State College, Olympia, Washington USA

Barbara Hayford[1] & Andrew Fasbender[2]

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The 18th Biennial Field Meeting of the North American Dipterists Society is scheduled for July 15–19 at the Evergreen State College (TESC; Fig. 1), located in the city of Olympia in Washington state. Located at the south end of Puget Sound, TESC is located about an hour's drive southwest of Seattle-Tacoma International Airport, a regional hub for the Pacific Northwest serviced by most major airlines. Registration for the Field Meeting is anticipated to be around $510, which includes lodging along with dinner and breakfast from the night of the 15th through the morning of the 19th (parking is an extra $6 per day for each vehicle). Accommodation consists of apartment suites made up of lockable private bedrooms sharing a common area and bathroom. Participants will also have access to a lab space with stereomicroscopes for sorting and identifying specimens.

The tentative schedule for the meeting starts on Monday the 15th with participant check-in through the afternoon and an opening presentation following dinner. The 16–18th will consist of field excursions to collecting sites in the southern portion of the Olympic Peninsula during the day, while evenings will host a series of 10–15 minute talks from participants on their research. All attendees are encouraged to submit a presentation, whether they are a first year student or old hand with decades in the field. Friday the 19th will consist of a "goodbye" breakfast and checkout, and whatever further activities you want.

Fig. 1. Purce Hall, The Evergreen State College. September 2023, B.L. Hayford.

Collecting opportunities start on TESC's campus, situated on 400 hectares of forest crossed with numerous hiking trails (Fig. 2). The north side of the property offers coastal frontage on Eld Inlet of Puget Sound, and there are multiple streams which flow through the campus. Multiple habitat types are easily accessible for setting up Malaise traps and black-lighting. The nearest off-site collecting location is Capitol State Forest, located a 15-minute drive to the southwest of TESC. An actively managed timberland, Capitol State Forest offers habitats in a variety of stages of succession from fresh clear cuts to 50–60 year old secondary growth dominated by Douglas Fir, Western Hemlock and Western Red Cedar (Washington DNR 2005). Clear cut by two logging companies in the mid-20th century, this 37,000+ hectare parcel was turned over to the state of Washington in the late 1950s to avoid paying property tax (Washington DNR 2017). The area is mountainous (with peaks of 800 meters), consisting of a series of ridges and draws crisscrossed with gravel roads and over 150 miles of trails. As with all the following collecting areas, there are numerous streams flowing through the forest.

Olympic National Forest (Fig. 3), located about forty miles northwest of TESC (about an hour by car), offers higher mountains and more mature forest than Capitol State Forest. The southeast portion of the Olympic Mountains is largely drained by the Skokomish River, a 20–30m wide stream which flows into Puget Sound. There are also several large lakes/reservoirs in the area, including Lake Cushman. To the north of Lake Cushman there is access to subalpine and alpine areas at the Mount Ellinor and Mount Washington trailheads. There are some opportunities to access old growth forest in this area, particularly in the upper part of the South Fork of Skokomish river drainage.

Figs. 2–3. 2 (left) Hiking Trail, The Evergreen State College campus. 3. (right). Unnamed Stream, Olympic Mountains. September 2022, B.L. Hayford.

A 90-minute drive from TESC is the Grays Harbor area and Pacific Ocean, offering many intertidal and estuarine habitats (Fig. 4) along with coastal bogs (Fig. 5) and lowland temperate rainforest. Our proposed collecting program should allow participants opportunities to sample many of the aforementioned habitats along with others such as coastal prairies. The Olympic Peninsula and adjacent Pacific Coast of Washington are noted for endemism among many taxa (including Diptera), giving participants an opportunity to locate these rare and sometimes undescribed species. Registration will open in early 2024 at: https://dipterists.org/field_meetings.html, and will also be announced on the Dipterists mailing list. Overall, we think the Olympic Peninsula of Washington will provide fruitful collecting and a great opportunity to meet with colleagues both old and new. We hope to see you there!

Figs. 4–5. 4 (left). Pacific Ocean shore, Grays Harbor County. 5 (right). Coastal Bog, Grays Harbor County. November 2023, B.L. Hayford.

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North American Dipterists Society organized meeting wrap-up (National Harbor, Maryland, USA)

Jessica P. Gillung

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The Organized Meeting of the North American Dipterists Society took place on Tuesday, November 7 from 19:00–21:00 EST during the Annual Meeting of the Entomological Society of America held in National Harbor, Maryland, USA. We did not receive any abstract submissions, so the meeting was reclassified as a "Function" by the ESA organizers. As a result, we did not have access to a projector or computer. A total of 21 people attended the Organized Meeting, most of them dipterists, as expected. We also successfully engaged coleopterists, neuropterists, and lepidopterists with an appreciation for the best insect order: Diptera. This wide range of interests and backgrounds ensured a dynamic and engaging conversation among attendees: we got to know each other, discuss our favorite flies, and explore the most interesting places we have been to in order to collect flies.

Moving forward, the Dipterists Society's Board of Directors is exploring options to provide snacks and drinks to attendees after the presentations, as a means to boost participation and engagement, as this meeting has always been part presentations and part social. The Board will also seek ways to convert the Organized Meeting to a hybrid format, thus offering an opportunity for engaging dipterists who were not able to attend the ESA meeting in person. This way, remote participants would have the opportunity to attend all talks, participate in the discussions, and deliver presentations remotely. Stay tuned for future developments!

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The past five international forums for surveillance and control of mosquitoes and vector-borne diseases (2015, 2017, 2019, 2021, and 2023)

Rui-De Xue

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Dr. Rui-De Xue from the USA and Dr. Tong-Yan Zhao from China discussed to create and organize the international forum for surveillance and control of mosquitoes and vector-borne diseases in China in 2008 and the proposal received the support by the Entomological Society of China (ESC) and the State Key Laboratory of Pathogen and Biosecurity (SKLPB), Beijing, China as a part of the international professional exchange activity. The meeting has been in conjunction with the National Congress of Medical and Veterinary Entomology (NCMVE) of the ESC and held in China every other year. The purpose of the meeting provides an opportunity to discuss current status and future challenges of mosquito and vector-borne disease surveillance and control programs in the world. Other objectives included: identifying possible areas of collaboration for research and development, sharing information about possible training and fund resources, and promoting new technology for surveillance and control of mosquitoes and vector-borne diseases. In this meeting, oral presentations are required for English only. Two projectors (one showing presentations in English and another showing presentations translated into Chinese) are used to facilitate communication. The programs were printed in both English and Chinese and published on the websites the Asian Society of Vector Ecology and Mosquito Control (ASVEMC)'s website at www.asiansvemc.org and www.mosquitoforum.net . The presentations and abstracts were brought together on a single DVD that was distributed to all participants in the 4th, 5th, and 6th.

In 2009, Dr. Rui-De Xue on behalf of the Society of Vector Ecology contacted and collaborated with Dr. Tong-Yan Zhao and Dr. Qi-Yong Liu from China about the creation of the Society of Vector Ecology (SOVE)'s Asian Branch as the Asian Society of Vector Ecology and Mosquito Control (ASVEMC)(www.ansiasvemc.org). In late May 2013, the ASVEMC's officers were installed by Dr. William Walton, President-Elect of the SOVE and the 1st member meeting was held before the 3rd IFSCMVD, Suzhou, China. The 1st Board of the ASVEMC was formed & Dr. Tong-Yan Zhao was elected as the 1st President. Then the Board decided to use the opportunity of the IFSCMVD to have the ASVEMC's Board meeting, member meeting, and officer rotation before the IFSCMVD every other year in the future (Fig. 10).

The 1st, 2nd, and 3rd IFSCMVD meeting summaries have been published in Wing Beats in 2010, 2012, and 2014. The 4th–8th IFSCMVDs in conjunction with the 10th–14th ESC's Congress of the Medical and Veterinary Entomology, and the 2nd–6th ASVEMC member meetings have been held in different cities with different dates in China (Table 1).

The 4th IFSCMVD, 10th NCMVE, 2nd ASVEMC meeting were held in Guangzhou, China, May 25–28, 2015 where was a fitting venue for the meeting as a dengue fever outbreak occurred in 2014. The meeting theme was **promotion of biorational and environmental methods for control of mosquito-borne diseases**. Dr. William Walton (Fig. 5) gave the keynote speaking about biorational and environmental control methods. The meeting provided an opportunity to review and discuss environmental and biorational control methods for mosquito-borne diseases, future directions and challenges, and celebration of Professor Bao-Lin Lu's achievement and accomplishment in the field of environmental and biocontrol of vector mosquitoes in China. More than 50 leading scientists in the fields of mosquito surveillance and control of mosquito & vector-borne diseases from 16 countries were invited to give presentations. A total of 30 presentations were given in the 3 panel sessions, and more than 50 presentations given in the 8 sections. The meeting attracted about 246 attendees from 15 countries, 29 Chinese provinces, and 15 private companies achieving both national and international attention. All participants in the 1st day were photographed together in front of the hotel (Fig. 1). All international scientists and participants were invited to visit Guangzhou Provincial CDC after the meeting.

The 5th IFSCMVD, 11th NCMVE, and 3rd ASVEMC meeting were held in Nanjing, China, May 22–26, 2017.
The meeting theme was **promotion of new technology for control of mosquito-borne diseases** and Dr. Daniel Strickman (Fig. 6) from the Gates Foundation gave the keynote speaking about innovation of technology for surveillance and control of vector-borne diseases. A total of 75 presentations were given in 10 sessions. The meeting attracted more than 270 participates from 13 countries. Due to rain fall, no photograph was taken for all participates.

The 6th IFSCMVD, 12th CMVE, and 4th ASVEMC meeting were held in Xiamen, China, May 26–30, 2019 where the medical entomology was born 100 years ago.
The meeting theme was **memorizing the history and exploring the future for control of mosquitoes and vector-borne diseases** and Dr. Graham White (Fig. 7) gave the keynote speaking about Dr. Patrick Manson – progenitor of Medical Entomology: from his inspiration by China to current elimination of malaria and filariasis. A total of 83 presentations were given in 12 sessions. The meeting attracted about 280 participates from 13 countries. All participates (Fig. 2) were photographed in the 1st day.

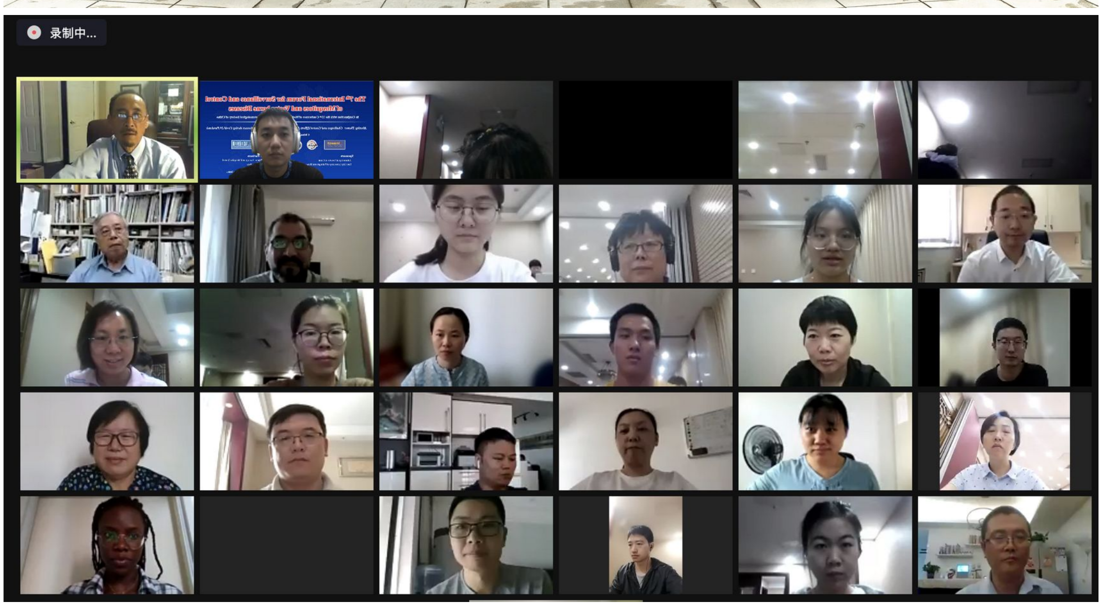
The 7th IFSCMVD, 13th CMVE, and 5th ASVEMC meeting were held by virtual, Beijing, China, August 15–18, 2021. The meeting was held in virtual zooming due to pandemic of COVID-19. The meeting theme was **Challenges and control efforts for mosquitoes and vector-borne diseases during COVID-19 pandemic.** The keynote speaker was Dr. Michael Turell (Fig. 8) who gave his presentation about How to recognize a vector and its importance. A total of 52 presentations were given in 6 sessions. The meeting attracted about 180 participates (Fig. 3) from 11 countries.

The 8th IFSCMVD, 14th CMVE, and 6th ASVEMC meeting were held in Beijing, China, October 23–27, 2023. This meeting was the 1st time in persons after the COVID-19 pandemic from year 2020 to early 2023. The meeting theme was **the world needs mosquito control: innovation and application of new technology for control of mosquito and vector-borne diseases**. Dr. J. Lyell Clarke (Fig. 9) gave the keynote speaking about the World Needs mosquito control: the industries play an important role in the pipeline. A total of 62 presentations were given in 13 sessions. The meeting attracted about 200 registrations and participates from 6 countries. The most participates were photographed in the 1st day of the meeting (Fig. 4).

The conference's council, program, and local arrangement committees and authors are very appreciating and thanking the ESC, SKLPB, ASVEMC, all colleagues, friends, and students, industry's sponsors, and all participates for their contributions to make each meeting successfully. The conferences could not be held and succussed without the hard working and timeless help of Dr. Tong-Yan Zhao, Dr. Chun-Xiao Li, Mrs. Ming-Yu Wu, Mr. Yan-De Dong, Dr. Hong-Liang Chu, and local CDCs from China. This article dedicates in memory of Dr. William Walton and Dr. Daniel Strickman who gave the keynote speaking at the 4th and 5th IFSCMVD and other help/support.

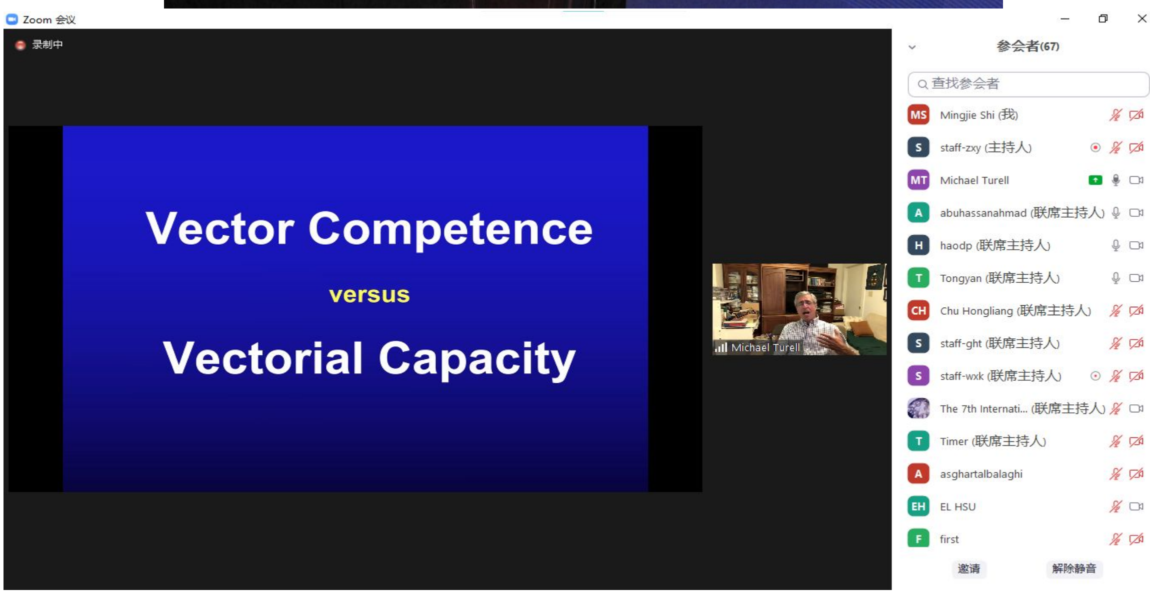
Table 1. The date, locations, meeting themes, the titles of the conference presidential address, keynote speakers with their presentation titles, and the leader scientists with their major topics at the 4th-8th International Forums for Surveillance and Control of Mosquitoes and Vector-borne Diseases organized by the Entomological Society of China, the State Key Laboratory of Pathogens and Biosecurity, and the Asian Society of Vector Ecology and Mosquito Control

Meeting, Location, and Date	**Meeting Theme**	**Title of the Conference Presidential Address**	**Keynote Speakers and their Organization**	**Titles of Presentations from Keynote speakers**	**Panel topics and Presenters**
4th IFSCMVD, Guangzhou, May 25–28, 2015	Promotion of Biorational AND Environmental Methods for Control of Mosquito-borne Diseases	Challenges and opportunities by Dr. Rui-De Xue	Dr. William Walton, Professor & Vice Chair, Department of Entomology, University of California, Riverside, CA, USA	Highlights of biorational and environmental methods for mosquito control	Dr. Wu-Chun Cao, Professor & Director, State Key Laboratory of Pathogen and Biosecurity (SKLPB), Beijing, **Status of vector-borne disease: emerging tick-borne diseases** Dr. Tong-Yan Zhao, Professor & Director, Department of Vector Biology and Control, **Professor Bao-Lin Lu's contribution and achievement for biological and environmental control of vector mosquitoes**
5th IFSCMVD, Nanjing, May 22–26, 2017	Promotion of New Technology for Control of Mosquito-borne Diseases	What is next for outbreak of potential mosquito-borne diseases by Dr. Rui-De Xue	Dr. Daniel Strickman, Senior Officer, Gates Foundation, Seattle, WA, USA.	Highlights of innovations of techniques and methods for control of vector mosquitoes	Dr. Wu-Chun Cao **for ticks**, Dr. Err-Lieh Hsu for **dengue in Taiwan**, Dr. G. Muller for **ATSB**, Dr. Tong-Yan Zhao for **New tech** in China. Dr. Randy Gaugler for **midges**, and Uli Bernier for **new repellents**
6th IFSCMVD, Xiamen, May 26–30, 2019	Memorizing the History and Exploring the Future for Control of Mosquito and Vector-borne Diseases	The past and future about the IFSCMVD by Dr. Rui-De Xue	Dr. Graham B. White, Emeritus Professor, University of Florida, USA.	Patrick Manson-Progenitor of Medical Entomology: from his inspiration by China to current elimination of malaria and filariasis	Dr. Le Kang for special lecture, Dr. Xiao-Nong Zhou for **tropical disease**, Dr. Tong-Yan Zhao for **Medical Entomology** in China, Dr. Qi-Yong Liu for **surveillance history** in China, Dr. Err-Lieh Hsu for **vector-borne disease** history in Taiwan, Dr. Uli Bernier for **National programs**
7th IFSCMVD, Virtual (Beijing, August 15–18, 2021	Challenge and Control Effort for Mosquito and Vector-borne Diseases during COVID-19 Pandemic	How has COVID-19 pandemic impacted on our mosquito surveillance and control program? Dr. Rui-De Xue	Dr. Michael Turell, Arbovirologist (Retired from US Army Institute, USA	How to recognize a vector and its importance	Dr. Wu-Chun Cao for **tick genomes and microbiome**, Dr. Xiao-Nong Zhao for **malaria elimination**, Dr. Tong-Yan Zhao for **IMM in rice field,** Dr. Qi-Yong Liu for ***Anopheles* and malaria control**, Mrs. Wendy Wei for **the BMGF China malaria**, Dr. Err-Lieh Hsu **for Surveillance**, and Dr. Xiao-Guang Chen for **the interspecific mating** between two *Aedes* mosquitoes
8th IFSCMVD, Beijing, May 23–27, 2023	The World Needs Mosquito Control: Development and Application of New Technology	Challenge and direction to use AI technique, robot, and drone for surveillance and control of mosquitoes and vectors by Dr. Rui-De Xue	Dr. John Lyell Clarke, CEO. The Clarke Mosquito Control, IL, USA	The World needs mosquito control: The industries play an important role in the pipeline	Dr. Wu-Chun Cao for tick-borne pathogens, Xiao-Nong Zhou for national malaria, Tong-Yan Zhao for *Aedes aegypti* in China, Xiao-Guang Chen for gene 7 dengue vector, Dr. Si-Bao Wang for symbiotic bacteria



Figs. 1–3. 1 (top). The 4th IFSCMVD, Guangzhou, May 25–28, 2015. 2 (middle). The 6th IFSCMVD, Xiamen, May 26–30, 2019. 3 (bottom). The 7th IFSCMVD, Virtual meeting, Beijing, August 15–18, 2021.

Figs. 4–6. 4 (top). The 8th IFSCMVD, Beijing, October 23–27, 2023. 5 (middle). Keynote speaker Dr. William Walton (deceased), Guangzhou, 2015. 6 (bottom). Keynote speaker Dr. Daniel Strickman (deceased), Nanjing, 2017.



Figs. 7–9. Keynote speakers. 7 (top). Graham White, Xiamen, 2019. 8 (middle). Mike Turell, Virtual, Beijing, 2021. 9 (bottom). Dr. John Lyell Clarke, Beijing 2023.

Fig. 10. Dr. Dan Kline (President-Elect of the Society of Vector Ecology, center) presented the certificate to Dr. Qi-Yong Liu (New President of the Asian Society of Vector Ecology & Mosquito Control, right), and moderator Dr. Rui-De Xue (left) prior to the 5th International Forum for Surveillance and control of Mosquitoes and Vector-borne Diseases, Guangzhou, China, May 25–28, 2015

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OPPORTUNITES

Postdoctoral Position Announcement,
Diptera Morphology and Evolution Lab.

Insect biodiversity in an Amazon tropical forest: species richness, vertical structure and faunistic turnover

Dalton de Souza Amorim

Departamento de Biologia, FFCLRP/USP, Av. Bandeirantes, 3900, Monte Alegre,
Ribeirão Preto, SP, Brazil; dsamorim@usp.br

[Photo, Craig Curler]

We are pleased to announce a Postdoctoral position in the Diptera Morphology and Evolution Lab, in the University of São Paulo at Ribeirão Preto, Brazil.

The project involves barcoding, taxonomy and ecological analyses of large-scale insect sampling in the Amazon Rainforest with by MiniON Oxford Nanopore NGS equipment. Sequencing protocols will be based on a reverse workflow process and use 648-bp COI barcode.

Applicants are required to have insect taxonomy morphology background and expertise in molecular NGS sequencing protocols. Applicants should have good communication skills in English. Familiarity with MiniON sequencer and basic skills in Portuguese are desirable.

Applicants must hold a PhD and send an application letter with their research interest and skills, along with an updated CV, to Prof. Dalton de Souza Amorim (dsamorim@usp.br).

The position is open to Brazilians and foreigners. The selected Postdoctoral fellow will receive a Postdoctoral scholarship from FAPESP (~US$ 1.8K/month paid in R$) and an additional 10% for research spending.

Project: Insect biodiversity in an Amazon tropical forest: species richness, vertical structure and faunistic turnover (FAPESP Proc. 2021/14092-0)
Working area: Zoology: Taxonomy
Number of positions: one
Begining: 1 February 2024
Principal investigator: Dalton de Souza Amorim
Institution: Departamento de Biologia, Faculdade de Filosofia Ciências e Letras, Universidade de São Paulo
Submission deadline: 10 January 2024
Address: Departamento de Biologia, FFCLRP/USP, Av. Bandeirantes, 3900, Monte Alegre, Ribeirão Preto, SP BRAZIL
Applications to: dsamorim@usp.br

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Diptera ARE Amazing!

A teneral female *Platypeza* sp. rests on the bark of a young tree. Manual high-mag focus stack by Zachary Dankowicz.

A pair of *Tachytrechus rotundipennis*, locked in a fearsome embrace in the fight for territory. Photograph by Zachary Dankowicz.

This beautiful robber fly (Asilidae) was found by Aleida Ascenzi, Valerio Caruso and Niina Kiljunen, on a branch of a tree with its lepturine cerambycid prey, near Fallen Leaf Lake, South Lake Tahoe, California, during a nature trip after the ICDX. Photo by Niina.

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BOOKS AND PUBLICATIONS

Book review:
Manual of Afrotropical Diptera.
Volume 3. Brachycera—Cyclorrhapha, excluding Calyptratae

Severyn Korneyev

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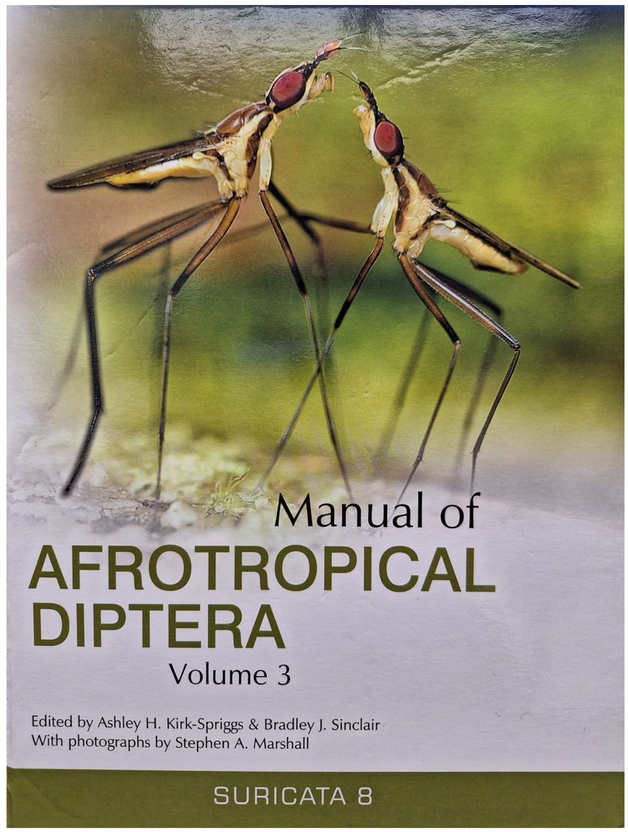


Kirk-Spriggs, A.H. & Sinclair, B.J. (Editors). 2021. Manual of Afrotropical Diptera. Volume 3. Brachycera—Cyclorrhapha, excluding Calyptratae. Suricata 8. South African National Biodiversity Institute, Pretoria, xv + 1365–2379.

In September 2022 I was sorting and identifying Diptera at the Bohart Museum of Entomology (University of California, Davis). Most of the material was from California, but eventually three drawers of flies from Democratic Republic of the Congo appeared on my table. I took the recently delivered Manual of Afrotropical Diptera Volume 3 from the shelf and dived into the colorful world of Afrotropical Diptera, mostly Chloropidae, Ephydridae and Platystomatidae. The book showed itself to be extremely helpful to sort through different genera in a very little time.

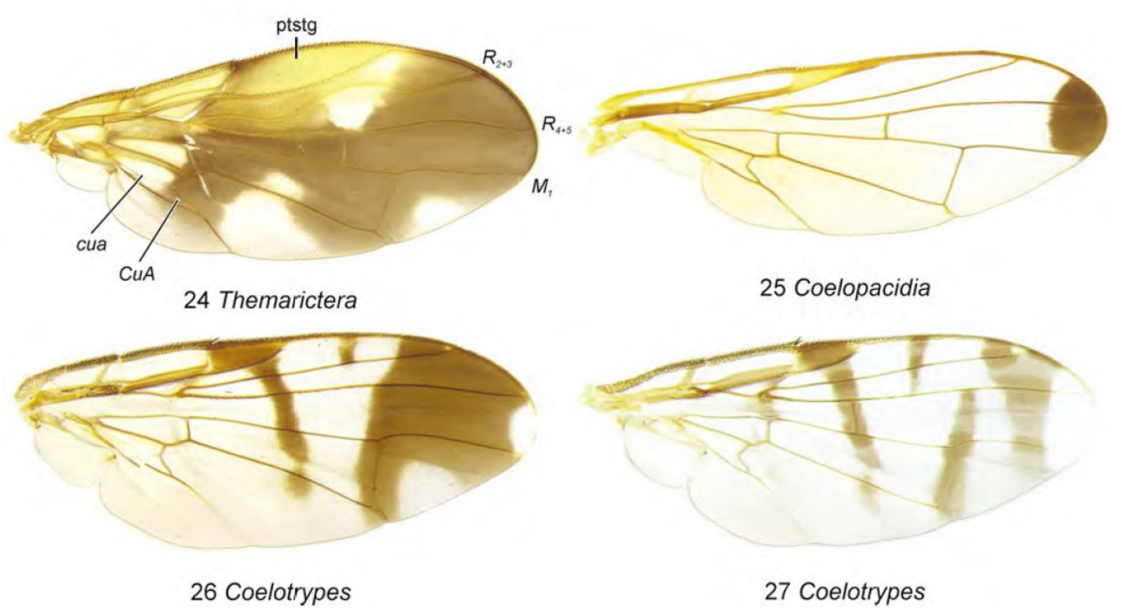
Volume 3 is an incredible tool for 51 families of Diptera, covers the Brachycera through Cyclorrhapha, written by 53 authors. It has 1032 pages with 3,440 illustrations: 1,746 color, 101 black and white images and 1,600 line drawings of flies. I would like to thank Ashley H. Kirk-Spriggs and Bradley J. Sinclair – without them, this book would never have happened. Publication of this book opens a portal to the world of Afrotropical Diptera that will significantly boost research in the region due to creating a new level of entry for researchers interested in Diptera.

Some chapters follow the traditional approach given in the Manuals of Nearctic and Neotropical Diptera, using only drawings, but most of the chapters are incredibly well illustrated with both high-resolution photos and drawings, which make it very easy to follow the given identification keys. Each chapter has a diagnosis of the family, information about biology and immature stages, economic importance, and an identification key to the genera. Also, the synopsis of the Afrotropical fauna for each genus is given, arranged in alphabetical order.

I am mostly interested in Tephritoidea and in this review I focus most on these chapters within my expertise. I used these chapters for the last year to process material at the California State Collection of Arthropods (in Sacramento, where I work), the Bohart Museum of Entomology, and the California Academy of Sciences (in San Francisco).

Tephritoidea

There is no one who knows African **Tephritidae** as well as the authors of this chapter. David Hancock is a unique expert with incredibly deep knowledge of African, Oriental, and Australian Tephritidae. Amnon Friedberg is probably the only person in the world who knew what is really going on in the subfamily Tephritinae on the African continent. Sadly, Amnon passed away on 10 October 2020 and Ariel-Leib-Leonid Friedman finished his part of this work. Together, Amnon and David, created an incredible tool for identification of 149 described and 3 undescribed genera of Tephritidae. With so many taxa, it was an incredible achievement to give high resolution pictures of wing patterns for all genera (Fig. 1). I personally would love to see habitus photos for African Tephritidae and other characters, given as separate figures. I believe this would reduce the number of incorrect identifications and make the key easier to follow, but even without habitus pictures the chapter is gorgeous.



Figs 71.22–27. Wings of Tephritidae (dorsal views): (22) *Ocnerioxa pennata* Speiser; (23) *Ptiloniola tripunctulata* (Karsch); (24) *Themarictera flaveolata* (F.); (25) *Coelopacidia punctum* (Enderlein); (26) *Coelotrypes major* (Bezzi); (27) *C. pulchellinus* (Hering).

Fig. 1. Manual of Afrotropical Diptera Volume 3, page 1675, wings of Tephritidae.

Ulidiidae, Pyrgotidae and **Ctenostylidae** chapters are given in classic monochrome style, and I enjoyed working with these chapters, as they were clear and straightforward.

Among Tephritoidea I was really amazed by the **Platystomatidae** chapter, prepared by Andrew E. Whittington and Ashley H. Kirk-Spriggs. For me, this chapter significantly simplified processing of African Platystomatidae from the California State Collection of Arthropods. The chapter has all the genera representatives photographed laterally, and in addition, separately all the wing patterns and all the important pictures for identification are given. Also, the chapter has a lot of incredible photos by S.A. Marshall with the files in the wild. Unfortunately, it is the only chapter of Tephritoidea with multiple colorful pictures in nature. Having habitus pictures all together (Fig. 2), allows me to learn and memorize genera faster and significantly increase speed of identification. This is incredibly helpful for people who work in pest diagnostics with a broad range of insects, having everyday multiple samples and limited time.

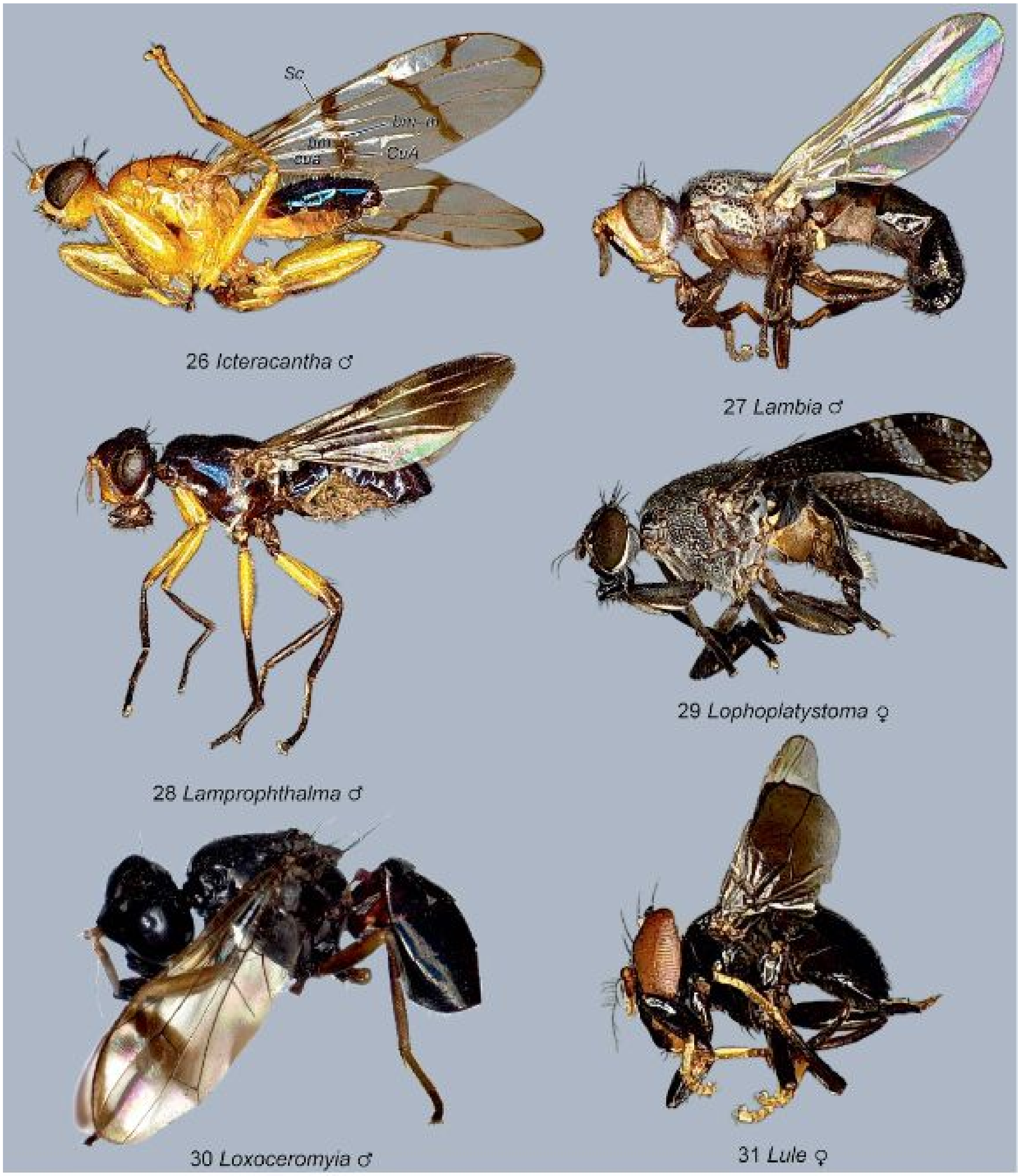


Fig. 2. Manual of Afrotropical Diptera Volume 3, page 1625, habitus of Platystomatidae.

Non-tephritoids
Among the material from the Democratic Republic of the Congo at the Bohart Museum were a lot of **Chloropidae** flies. I had no experience before with African chloropids, so for me it was very entertaining to go through them together with the excellent chapter by John & Barbara Ismay and John Deeming.

A similar approach, with high resolution photographs, was used by Steve Gaimari for his chapters: **Lauxaniidae, Celyphidae, Chamaemyiidae** and **Odiniidae**, and for all of them the keys worked very well and the chapters were well written. Chapters about **Syrphidae, Conopidae, Milichiidae** and **Drosophilidae** are also look incredible and work very well.

So far, I was not able to test the book on every family, but I hope I will have such chance in the future.

The book costs a reasonable price for such a gigantic volume – £135.00. PDF files of the chapters can be found on the internet or are shared by the authors.

Personally, I can't wait to see Volume 4 with the chapters about Calyptratae flies published.

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SOCIETY BUSINESS

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

No documents are provided in this issue, as the minutes of the annual meeting of Directors, held on 10 December 2023, will be approved before and published in the next issue.

However, we do have some information of immediate import and effect:

1) We here announce that the name of the society has been changed! The new name is "**The Dipterists Society**", with the subtext "**An International Society for Dipterology**". There is still considerable work to do to make it fully realized, e.g., in legal documents, on the website, etc., but just letting everyone in on the fact that we have a new name. There will be a more formal announcement, with details, in the next issue of *Fly Times*!
2) We here welcome a new Officer, Giar-Ann Kung (Natural History Museum of Los Angeles County), as our Education Chairperson

As of this writing, following are the Directors and the Officers of the Society.

Directors

Stephen Gaimari
Jessica Gillung
Martin Hauser
Christopher Borkent

Officers

Stephen Gaimari, President
Martin Hauser, Vice President
Christopher Borkent, Treasurer
Giar-Ann Kung, Education Chairperson
Jessica Gillung, Meeting Chairperson
Barbara Hayford, Field Meeting Co-Chair
Andrew Fasbender, Field Meeting Co-Chair

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