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HOSTING

Fly Times is a publication of the North American Dipterists Society. All issues are hosted on the webpages of the Society, at both the new website https://dipterists.org and the longstanding website http://nadsdiptera.org. The new website is fully hosted by the Society, with webmaster Steve Gaimari. The latter website, with Jim O'Hara as webmaster, is kindly hosted by the University of Guelph through arrangement with Steve Marshall.

DISTRIBUTION

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

SCOPE

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

INSTRUCTIONS TO AUTHORS

Although not a peer-reviewed journal, all submissions are carefully considered by the editor before acceptance. We encourage submissions from dipterists worldwide on a wide variety of topics that will be of general interest to other dipterists, 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 .doc preferred) along with separate image files (.jpg or .png preferred).

Following are some specific do's and don't's, bearing in mind that consistency among manuscripts is important:

1) *Do not* embed images into the text file (but *do* indicate in the text file approximately where each image should be placed).
2) *Do* submit image files of a reasonable size (no more than about 2MB per image file).
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 (spring issue) and early December (fall issue). For larger manuscripts your submissions may be considered for inclusion in the *Fly Times Supplement* series.

Please submit manuscripts to the editor-in-chief, Stephen Gaimari, at:

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and cc sgaimari@dipterists.com

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The North American Dipterists Society is a 501(c)(3) nonprofit organization, 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.

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*! This issue is once again brought to you during the Covid-19 pandemic, and with new variants seemingly popping up all too often. We are all hopeful that things will improve as time passes. As usual, I am impressed with the variety of excellent submissions, and I hope they are enjoyable to the readers. And as seems to be typical, I am right at the edge of this being a true spring issue. My intention is always to have it out a bit earlier, but manuscripts seem to come in until the last minute! Please consider writing an article or two for the next issue, which is slated for fall of 2022. And for larger works, please consider the *Fly Times Supplement* series, which can be found at https://dipterists.org/fly_times_supplement.html.

Also note, I am hoping to improve the front and back covers of the *Fly Times*. Some of you clever dipterists might have good ideas for this – please consider submitting them! There are several options – to have different covers with each issue, or like most journals, to have a static cover issue to issue. Or even to switch it up each year, or every once in a while. So please send your design ideas (8-1/2" x 11") to me at sgaimari@gmail.com (cc sgaimari@dipterists.org).

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NEWS

An extensive collection of the insect fauna of the Valdivian forest, in the Parque Nacional Puyehue, southern Chile

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Back in 2011, there was a field trip in Chile for 5-weeks by a team with David Yeates, Bryan Lessard and one of us (DSA). The trip covered from the Parque Nacional Bosque Fray Jorge (30° 39’ S), north of Santiago, to the Parque Nacional Alerce Andino (41° 34’ S), south of Puerto Montt. The core goal of the trip was to collect tabanids across this large portion of Chile. Specimens collected were later used in a study of the evolution of Scionini pangonines, disjunct between Australia and southern South America (Lessard et al. 2013, Lessard 2014), in which conclusions corroborated a hypothesis of a late (Cenozoic) vicariance in southern Gondwanan terranes (Amorim et al. 2009, Almeida et al. 2012).

The austral temperate flora and fauna has long attracted the attention of science. Joseph D. Hooker (1817–1911) published three large volumes on the flora of the southern end of the world—*Flora Antarctica* (1844–1847), *Flora Novae-Zelandiae* (1851–1853) and *Flora Tasmaniae* (1853–1859). The uniqueness of the insect fauna of Chile has also attracted the attention of numerous entomologists and collectors for over 150 years. There is a nice summary on entomology in Chile prepared by Cortés & Herrera (1989) (see also O’Hara et al. 2021). After the pioneer studies of Claudio Gay (1844–1871), Francis Walker (1849), C.E. Blanchard (1854), R.A. Philippi (1865), the Shannon-Edwards expedition to Patagonian Argentina and southern Chile in the 1920s produced one of the most impressive reports ever prepared on a fly regional fauna. The *Diptera of Patagonia and South Chile*, published by the British Museum (Natural History) between 1929 and 1951, was mainly based on the Shannon-Edwards expedition material at the British Museum (Natural History), with some few other sources of material. These books were authored by leading dipterists of the time and provided the foundation for the study of southern South American flies (Alexander, 1929; Tonnoir, 1929; Edwards, 1929a-d; van Duzee, 1930; Aubertin, Krober & Edwards, 1930; Collin, 1933; Malloch, Edwards & Bromley, 1932; Schmitz, 1929; Schmitz, Collin, Richards & Cresson 1931; Edwards, Shannon, Aubertin & Malloch, 1933; Malloch, 1933, 1934a, 1948; Aldrich, 1934; Malloch, 1934b; Hall & Smart, 1937). Slightly later, there was a Danish scientific expedition to Patagonia and Tierra del Fuego that collected 6,530 specimens of flies from localities primarily of southern Argentina, but also some from Chile, including a sample from the Parque Nacional Puyehue (Schmidt Nielsen 1980).

The growth of dipterology in Chile in more recent years had important contributions from Raúl Cortés, working mostly with tachinids (see complete references in O'Hara et al. 2021) and Christian González (e.g., González et al., 2019; González & Elgueta 2020; González et al. 2021; O'Hara et al. 2021). The Chilean coleopterist Luis Enrique Peña made extensive collections in different parts of Chile, acquired especially by the Canada National Collection, the Museu de Zoologia da Universidade de São Paulo, and the United States National Museum. We should also add, in terms of fieldwork in Chile, the collecting of Lubomir Masner, Sixto Coscarón, Mounty Wood, Marc Pollet, Brian Brown, and Steve Marshall, among others. Some coleopterists, such as Alfred F. Newton, Margareth K. Thayer and Stewart B. Peck, made extensive collections in southern Chile (see, e.g., Bickel 2007).

A taxon-oriented geographically spread design of fieldwork is necessary to answer some questions underlying biodiversity studies. But there are other ways, however, to address biodiversity questions. The importance of single-site knowledge of "All Diptera" fauna received international attention with the study in Zurquí, Costa Rica, led by Brian V. Brown and Art Borkent (Brown & Borkent 2012, Brown et al. 2018, Borkent et al. 2018). We want to report here an ongoing project with intensive collecting in southern Chile, which may move to another level our understanding of the fly temperate fauna of South America. In 2017, we decided to make an intensive, single-site approach of flies in southern Chile. At a first stage, with the support of Mario Elgueta, at the Museo Nacional de Historia Natural, Santiago, and of Christian González, from the Universidad Metropolitana de Ciencias de la Educación, we obtained the due permits, paperwork and connections to collect in the Parque Nacional Puyehue for 4-weeks in late January/early February 2017.

The Parque Nacional Puyehue has 1,070 Km² and is at the latitude of Osorno, near 40.7°S, about five kilometers east of Lake Puyehue (Fig. 11). The park harbors its administration at Aguas Calientes, which also has a small public exhibition and a hotel. This sets exceptionally convenient logistics for intensive collecting, avoiding time spent daily with traveling. The site of Aguas Calientes is at the altitude of about 440 m; the locality of Antillanca, also within the park, is at the higher limit of vegetation in that area, between 1,000 and 1,300 m. Aguas Calientes had a number of timber mills until the 1930s, and the park was created in 1941. During these last 80 years, the park protected the original vegetation, and impacted areas have recovered continuously.

The sampling of the fly fauna in Parque Nacional Puyehue in 2017 rendered some rare findings. Among many other interesting catches, this includes:

(1) the fungus gnat *Freemanomyia elongata* (Freeman), of unclear family affinity, known from only a few recorded specimens (Fig. 1);
(2) the second record of the minute empidoid *Gondwanamyia chilensis* Cumming & Saigusa (Fig. 2), a genus also known from New Zealand (Sinclair et al. 2016);
(3) the sciadocerine phorid *Archiphora patagonica* White (Fig. 3), that has the New Zealand *Sciadocera rufomaculata* White as its sister species;
(4) the nephrocerine pipunculid *Protonephrocerus flavipilus* Skevington, Marques & Rafael (Skevington et al. 2021) (Fig. 4);
(5) an important number of southern South American heleomyzid genera (Fig. 5);
(6) the acalyptrate *Mayomyia diversipennis* Malloch (Fig. 6) and *Melantomyza polita* Malloch (Fig. 6), also with unclear affinities;
(7) two teratomyzid species, *Teratoptera chilensis* Malloch (Fig. 7), described from Chile, and another species of *Teratoptera* (Fig. 8);
(8) pallopterid species of the genera *Aenigmatomyia* and *Pseudopyrgota*;

(9) species of the paraleucopid genera *Mallochianamyia* and *Schizostomyia*; and, most unexpectedly,

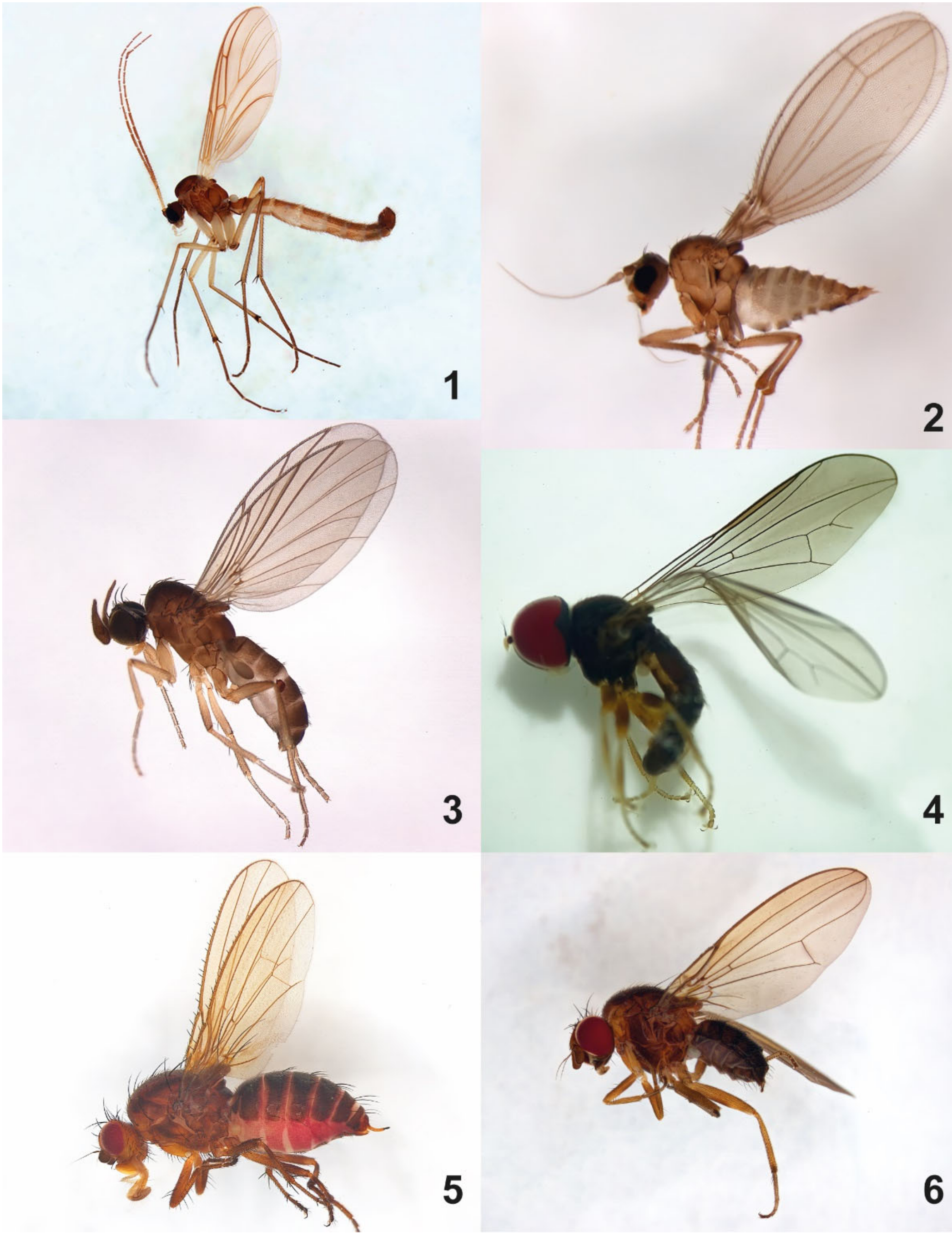
(10) the first non-Palearctic species of Opetiidae, *Puyehuemyia chandleri* Amorim, Silva & Brown (Amorim et al. 2018) (Fig. 10).

The findings of that expedition were so important that we decided to move on to a second stage, with trapping along one full year. The collaboration with the entomologists in Santiago and the participation of park authorities and personnel brought up the necessary solutions. In November 2019, one 6 meters Gressitt-style Malaise trap and two Townes-style Malaise traps were set in the area of Aguas Calientes, close to the park base; and two Townes-style Malaise traps were set at Antillanca. Park rangers were responsible for managing the traps, with regular online communication. The first plans were to have sampling until November 2020, with a major blitz for sorting at the park, including additional collecting and outreach activity. The pandemic, of course, forced a revision of the plans. The park administration and the rangers were committed to the process of collecting samples even along the period with some restrictions. Collecting in Antillanca went until June 2020, when the traps were destroyed by the snow—quite expectedly. Collecting in Aguas Calientes went until July 2021, with a gap between July and December 2020, when there were no personnel available to manage the traps.

The samples were kept safe at the park until November 2021, when they could be processed and transported. We now began the process of sorting the huge amount of material available. Late autumn and early winter samples are considerably small, but the late spring and early summer samples are massive. For this second stage of the project, there are 75 two-week samples available from the sites.

Because of marked seasonality for many groups of insects, the lack of late winter and early spring samples restrains our knowledge of the fauna. One of the July samples at Termas, for example, has five females of *Perissomma congrua* Colless, entirely absent in other samples we have seen so far. There is evidence at least for some Diptera present exclusively in late autumn, winter and/or early spring in a subantarctic forest of southernmost Chile (Elgueta 1993). We decided, hence, to move to another stage. Traps will be set again in June 2022 and kept running until December 2022. We will add as collecting sites the area with the best-preserved forest patches, in Lago Toro, and have as well traps at the level of Lake Puyehue (~200 m of altitude).

This kind of long-term approach is highly dependent on solutions for logistics and large collaboration. The first step, in 2016, was an agreement with the Museo Nacional de Historia Natural, in Santiago, on a protocol dealing with types and identified material: all holotypes, half of the paratypes and half of the identified specimens will go back to Chile. This is just the standard arrangement with other natural history museums. There are some rumors going around, however, that seem to discommend having types at the Museum in Santiago. This is untruthful and damaging gossip. The reason for this rumor may be that Philippi's types are not at the museum in Santiago. This issue has been largely clarified more with the information that Philippi used to sell or exchange parts of his collections (Evenhuis & Greathead 1999, Fitzgerald et al. 2020, González et al. 2018, Sforzi & Sommaggio 2021), including some Coleoptera species described by him and his son (Bezděk & Hájek 2009, 2010a,b, Hájek & Švihla 2012). Some types may have been destroyed since the Diptera collection did suffer damage in the first half of the last century, maybe by effects of earthquakes (see Marston 1970) and because there were long periods without entomologists or technicians.



Figures 1–6. 1. A male of *Freemanomyia elongata* (Freeman). The genus has been assigned to the family Mycetophilidae, but the wing venation suggests that the species is at best sister to all remaining members of the family. **2.** A female of the empidoid *Gondwanamyia chilensis* Cumming & Saigusa. The original description was based on yellow pan traps and this single specimen was collected with sweeping. We have now a good number of additional specimens collected in Malaise traps. **3.** A female of *Archiphora patagonica* White (Phoridae: Sciadocerinae), described over a century ago, but not known from many specimens. **4.** The nephrocerine pipunculid *Protonephrocerus flavipilus* Skevington, Marques & Rafael. There was a species described from the Isla de Chiloé, but maybe additional material was misidentified. We still do not have specimens from Termas, but we now have rather abundant material from Antillanca. **5.** *Blasochaetoptera*. **6.** *Mayomyia diversipennis* Malloch, a species originally left unplaced, but fit into the Heleomyzidae in some more recent classifications.

Figures 7–8. Teratomyzidae. **7.** *Teratoptera chilensis* Malloch. **8.** *Teratoptera* sp.

The question of Philippi's types is also stated in O'Hara et al. (2021), who mention that in the mid-1800s the specimens were trickling "back" to Europe into institutional and private collections. Some conflicts of interest may have also led to this idea. The matter of fact is that the collection of the Museo Nacional de Historia Natural in Santiago is very well organized and safe. The museum was able to go through the 1960, 1971, 1985, and 2010 earthquakes in Chile largely unaffected in relation to its collections. Other smaller insect collections existed in Chile, some of which have been merged with larger collections. One of the collections still standing is the Museo Entomológico Luis Peña, Departamento de Sanidad Vegetal de la Facultad de Ciencias Agronómicas, Universidad de Chile, Santiago. The collection of the Instituto de Entomología de la Universidad Metropolitana de Ciencias de la Educación, Santiago is important. The insect collection of the Estación Experimental Agronómica, Universidad de Chile, Maipú, Santiago is now merged with the Museo Entomológico Luis Peña.

The local support from the Park staff for this project was also shown to be crucial in our project. A nice solution came up in the process: at the beginning of the project, feedback on the scientific findings of our expedition was given to the park. A talk to the park rangers in 2017—"The treasure jewels"—raised a lot of interest in the project. Actually, rangers (anywhere on the planet) used to take care of highly valuable biodiversity which they are seldom told about. On a worldwide scale, rarely do taxonomists give back to the park and biological reserve staff information on the fauna collected or discovered. We later made arrangements for a wider outreach project: posters with some of the more iconic species found in the park were prepared for the public exhibition (e.g., Fig. 10) and some educational activities are being prepared for school kids at the Park in November 2022. The park administration immediately loved the idea back in 2017 and began to support the project. We trained the rangers to manage the Malaise traps and we keep updating information on family, genera and species recognized in the material.

The agreement with the Museo Nacional de Historia Natural, Santiago, includes, as mentioned above, the question of housing types. But it also has to deal with the process of handling subsamples in the long run. This concerns the issue of labs not "taking property" of samples informally, i.e., not ever returning material and sometimes never actually even working with the samples (for this part of the taxonomic impediment, see, e.g., Evenhuis 2007).

This is a complex issue and the Museum National d'Histoire Naturelle, Paris, set an interesting model of collective collaboration that includes a signed agreement for the Mitaraka biodiversity project (Touroult et al. 2018, 2021):

> "*One of the lessons learned about the organization of the specimen processing chain is the key role of the taxonomic group coordinators, as this is an effective way of multiplying and facilitating access to the collected material by a large community of experts worldwide. However, while this is undoubtedly a plus, choosing the right person for this job is all but sufficient: it is as crucial to ensure the commitment of both group coordinators and expert taxonomists by negotiating beforehand the terms of a signed agreement.*" (Touroult et al. 2021, p. 822)

For this project in Puyehue, committed coordinators for many insect orders and most fly families have been chosen to handle the samples in the long run. The responsibilities, however, on some of the dark taxa (in the sense of Hausmann et al. 2020)—e.g., collembolans, psocopterans and chironomids—are still pending. The lack of available expertise to work with so many biodiversity projects worldwide is of course part of the taxonomic impediment, in times in which there is a lot of greenwashing talks about the importance of biodiversity but scarce or no funding for the real production of primary biodiversity knowledge.

Objective goals and a time schedule are also being set to deal with the Puyehue samples and on published papers. As a first step, a technical paper should come out soon on features of the local environment, the core issues being addressed with the project and some of the initial list of findings for Diptera. In a second step, after the primary sorting is accomplished and specimens reach the specialists, identification of the material down to genera should be done within one year—to generate a paper with an overview of the fauna. Specialists who cannot invest further into the taxonomy of the group should return the material by then. In a third step, there would be another year for taxonomic studies within families/genera, after which the material should be returned to Santiago or have another agreement signed.

In the long run, we have some hypotheses to be tested, especially if we can sequence certain target families, with sampling at a number of west-east sites (Fig. 9):

(1) Is there a significant faunal turnover between the forest at lower altitudes (~500 m) and the fauna at the higher limit of the vegetation at this latitude (~1,000 m)? In other words, is there large-scale conspecificity between elements at different altitudes at the same latitude?

(2) For the cases of shared species between both altitudes, is there significant genetic divergence between the elements of lower areas and the elements of higher areas?

(3) Are there unique components of lower areas and higher areas?

(4) Is there a significant difference between the faunal composition in areas with secondary vegetation or in recovery (Aguas Calientes and Antillanca) and areas with pristine, original vegetation (Lake El Toro)?

(5) Is there a large faunal turnover between areas of the Andean mountain range (dominated by *Nothofagus dombeyi*) and the fauna of the Central Valley (200 m, dominated by *Nothofagus obliqua*)?

(6) Is there a large faunal turnover between areas of the Andean mountain range and the coastal mountain range. For the shared species between both areas, what is the percentage of genetic divergence?
(7) Where is the faunal turnover between the fauna at the latitude of Puyehue and the faunal of the Valdivian temperate rainforest to the north and to the south?

Figure 9. Map of the region with the position of Aguas Calientes and Antillanca (modified from Google Earth).

At this time, we still have a limited inference of the size of the 2-week samples. It is possible, however, that this is the largest single-site collection ever made of insects in the Valdivian forest. The mid Autumn samples (April 28 to May 12, 2021), which are considerably small, have a total of 2,715 specimens of arthropods (mostly hexapods, but also spiders, opiliones, and acari); and there are 3,221 specimens in the 6 m trap sample for the same period. The large samples may have at least three times this number of specimens.

We have high hopes that rare species from southern Chile will now be well-documented and represented in good numbers in collections. As well, that new supra-specific taxa of the southern temperate fly clades come to be discovered, as was the case of the new opetiid genus. This material will also help clarifying the position of some enigmatic genera of acalyptrates at higher-level phylogenies of flies. This kind of massive sampling will be increasingly important in the near future when barcode sequencing in a large scale will allow an acceleration of taxonomy knowledge with limited cost (Wang et al. 2018, Yeo et al. 2021, Hartop et al. preprint, Srivathsan et al. preprint).

This project also raises the Museo Nacional de Historia Natural of Santiago to the condition of a key player in international studies of the temperate Neotropical insect fauna. A takeaway lesson from our project is that much more can be achieved with collaboration—much more than can be done traveling around for a few weeks, with barely any sampling of a fauna that has strong seasonal variation. Collaboration and agreements with the museum in Santiago can help with permits, planning, logistics and contacts at the national level (central office of Corporación Nacional Forestal, CONAF, responsible of the National Protected Areas), regional level (office at the respective administrative region where is located the protected area), and park staff.

UMCE

Museo Nacional de Historia Natural Chile

Parque Nacional Puyehue

Puyehuemyia chandleri Amorim, Silva & Brown 2018

Esta especie pertenece a una familia de moscas denominada Opetiidae de la que solo se conocía el género *Opetia*, con tres especies de Europa y Asia. Así que el género *Puyehuemyia* es el único representante de esta familia conocido de todo el Hemisferio Sur.

El género y su única especie se describieron en base a un único ejemplar (una hembra) recolectado en los bosques de Puyehue. Es una de las especies más particulares que se ha recolectado en el Parque Nacional Puyehue nominándose el género con el nombre *Puyehuemyia* (mosca de Puyehue) como un homenaje al parque. El único ejemplar conocido se guarda en la colección científica del Museo Nacional de Historia Natural, en Santiago.

Es posible que las larvas de esta especie sean parasitoides de otros insectos.

Figure 10. Example of a poster with a species found in the park, with the photo of *Puyehuemyia chandleri* Amorim, Silva & Brown.

Finally, giving information back to the park and to the population does not take much time, is a lot of fun, brings further help to the fieldwork, with information about best spots for trapping etc. This should be part of our business as taxonomists: part of the population is eager to know more about biodiversity. Along with previous steps of this project, two articles in local newspapers were published (https://www.soychile.cl/Osorno/Sociedad/2021/12/12/735911/descubren-nueva-especie-insecto-puyehue.aspx and https://paginav.cl/2021/12/13/entomologos-investigan-rica-diversidad-de-insectos-en-parque-nacional-puyehue/) and one radio interview was aired about the ongoing research.

We strongly recommend following Chilean legislation (CONAF rule II.A.8: "8. Si, como fruto de la investigación, se describen nuevas especies para la ciencia, el material de holotipos deberá ser entregado a la custodia del Museo Nacional de Historia Natural de Chile" (CONAF 2013)) that primary types, part of paratypes and identified specimens be sent back to Chile, and that faunal research projects be developed in collaboration with Chilean specialists. The rationale of our project, hence, is to strengthen Chilean dipterology and the Santiago National Museum of Natural History as the institutional backbone working with foreigners participating in this process. If you are a specialist that agrees with the terms and is interested to deal with questions pointed out here or know others that may be interested in collaborating, please contact the project's Chilean entomological leadership.

References

Alexander, C.P. 1929. Crane-flies (Tipulidae, Trichoceridae, Tanyderidae), pp. 1–240. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History).* Part I. British Museum Natural History, London.

Aldrich, J. M, 1934. Tachinidae, pp. 1–170. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VII-Fascicle 1. British Museum of Natural History, London.

Almeida, E.A.B., M.R. Pie, S.G. Brady, and B.N. Danforth. 2012. Biogeography and diversification of colletid bees (Hymenoptera: Colletidae): emerging patterns from the southern end of the world. *Journal of Biogeography* 39: 526–544.

Amorim, D.S., C.M.D. Santos, and S.S. Oliveira. 2009. Allochronic taxa as an alternative model to explain circumantarctic disjunctions. *Systematic Entomology* 34: 2–9.

Amorim, D.S., Silva, V.C. & Brown, B.V. 2018. *Puyehuemyia chandleri*, gen.n., sp.n. (Diptera: Opetiidae): remnant of a Cretaceous biota in Chile. *American Museum Novitates* 3892:1–27.

Aubertin, D., 1930. Stratiomyidae, pp. 93–105. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part V-Fascicle 2. British Museum (Natural History), London.

Aubertin, D. & J.R. Malloch. 1933. Conopidae, pp. 171–175. *In*: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 3. British Museum of Natural History, London.

Borkent, A., Brown, B.V., Adler, P.H., Amorim, D.S., Barber, K., Bickel, D., Boucher, S., Brooks, S.E., Burger, J., Burington, Z.L., Capellari, R.S., Costa, D.N.R., Cumming, J.M., Curler, G., Dick, C.W., Epler, J.H., Fisher, E., Gaimari, S.D., Gelhaus, J., Grimaldi, D.A., Hash, J., Hauser, M., Hippa, H., Ibáñez–Bernal, S., Jaschhof, M., Kameneva, E.P., Kerr, P.H., Korneyev, V., Korytkowski, C.A., Kung, G.–A., Kvifte, G.M., Lonsdale, O., Marshall, S.A., Mathis, W., Michelsen, V., Naglis, S., Norrbom, A.L., Paiero, S., Pape, T., Pereira–Colavite, A., Pollet, M., Rochefort, S., Rung, A., Runyon, J.B., Savage, J., Silva, V.C., Sinclair, B.J., Skevington, J.H., Stireman III, J.O., Swann, J., Vilkamaa, P., Wheeler, T., Whitworth, T., Wong, M., Wood, D.M., Woodley, N., Yau, T., Zavortink, T.J. & Zumbado, M.A. 2018. Remarkable fly (Diptera) diversity in a patch of Costa Rican cloud forest: Why inventory is a vital science. *Zootaxa* 4402: 53–90.

Bezděk, A. & Hájek, J. 2009. Catalogue of type specimens of beetles (Coleoptera) deposited in the National Museum, Prague, Czech Republic - Scarabaeoidea: Bolboceratidae, Geotrupidae, Glaphyridae, Hybosoridae, Ochodaeidae and Trogidae. *Acta Entomologica Musei Nationalis Pragae* 49(1): 297–332.

Bezděk, A. & Hájek, J. 2010a: Catalogue of type specimens of beetles (Coleoptera) deposited in the National Museum, Prague, Czech Republic. Scarabaeidae: Dynamopodinae, Dynastinae, Melolonthinae and Rutelinae. *Acta Entomologica Musei Nationalis Pragae* 50: 279–320.

Bezděk, A. & Hájek, J. 2010b: Catalogue of type specimens of beetles (Coleoptera) deposited in the National Museum, Prague, Czech Republic. Scarabaeidae: Cetoniinae (including Trichiini) and Valginae. *Acta Entomologica Musei Nationalis Pragae* 50: 629–655.

Bickel, D. 2007. *Pharcoura* (Diptera: Dolichopodidae), a new genus from Chile. *Tijdschrift voor Entomologie* 150: 5–12.

Blanchard, C.E. 1854. Orden IX. Dipteros, pp. 327–468. In: Gay C (Ed), *Historia física y política de Chile según documentos adquiridos en esta república durante doce años de residencia en ella y publicada bajo los auspicios del supremo gobierno. Zoologia*. Tomo sétimo. Paris, 471 pp. [Cover of volume bears date of 1852.]

British Museum (Natural History) Department of Entomology 1929-1951. *Diptera of Patagonia and South Chile: based mainly on material in the British Museum (Natural History)*. London, British Museum. 7 v.

Brown, B.V., Borkent, A., Adler, P.H., Amorim, D.S., Barber, K., Bickel, D., Boucher, S., Brooks, S.E., Burger, J., Burington, Z.L., Capellari, R.S., Costa, D.N.R., Cumming, J.M., Curler, G., Dick, C.W., Epler, J.H., Fisher, E., Gaimari, S.D., Gelhaus, J., Grimaldi, D.A., Hash, J., Hauser, M., Hippa, H., Ibáñez–Bernal, S., Jaschhof, M., Kameneva, E.P., Kerr, P.H., Korneyev, V., Korytkowski, C.A., Kung, G.–A., Kvifte, G.M., Lonsdale, O., Marshall, S.A., Mathis, W., Michelsen, V., Naglis, S., Norrbom, A.L., Paiero, S., Pape, T., Pereira–Colavite, A., Pollet, M., Rochefort, S., Rung, A., Runyon, J.B., Savage, J., Silva, V.C., Sinclair, B.J., Skevington, J.H., Stireman III, J.O., Swann, J., Vilkamaa, P., Wheeler, T., Whitworth, T., Wong, M., Wood, D.M., Woodley, N., Yau, T., Zavortink, T.J. & Zumbado, M.A. 2018. Comprehensive inventory of true flies (Diptera) at a tropical site. *Biology Communications* 1(21): 1–8.

Collin, J.E. 1933. Empididae, pp. 1–334. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part IV. British Museum Natural History, London.

CONAF (Corporación Nacional Forestal, Gerencia de Áreas Silvestres Protegidas). 2013. Manual de Procedimientos, Requisitos y Obligaciones para Proyectos de Investigación Científica en el Sistema Nacional de Áreas Silvestres Protegidas del Estado – SNASPE (available at https://www.conaf.cl/wp-content/files_mf/1377635097ReglamentodeInvestigacionesenelSNASPE2013.pdf).

Cortés R. & Herrera J. 1989. Antecedentes historicos y bibliograficos para una historia de la entomologia en Chile. *Acta Entomológica Chilena* 15: 297–321.

Cresson, E. T., Jr. (1931). Family Ephydridae, pp. 85–116. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History).* Part VI-Fascicle 2. British Museum of Natural History, London.

Edwards, F. W. 1930c. Bibionidae, Scatopsidae, Cecidomyiidae, Culicidae, Thaumaleidae (Orphnephilidae), Anisopodidae (Rhyphidae), pp. 77–119. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part II-Fascicle 3. British Museum Natural History, London.

Edwards, F.W. 1933. Ephydridae (Supplement), pp. 117–119. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 3. British Museum of Natural History, London.

Edwards, F.W., 1929. Blepharocidae, pp. 33–75. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part II-Fascicle 2. British Museum Natural History, London.

Edwards, F.W., 1931. Chironomidae, p. 233–324. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part II-Fascicle 5. British Museum Natural History, London.

Elgueta D., M. 1993. Invertebrados asociados a suelo en bosque de *Nothofagus pumilio* (Poepp. et Endl.) Krasser, XII Región – Chile, con especial referencia a Insecta. *Revista Chilena de Entomología* 20: 49–60.

Evenhuis, N.E. 2007. Helping solve the "Other" taxonomic impediment: completing the eight steps to total enlightenment and taxonomic Nirvana. *Zootaxa* 1407(1): 3–12.

Evenhuis, N.L. & Greathead, D.J. (1999) World catalog of bee flies (Diptera: Bombyliidae). Backhuys Publishers, Leiden, xlviii + 756 pp. [I do not have this book, but you can see p. 25, 164, and 540]

Fitzgerald, S.J., González, C.R. & Elgueta, M. 2020. A catalog of the Bibionidae (Diptera: Bibionomorpha) of Chile. Zootaxa, 4766(1): 48–60. [see p. 56]

Freeman, P., 1951. Mycetophilidae, p. 1–138. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part III. British Museum of Natural History, London.

Gay, C. 1844-1871. *Historia física y política de Chile según documentos adquiridos en esta república durante doce años de residencia en ella y publicada bajo los auspicios del supremo gobierno*. Paris en casa del autor. Chile en el Museo de Historia Natural de Santiago.

González, C.R., Elgueta, M. & Ramírez, F. 2018. A catalog of Acroceridae (Diptera) from Chile. *Zootaxa*, 4374(3): 427–440.

Hájek, J. & Švihla, V. 2012: Catalogue of the type specimens of beetles (Coleoptera) deposited in the National Museum, Prague, Czech Republic. Trogossitidae, Cleridae, Prionoceridae, Melyridae, Dasytidae, Rhadalidae and Malachiidae. *Acta Entomologica Musei Nationalis Pragae* 52: 603–654.

Hartop, E., Srivathsan, A., Ronquist, F.& Meier, R. 2021. Large-scale Integrative Taxonomy (LIT): resolving the data conundrum for dark taxa. *BioRXiv* (pre-print).

Hausmann, A., Krogmann, L., Peters, R., Rduch, V., Schmidt S. 2020. GBOL III: Dark Taxa. Available from https://ibol.org/barcodebulletin/research/gbol-iii-dark-taxa/.

Ingram, A. & Macfie, J. W. S. 1931. Ceratopogonidae, pp. 155–232. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part II-Fascicle 4. British Museum Natural History, London.

Lessard, B.D. 2014. Revision of the austral horse fly tribe Scionini (Diptera: Tabanidae). *Austral Entomology* 53: (2), 203–239. https://doi.org/10.1111/aen.12076

Lessard, B.D., Cameron, S.L., Bayless, K.M., Wiegmann, B.M. & Yeates, D.K. 2013. The evolution and biogeography of the austral horse fly tribe Scionini (Diptera: Tabanidae: Pangoniinae) inferred from multiple mitochondrial and nuclear genes. *Molecular Phylogenetics and Evolution* 68 (3): 516–540. https://doi.org/10.1016/j.ympev.2013.04.030

Malloch, J.R. (1932*)*. Rhagionidae (Leptidae), Therevidae, Scenopenidae, Mydaidae, Asilidae, Lonchopteridae, pp. 1–257. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History).* Part V-Fascicle 3. British Museum of Natural History, London.

Malloch, J.R. 1933. Acalyptrata (Helomyzidae, Trypetidae, Sciomyzidae, Sapromyzidae, etc.), pp. 177–391. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 4. British Museum of Natural History, London.
Malloch, J.R. 1934. Acalyptrata (concluded) , pp. 393–489. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 5. British Museum of Natural History, London.
Malloch, J.R. 1934. Muscidae, p. 171–346. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VII-Fascicle 2. British Museum of Natural History, London.
Malloch, J.R., 1948. Key to the families of Acalyptrata, with notes on some of the families, pp. 491–507. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 6. British Museum of Natural History, London.
Marston, N. 1970. Revision of New World Species of *Anthrax* (Diptera: Bombyliidae), other than the *Anthrax albofasciatus* group. *Smithsonian Contributions to Zoology* 43: 1–148.
O'Hara, J.E., Wood, D.M. & González C.R. 2021. Annotated catalogue of the Tachinidae (Insecta, Diptera) of Chile. *ZooKeys* 1064: 1–200. https://doi.org/10.3897/zookeys.1064.62972
Philippi, R.A. 1865. Aufzählung der Chilenischen Dipteren. Verhandlungen der Zoologisch-Botanischen Gesellschaft in Wien. *Verhandlungen der Zoologisches-botanisches Gesellschaft Wien* 15: 595–782.
Schmidt Nielsen, E. 1980. Entomology. Pp. 9–13. In: Madsen, H.B., Nielsen, E.S. & Ødum, S. (eds.). 1980. The Danish Scientific Expedition to Patagonia and Tierra del Fuego. *Geografisk Tidskrift* 80: 1–28.
Schmitz, H. 1929. Sciadoceridae and Phoridae, p. 1–42. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History).* Part VI-Fascicle 1. British Museum of Natural History, London.
Sforzi, A. & Sommaggio, D. 2021. Catalog of the Diptera types described by Camillo Rondani. *Zootaxa* 4989(1): 1–438.
Shannon, R.C. & Aubertin, D. 1933. Syrphidae, pp. 120–170. In*: Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part VI-Fascicle 3. British Museum of Natural History, London.
Srivathsan, A., Lee, L., Katoh, K., Hartop, E., Kutty, S.N., Wong, J., Yeo, D. & Meier, R. In press. MinION barcodes: biodiversity discovery and identification by everyone, for everyone. *bioRxiv*. https://doi.org/10.1101/2021.03.09.434692
Tonnoir, A.L. 1929. Psychodidae, pp. 1–32. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part II-Fascicle 1. British Museum Natural History, London.
Touroult, J., Pollet, M. & Pascal, O. 2018. Overview of Mitaraka survey: research frame, study site and field protocols. In: Touroult, J. (ed.), "Our Planet Reviewed" 2015 large-scale biotic survey in Mitaraka, French Guiana. *Zoosystema* 40 (13): 327–365.
Touroult, J., Pascal, O., Barnier, F. & Pollet, M. 2021. The "Our Planet Reviewed" Mitaraka 2015 expedition: a full account of its research outputs after six years and recommendations for future surveys. In: Touroult, J. (ed.), "Our Planet Reviewed" 2015 large-scale biotic survey in Mitaraka, French Guiana. *Zoosystema* 43(32): 811–833. http://zoosystema.com/43/32, https://doi.org/10.5252/zoosystema2021v43a32
Van Duzee, M.C. 1930. Dolichopodidae , pp. 1–92. In: *Diptera of Patagonia and South Chile based mainly on material in the British Museum (Natural History)*. Part V-Fascicle 1. British Museum (Natural History), London.
Walker, F. 1849. *List of the specimens of dipterous insects in the collection of the British Museum*. Part IV. British Museum, London. [3] + 689–1172 pp.

Wang, W.Y., Srivathsan, A., Foo, M., Yamane, S.K. & Meier, R. 2018. Sorting specimen-rich invertebrate samples with cost-effective NGS barcodes: validating a reverse workflow for specimen processing. *Molecular Ecology Resource*s 18: 490–501.

Yeo, D., Srivathsan, A., Puniamoorthy, J., Maosheng, F., et al. 2021. Mangroves are an overlooked hotspot of insect diversity despite low plant diversity. *BMC Biology*. https://doi.org/10.1101/2020.12.17.423191

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Kleptoparasitic chloropids and acacia ants in Guanacaste, Costa Rica

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A couple of recent notes and papers have drawn attention to the association between Chloropidae and *Pseudomyrmex* ants that occur only on host bull's horn acacias (*Vachellia* species). The first report of chloropids acting as kleptoparasites by stealing the extrafloral nectar provided by the acacias for their *Pseudomyrmex* inhabitants was by Barrantes et al. (2018), who reported *Notaulacella octicola* Sabrosky interacting with *Pseudomyrmex spinicola* on *Vachellia collinsi* in Guanacaste Province, Costa Rica. A note in *Fly Times* by Ana Rita Gonçalves (2019) added some further observations of a similar association between *Pseudomyrmex* sp. and unidentified chloropids in Mexico, and I here add a little more to the story with observations and photos of another genus of Chloropidae associated with a different species of *Pseudomyrmex* on the Guanacaste coast.

During a brief visit to Playa Hermosa, Guanacaste on May 15, 2022 (while waiting to catch a flight out of Liberia) I walked up a trail off the north end of the beach to check out a patch of bull's horn acacia (probably *V. cornigera*) populated by *Pseudomyrmex flavicornis.* The attached photos show the hollow thorns housing the nests and some of the extrafloral nectaries provided for the ants, and they also show a small chloropid feeding from the extrafloral nectaries. There were several of these chloropids, dashing around the ants and feeding on the nectaries, apparently without provoking any reaction from the ants. The chloropids appear to be an *Olcella* species (not *Notaulacella*, which has long ocellar bristles).

It is remarkable that two sympatric *Pseudomyrmex* species seem to be associated with apparently specialized nectar-robbing chloropids in two different genera, and perhaps even more remarkable that these conspicuous associations remained unreported until 2018. Other *Olcella* are common kleptoparasites associated with large predators and I've photographed them on ants as well as other insects as they are being consumed by spiders, reduviids and asilids, but never on extrafloral nectaries and never in association with living ants. Unfortunately, my collecting and export permits did not cover Guanacaste, so these opportunistic observations are not vouchered. Specimens are needed to identify the fly species and to further investigate chloropid-ant associations in other *Pseudomyrmex*-*Vachellia* communities.

Acknowledgements

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References

Barrantes G, Valverde-Hernández J.C, Vargas-Rodríguez A, Amador-Vargas S. 2018. Another one breaking through an ant plant mutualism. *Boletín de la Sociedad Zoológica del Uruguay* (2a época). 2018. Vol. 27 (1): 11–13

Gonçalves Ana Rita. 2018. Observations on how some very interesting flies benefit from ant behaviour to rob their resources. *Fly Times* 63: 13–14.

Figure 1. *Pseudomyrmex flavicornis* with four chloropids.

Figure 2. *Pseudomyrmex flavicornis* at an extrafloral nectary, with three chloropids

Figure 3. An *Olcella* sp. feeding at an extrafloral nectary provided by *Vachellia* for its *Pseudomyrmex flavicornis* associate.

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Untangling tangle-veined flies (Nemestrinidae)

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Many dipterists, pollination biologists and keen naturalists may be aware of the handful of Nemestrinidae species in southern Africa with wildly long proboscides that have had their adult ecology rather well studied. They are mesmerising creatures, visiting long tubed, showy flower after flower (Fig. 1) (Manning & Goldblatt, 2000). In southern Africa, as a family, the Nemestrinidae likely visit several hundreds of flower species and a handful of long-proboscid species are known to be the sole pollinators of ~150 plant species (Manning & Goldblatt, 2000; Potgieter & Edwards, 2005; Anderson & Johnson, 2009; Newman, Manning, & Anderson, 2014). Their role as important pollinators of numerous plants, including rare and endangered species makes them a group of particular interest to study. Unfortunately, the southern African genera have not received much taxonomic attention in recent years and little to no molecular data is available for the family with which to work.

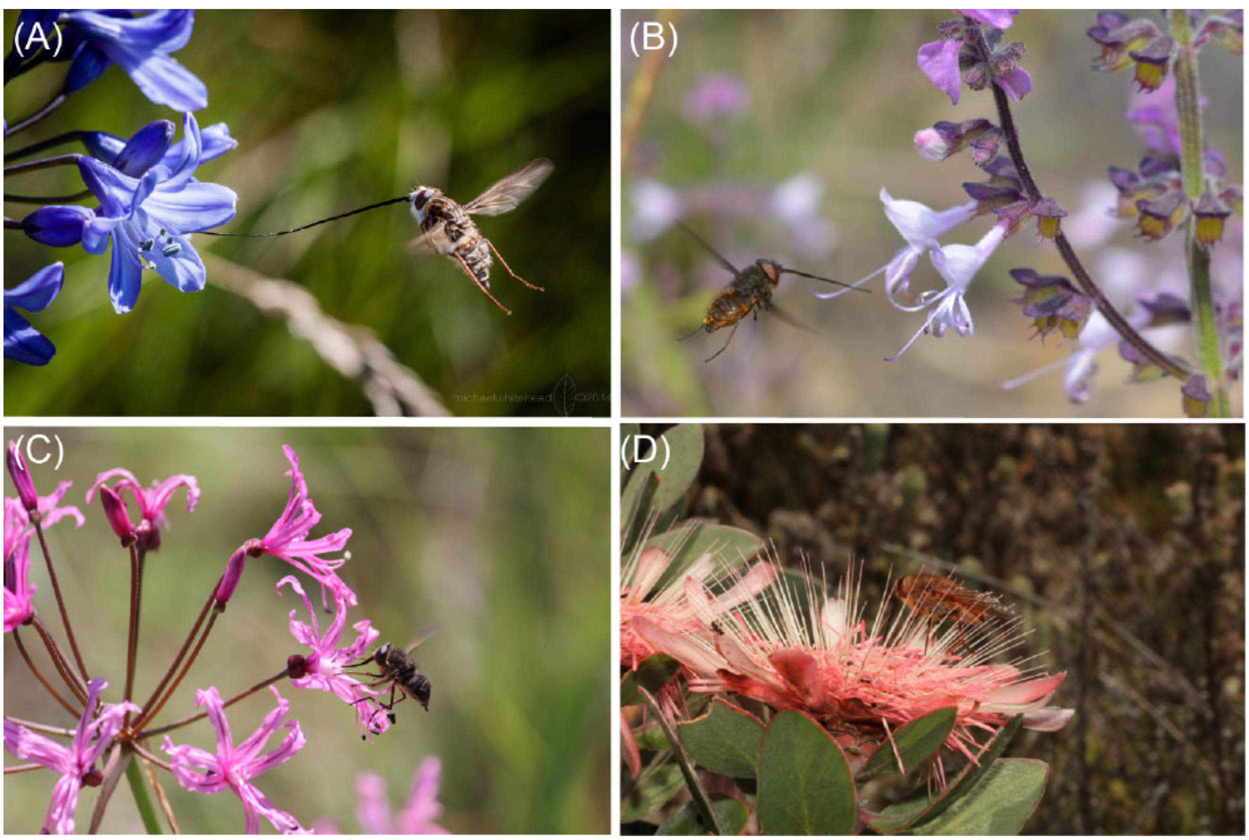

Figure 1. Photographs of *in situ* adult *Prosoeca* species visiting flowers. (**A**) *Prosoeca ganglbaueri* visiting an *Agapanthus sp.* (**B**) *Prosoeca* sp. visiting a Lamiaceae sp. (**C**) *Prosoeca umbrosa* visiting *Nerine angustifolia* (**D**) *Prosoeca robusta* visiting a *Protea punctata.* Photo credits: (A) Michael Whitehead, (B) Ruth Cozien, (C) Genevieve Theron, (D) Steven Johnson.

During my PhD I was fortunate enough to travel across South Africa, spending many hours in the field watching, admiring, and collecting these flies. I managed to include 58 morphospecies in a phylogenetic tree, using four gene regions of the three southern African Nemestrininae genera. The topology of this tree suggests that the southern African genera are in urgent need of a review, with *Prosoeca* emerging as paraphyletic, with *Stenobasipteron* nested within *Prosoeca.* Furthermore, at least half of the species that we included in our tree are currently undescribed, highlighting a substantial taxonomic impediment in this group (Theron, 2021).

As part of my thesis, we investigated the *Prosoeca peringueyi* complex that was thought to be the sole pollinators for over 28 flower species. Upon further investigation, the number of flowers pollinated by the long-proboscid flies in this system has increased to 42 and we have described a new nemestrinid species, *Prosoeca torquata* (Fig. 2) (Theron *et al.*, 2020). The new species was found to be sister to *P. peringueyi* but morphologically and genetically distinct from it. The description of *P. torquata* along with that of a third species in this system, *Prosoeca marinusi* (Barraclough *et al.*, 2018), complicates the once simple pollination story, making our knowledge of these plant-fly interactions ambiguous. In addition to the undescribed species that await description, there are a few species complexes in *Prosoeca* that remain to be resolved.

Figure 2. Photographs of *in situ* adult *Prosoeca peringueyi* visiting *Zaluzianskya* sp. (**A**) and visiting *Lapeirousia silenoides* (**B**), and *Prosoeca torquata* hovering over *Lapeirousia dolomitica* (**C**) and mating (**D**). Photo credits: (A-B) Steven Johnson, (C-D) Florent Grenier.

Recently I have started a new position as a postdoctoral fellow at the KwaZulu-Natal Museum (NMSA) with Dr. John Midgley. My aim, amongst others, is to establish a stable taxonomy for the southern African genera and, in collaboration with Dr. David Yeates, to reconstruct a phylogeny of the Nemestrinidae family. South Africa has six of the 15 genera worldwide, and I already have samples of these groups preserved in ethanol. A family level phylogeny will allow me to investigate Bernardi's (1973) hypothesis of generic relationships based on morphology as well as to examine evolutionary history and biogeographical patterns.

Call for newly collected Nemestrinidae

To reconstruct a phylogeny of the Nemestrinidae we need representative specimens from as many genera as possible. To test the monophyly of the genera it would be ideal to have 2-3 species from each genus if they are available. We plan to build a phylogenomic dataset using Anchored Hybrid Enrichment to generate sequence data. While our coverage in southern Africa is good, we have very few representatives from elsewhere in the world. We are seeking donations of specimens for the project, preferably in ethanol or freshly pinned, and would welcome any suggestions about the times and places to collect the different genera around the world. If you have an interest in the Nemestrinidae, like I do, I am open to discussing potential collaborations. For more information, feel free to contact me.

References

Anderson B. & Johnson S.D. 2009. Geographical covariation and local convergence of flower depth in a guild of fly-pollinated plants. *New Phytologist* 182: 533–540.

Barraclough D.A., Colville J.F., Karolyi F. & Krenn H.W. 2018. A striking new species of *Prosoeca* Schiner, 1867 (Diptera: Nemestrinidae): An important pollinator from the Bokkeveld Plateau, Northern Cape Province, South Africa. *Zootaxa* 4497: 411–421.

Bernardi N. 1973. The genera of the family Nemestrinidae (Diptera: Brachycera). *Arquivos de Zoologia* 24: 211–318.

Manning J.C. & Goldblatt P. 2000. The long-proboscid fly pollination system in southern Africa. *Annals of the Missouri Botanical Garden* 87: 146–170.

Newman E., Manning J.C. & Anderson B. 2014. Matching floral and pollinator traits through guild convergence and pollinator ecotype formation. *Annals of Botany* 113: 373–84.

Potgieter C.J. & Edwards T.J. 2005. The *Stenobasipteron wiedmanni* (Diptera, Nemestrinidae) pollination guild in eastern Southern Africa. *Annals of the Missouri Botanical Garden* 92: 254–267.

Theron G.L., Grenier F., Anderson B.C., Ellis A.G., Johnson S.D., Midgley J.M. & van der Niet T. 2020. Key long-proboscid fly pollinator overlooked: morphological and molecular analyses reveal a new *Prosoeca* (Nemestrinidae) species. *Biological Journal of the Linnean Society* 131: 1–13.

Theron G.L. 2021. *Evolution of tangle-veined flies: systematics, biogeography and functional traits in southern African Nemestrininae (Nemestrinidae)*. Doctoral Thesis. University of KwaZulu-Natal, Pietermaritzburg.

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A multiple species aggregation of *Archisepsis* (Sepsidae) flies in Costa Rica

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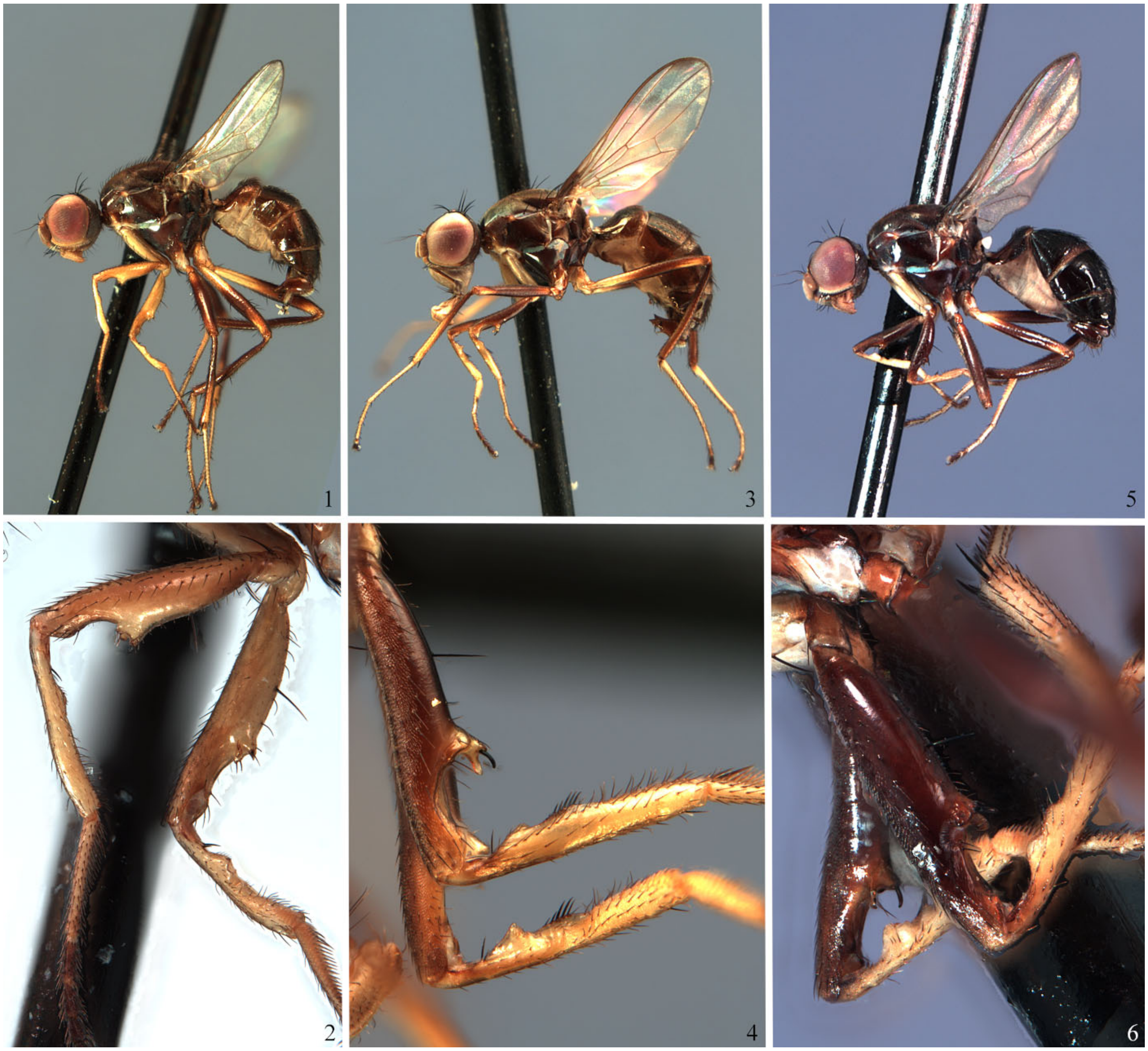
Black scavenger flies (Sepsidae) are well-known for their unusual mating systems and aggregations (Pont 1979, 1987; Eberhard 2000). While collecting in Heredia, Costa Rica near Trinidad, on 15.viii.1995 in a 50m elevation forest at the junction of the Rio Sarapiqui and Rio San Juan, the first author came across a relatively small dense aggregation of male and female sepsids on understory plants. The flies were aggregating on eight leaves of three adjacent plants, mostly on top of the leaves, with many observed wagging their wings and occasionally (presumably males) grappling. No copulations or riding of males on females was observed. No odour or scent was noticeable as has been reported in some other sepsid aggregations (Pont 1987). This was the only aggregation seen by the first author or other dipterists (e.g., Monty Wood) who were collecting flies at this location. The flies were not observed feeding on any liquids or other substances on the surface of the leaves. In addition, the aggregation site was not near dung or other decaying organic matter anywhere in the vicinity, which the flies might have been feeding or ovipositing on. After observing the aggregation for several minutes the first author swept the understory plants and collected the entire, or almost the entire, aggregation of flies, totalling 135 specimens comprising 80 males and 55 females.

The second author later identified the flies as *Archisepsis* Silva, with specimens surprisingly belonging to three separate species, namely *A. polychaeta* (Ozerov), *A. diversiformis* (Ozerov) and *A. excavata* (Duda) (Figs 1, 3, 5). Males of these species possess species-specific modifications of the fore legs (Figs 2, 4, 6) that are used to clasp the base of the female wing during mounting to stimulate conspecific females prior to copulation (Eberhard 2001, 2002). Most belonged to *A. polychaeta* (65 males and 49 females), whereas only 11 males of *A. excavata* and 4 males of *A. diversiformis* were present. Six additional females of *A. excavata* and/or *A. diversiformis* were collected, but could not be confidently assigned to either of these two species. These three species belong to a relatively large subset of the genus characterized by an abdomen that is transversely wrinkled dorsally in both sexes, and while black in base colour, is strongly iridescent. This form of wrinkled iridescent abdomen is unique within the Sepsidae and may possibly involve signalling between the sexes.

Initially the first author thought he was observing a lek of a single sepsid species, however no mounting of males on females, or copulations, were seen during the relatively short time (5–10 minutes) the aggregation was observed. Generally mating in sepsids occurs near oviposition sites such as dung or carrion (Pont 1979), but sexual activity can occasionally occur in some sepsid species in areas not tightly associated with oviposition sites. Eberhard (2000) discovered that small numbers of virgin females of *Microsepsis armillata* (Melander & Spuler) in Costa Rica copulated with males away from oviposition sites, whereas nonvirgin females commonly mated on dung with males, after laying their eggs. In addition, large aggregations of up to 100,000 individuals of *Sepsis fulgens* Meigen are known in England away from oviposition sites (Pont 1987), where most aggregating flies do not feed or engage in sexual interactions, even though rare matings may occasionally occur. Pont (1987) suggested that these huge aggregations, which usually form in this species during late summer and early fall, are actually hibernation swarms. This presumably is not the cause of the *Archisepsis* aggregation observed in Costa Rica. Why the *Archisepsis* flies observed here were aggregating, what precisely they were doing, and why three species of *Archisepsis* were found together, is still unclear.



Figures 1-6: *Archisepsis* males collected at study site: **1.** *A. polychaeta* (Ozerov); **2.** same, detail of fore legs; **3.** *A. diversiformis* (Ozerov); **4.** same, detail of fore legs; **5.** *A. excavata* (Duda); **6.** same, detail of fore legs.

Acknowledgement
Dr. Vera Silva (São Paulo, Brazil) is thanked for clarifying and confirming the identities of the species collected.

References

Eberhard, W.G. 2000. Behavior and reproductive status of *Microsepsis armillata* (Diptera: Sepsidae) flies away from oviposition sites. *Annals of the Entomological Society of America* 93(4): 966–971.

Eberhard, W.G. 2001. The functional morphology of species-specific clasping structures on the front legs of male *Archisepsis* and *Palaeosepsis* flies (Diptera, Sepsidae). *Zoological Journal of the Linnean Society* 133: 335–368.

Eberhard, W.G. 2002. Physical restraint or stimulation? The function(s) of the modified front legs of male *Archisepsis diversiformis* (Diptera, Sepsidae). *Journal of Insect Behavior* 15(6): 831–850.

Pont, A.C. 1979. Sepsidae. Diptera Cyclorrhapha, Acalyptrata. *Handbook for the Identification of British Insects* 10,5(c): 35 pp., 10 figs, 10 pls.

Pont, A.C. 1987. 'The mysterious swarms of sepsid flies': an enigma solved? *Journal of Natural History* 21(2): 305–317.

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Aquatic Empididae inhabiting tufa stream environments of tropical karst ecosystems in Thailand

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In a previous issue of *Fly Times*, I presented (Plant, 2019a) a short introduction to my ongoing project investigating the aquatic Empididae inhabiting tropical tufa-streams and other calcareous waters in Thailand. Thanks to generous support from the Thailand Research Fund (DBG6180024), I was able to contract Chonticha Kunprom (then a postdoctoral researcher in Pairot Pramual's lab at Mahasarakham), to extract and amplify DNA, and my wife Pia, as a driver, technician and "fixer." Over a two-year period, Pia and I intermittently travelled the length and breadth of Thailand, comprehensively exploring and sampling all of the karstic areas. A book could be written on the spectacular places visited, adventures had, encounters with people and wildlife etc; - but here we will be confined merely to the results!

There are no cold-water relicts

First mention is of a negative result. I had hypothesised that cold-adapted aquatic Empididae may have been marooned during cooler climatic periods associated with historical glacial maxima but might persist to this day in cold emergent groundwaters. In fact, cold springs could not be found, and it seems that limestone streams in Thailand's karsts are probably mostly allogenic with short subsurface residence times, so water temperatures essentially equilibrate with atmospheric temperatures at different elevations, not with deep geology. Emergent streams and springs may have once held cold-adapted empidids, but they were likely lost rapidly as the climate ameliorated.

Diversity, distribution & ecology

So, all calcareous lotic waters in Thailand are warm (typically 18–30°C) and these were found to support empidoid communities dominated by species of *Hemerodromia* Meigen (Clinocerinae and other Hemerodromiinae such as *Chelifera* Macquart were rare).

I had earlier described 20 new species of *Hemerodromia* from Thailand and recorded 5 species previously known only from China. The new study described 5 more species and greatly expanded knowledge of 11 previously known species. Six species had an obligate association with tufa, two were restricted to calcareous streams, three occurred on both tufa and calcareous streams and six were eurytopic species also found away from karst. A striking characteristic of the six obligate tufa species was an *absolute* association with rapid shallow flows of water over tufa at waterfalls, always in shaded locations. Ordination of environmental variables revealed that benthic substrate and canopy cover were very important factors for most calcareous species. As might be expected, species richness and abundance varied through the monsoon cycle (which produces seasonal rainfall extremes, spate, and drought conditions etc.) but there were few simple correlations with seasonal changes in water temperature, conductivity and pH. The seasonality of tropical stream insects is not particularly well understood and further analysis of these *Hemerodromia* communities is in progress.

Populations (especially of tufa-species) were generally highly localised and confined to widely dispersed, fragmented and rare habitats. Local and regional-scale endemism was apparent at species-level. Thus, there were species with patchy distribution patterns that were confined to northern, western, southern regions corresponding with some of the reasonably well-defined biogeographic boundaries now known to shape diversity of Empidoidea in Thailand. However, assemblage similarity in tufa habitats decayed with geographical distance and there was little evidence for community-level endemism at any geographic scale. Both species makeup and community characteristics appeared to be independent of underlying limestone geology (Carboniferous, Jurassic, Cretaceous etc.). Any limestone will do.

Genetic structure and population history

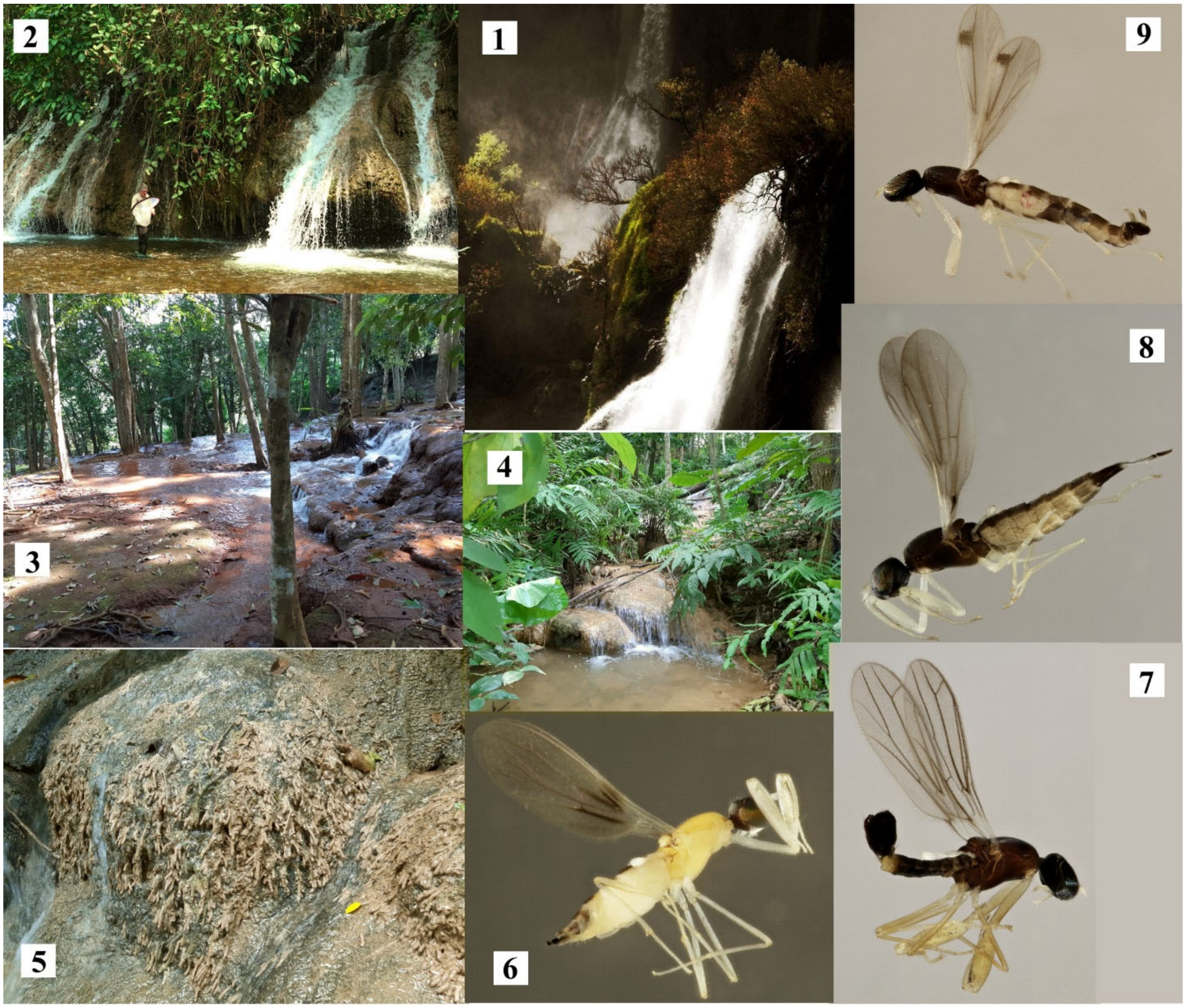
The population genetic structure of obligate tufa species that have fragmented distributions is of great interest and this was studied in detail for one exemplar species (*H. conspecta* Plant). A median-joining (MJ) haplotype network based on COI sequences indicated a high level of genetic structure and resolved 5 distinct lineages associated with geographically isolated clusters. Haplotype diversity, nucleotide diversity and pairwise comparisons of F_{ST} indicated considerable genetic differentiation among fragmented populations. However, none of the lineages were so differentiated as to be considered discrete species (as assessed by evolutionary distance using the Kimura 2-parameter). Rather, the evidence pointed to ongoing processes of allopatric speciation. Long path lengths in the MJ network suggest long isolation of communities and also, the very low haplotype diversity of one population points to it having experienced a severe population bottleneck (as is common in species with specialised ecology that ties them to rare, fragmented and unstable habitats). Mismatch distribution analysis suggested a late Pleistocene population expansion (~10,000 – 100,000 bp) of *H conspecta* populations, entirely consistent with evidence for a climatically driven period tufa deposition in the late Cenozoic which was followed by late Quaternary decline.

The allopatric distribution of geographically fragmented and genetically distinct lineages supports the view that vicariant diversification is driving active processes of speciation and microendemism occurring within a mosaic of multiple microrefugia set within a wider matrix of unsuitable habitats. Tufa-inhabiting *Hemerodromia* in Thailand are actively evolving in their own tiny world-fragments.

Conservation & Threats

Extinction risk is high in species with restricted ranges. Isolation and limited dispersal capabilities of karst species render them inherently vulnerable to environmental changes. Even within the scattered 'archipelagos' of karst landscapes, *active* tufa is a rare, highly fragmented and diminishing habitat viewed at evolutionary, spatial and temporal scales. Low-vagility species adapted to it can only survive by dynamically niche-tracking transient tufa habitats as they are modified or migrate under the predominant influence of fluvial processes such as riverine incisions, intermittent drying or water loss into sinkholes and fractured stream beds, changes to spring lines, downslope progradation or lateral migration of cascades etc. not to mention large-scale slippage and tectonic events. Interestingly, while in most karst habitats there is net removal of rock due to hydrological *dissolution* by meteoric water, with carbonates weathered from the rock being hydrologically transported away, tufa habitats represent a *depositional* zone where new rock is accreted. Tufa stenotopes must be able to avoid being trapped in the rapid deposition process (tufa formation can be astonishingly rapid in the tropics).

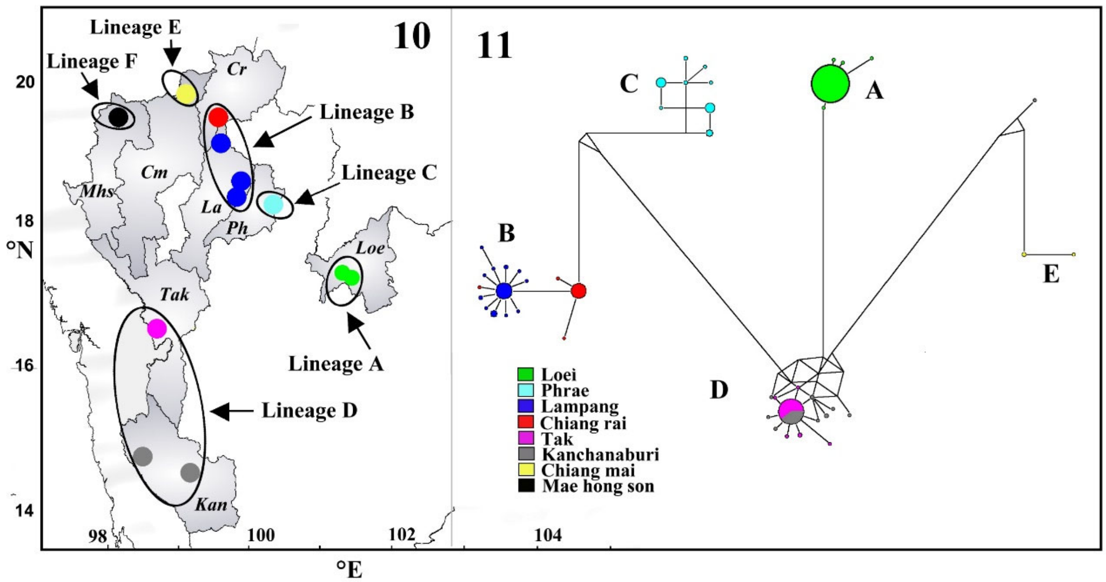
These already fragile tufa biotopes are particularly susceptible to activities of humans. Tourism, agricultural activities and water abstraction are pervasive at tufa sites with obvious and sometimes massively damaging effects including, for example: (1) physical destruction and polishing of tufa by both people and domestic animals, (2) management practices promoting vegetation removal, (3) over-abstraction, complete abstraction and capping at source leading to desiccated natural channels and "dead" tufa, or (4) siltation and contamination with agricultural run-off. Many sites have been destroyed or severely damaged centuries ago by incorporation into Buddhist temples and shrines etc. (although a few such sites have been well looked after). The special "magical" properties of tufa



Figures 1–9. **1**. Thi Lo Su; a 400m high tufa waterfall and terrace system in Tak Province. **2**. A small sidestream discharging via a low tufa waterfall into a large calcareous river at SuSa, Mae Hong Son Province. **3**. A huge tufa cone at Pa Wai, Tak Province is engulfing all in its path. The herb and shrub layers have been removed by human agency. The red colour is soil arising from agricultural runoff and is completely smothering the tufa system. There were no aquatic empidids at this site and very few aquatic insects in general. **4**. A healthy tufa stream at Paeng Din, Loei Province, supporting several specialist *Hemerodromia* spp. **5**. Close view of "fast films" flowing over tufa surfaces in shade; the habitat of all tufa specialists. The tubular structures are silk capture nets of philopotamid Trichoptera that have provided nucleation sites for calcite precipitation. Insect silks (including those of Diptera) contribute significantly to mineralisation and the accretion of new tufa. Insects are involved in growing rocks! A rarely seen biotic role in geology. **6**. *H. namtokhinpoon* Plant, a tufa stenotope confined to just two sites, only 800m apart in Loei province. **7**. *H. demissa* Plant is only found on larger calcareous rivers such as that shown in Fig. 2. **8**. *H. conspecta* Plant, is a tufa specialist that was used as an exemplar in population genetic studies. **9**. *H. anomala* Plant is another tufa stenotope. It has unusual morphology of head, thorax and wing venation but preliminary CO1 sequence date (San Namtaku, unpublished) does not suggest a unique phylogeny.

streams, upwellings and cave emergences have long been venerated. In a tradition stretching back thousands of years (and still alive today) a Buddhist monk might seek out such a place for quiet meditation. Of course, the locals inevitably find the monk, provide alms, build him shelters, install Buddha images, entrain the spring etc. and in no time, the original place for quiet contemplation has developed into a temple complex with access roads, car parks and many more monks. Sometimes, nothing remains of a tufa spring but its presence recorded in the name of the temple! It is fortunate that unlike in Europe (where tufa deposits have long been over-exploited, e.g. in the construction of the Roman Colosseum), I could find no evidence of tufa being used in monumental architecture at any of the many temples visited during fieldwork in Thailand.

The next steps? Well tufa is found throughout tropical SE Asia. New species discovery is highly likely in many areas of Myanmar, Vietnam and Lao PDR for example, so sampling visits to these areas would be productive. Perhaps, the results of this work can help educate improved conservation in a country where waterfall tourism is a major industry (some tufa systems receive >700,000 visitors per year… or at least they did, prior to Covid-19). One hopes so.



Figures 10–11. *Hemerodromia conspecta* Plant. **10**. Distribution in northern Thailand. Localities where the species occurs are colour-coded and their unique lineages designated in agreement with Fig. 11. **11**. A median-joining (MJ) haplotype network based on COI sequences indicating the major lineages.

References

Kunprom, C., Plant, A.R. & Pramual, P. (2021). Population genetic structure and microendemism in aquatic Empididae (Diptera) in transient tufa biotopes on tropical karst. *Entomological Science*. 24(2): 127–136.

Plant, A.R. (2019) Biomediation of tufa formation by silk capture nets of larval Philopotamidae (Insecta: Trichoptera). *Tropical Natural History* 19(2): 113–116.

Plant, A.R. (2019a). Inventory of Aquatic Hemerodromiinae and Clinocerinae (Diptera: Empididae) inhabiting Tufa Stream Environments of Tropical Karst Ecosystems in Thailand*. Fly Times* 62: 5–7.

Plant, A.R. (2020) New species of *Hemerodromia* Meigen (Diptera: Empididae: Hemerodromiinae) associated with limestone karstic waters in Thailand. *Zootaxa*.4758 (3): 549–560.

Plant, A.R. (2021). Tropical tufa and karsts streams support unique and threatened assemblages of aquatic Diptera, Empididae in Thailand. *Entomological Science*. 24: 169–182.

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Why did *Culex bahamensis* replace *Aedes taeniorhynchus* (Culicidae) on No Name Key, Monroe County, Florida, in 2007?

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The Florida Keys Mosquito Control District has been monitoring species composition, seasonal distribution, and relative abundance of mosquito species since 1998 (Hribar 2002a). Adult mosquitoes are collected with the aid of dry ice-baited light traps. *Aedes taeniorhynchus* (Wiedemann) is by far the most abundant mosquito in the Florida Keys, and the most widespread. Some years ago, when I was examining data from collections made in previous years, I noticed what appeared to be an anomalous inversion of the relative numbers of *Ae. taeniorhynchus* and *Culex bahamensis* Dyar & Knab on No Name Key (Fig. 1). This inversion did not occur on neighboring Big Pine Key, although there were more *Cx. bahamensis* than usual (Fig. 2).

Curious as to why this might have happened, and knowing that *Cx, bahamensis* numbers are affected by tides and rainfall (Hribar 2002b), I examined tide data from the closest site where actual measurements are made, Coast Guard Station Marathon. There were apparently normal tidal fluctuations during the period 2006–2008, although there did appear to be some lower tides at the end of 2007 and the beginning of 2008 (Fig. 3). More interesting, however, was an apparent inverse relationship between rainfall and *Cx. bahamensis* numbers (Fig. 4).

There was an interesting interplay among the four most commonly collected mosquito species on No Name Key, *Ae. taeniorhynchus*, *Deinocerites cancer* Theobald, *Anopheles atropos* Dyar & Knab, and *Cx. bahamensis* (Fig. 5). Figure 5 also reveals that the increase in numbers of *Cx. bahamensis* extended into 2008. All four species have different habitat requirements, although they occur in the same area, but it was only *Cx. bahamensis* that made a dramatic replacement of *Ae. taeniorhynchus*.

Not only was there below normal rainfall in 2007, but the years 2007–2008 were La Niña years (Okumura & Deser 2010). La Niña years are characterized in part by lower rainfall and less surface water (Schmidt & Luther 2002, Beckage et al. 2003, Abtew & Trimble 2010). Lower rainfall volume and less surface water, conditions present during a drought situation, reduce the number of oviposition sites available to mosquitoes, for example *Culex nigripalpus* Theobald (Shaman et al. 2002). Lower rainfall, especially in La Niña years, results in greater water salinity (Schmidt & Luther 2002).

Investigation of water quality requirements of the two species (*Ae. taeniorhynchus* and *Cx. bahamensis*) proved to be intriguing. I had reported some water quality parameters for *Cx. bahamensis* larvae on Vaca Key (Hribar 2010), and Van Der Kuyp (1954) also reported pH values for *Cx. bahamensis* larval habitats (Table 1). Several investigators had reported water quality parameters for *Ae. taeniorhynchus* (Table 2). Comparison of the two species' data revealed that *Cx. bahamensis* apparently can survive much higher seawater concentrations than can *Ae. taeniorhynchus* (Table 3). Since rainfall was less than usual in 2007, but tides were normal, it is possible that oviposition sites usually utilized by *Ae. taeniorhynchus* became too saline due to repeated inundation by tidal waters but no dilution of water by rainfall (DeSantis et al. 2007). If this process continued throughout the year the salinity of the oviposition sites may have increased to the point that the sites became more favorable to *Cx. bahamensis*.

Table 1. Water quality data for two *Culex bahamensis* larval habitats on Vaca Key, Florida (data from Hribar 2010).
[1]Van Der Kuyp (1954) reported pH values of 7.2 - 8.8 for *Culex bahamensis* larval habitats.

	Sample	
Parameter	Fountain	Pond
pH[1]	8.8	8.0
Ammonia	0	0
Hardness	50	120
Nitrate	0	0
Nitrite	0.5	0
Alkalinity	>300	80

Table 2. Water quality data for *Aedes taeniorhynchus* larval habitats.
[1]1, Pierce et al. (1945); 2, Van Der Kuyp (1954); 3, Peterson and Chapman (1970); 4, Carlson (1982); 5, Clark et al. (2004).

	Study[1]				
Parameter	1	2	3	4	5
pH[1]	8.1 – 9.6	7.4 – 8.0	3.3 – 8.1	6.8 – 7.4	3 – 11
Ammonia				0 – 27.1	
Nitrate				0 – 16.7	
Nitrite				0 – 2.1	

Table 3. Comparison of larval habitat water quality data for *Aedes taeniorhynchus* and *Culex bahamensis*.
[1] Data from Nayar (1969), Nayar & Sauerman (1970). [2] Data from Van Der Kuyp (1954). [3] SW = seawater.

	Species	
Parameter	*Ae. taeniorhynchus*	*Cx. bahamensis*
pH	3 – 11	8 – 8.8
Ammonia	0 – 27.1	0
Nitrate	0 – 46.7	0
Nitrite	0 – 2.1	0 – 0.5
Salinity[1]	10% – 25% SW[3]	50% SW
mg Cl/L[2]	150 – 24,000	150 – 46,000

Aedes taeniorhynchus is a mosquito that engages in long-distance dispersal flights. There is ample larval habitat on No Name Key, but No Name Key also receives some mosquitoes dispersing from small uninhabited islands such as Annette Key and Porpoise Key (Vlach et al. 2006). Examination of mosquito trapping data from Annette Key and comparison to No Name Key revealed that during the years 2004–2009 *Ae. taeniorhynchus* and *Cx. bahamensis* population levels essentially mirrored each other on the two islands (Figs 6 & 7). The concordant rise and fall of mosquito numbers of Annette Key and No Name Key, and the increase in numbers on Big Pine Key, indicate that this phenomenon, the surge in numbers of *Cx. bahamensis*, was not confined to one island. Something changed to facilitate increased reproduction by *Cx. bahamensis*, and that increase is coincident with 2007 and 2008 being La Niña years.

The El Niño / La Niña phenomena affect weather patterns in subtropical areas worldwide (Molles & Dahm 1990). There is ample evidence that these weather phenomena and rainfall patterns associated with them impact insect abundance (e.g., Frankie et al. 2005, Seal & Tshinkel 2010, Woli 2014, Szyniszewska et al. 2020). These events also influence mosquito numbers and life cycles and can have definite impact on disease transmission by mosquitoes (Reisen et al. 2008, Miley et al. 2020).

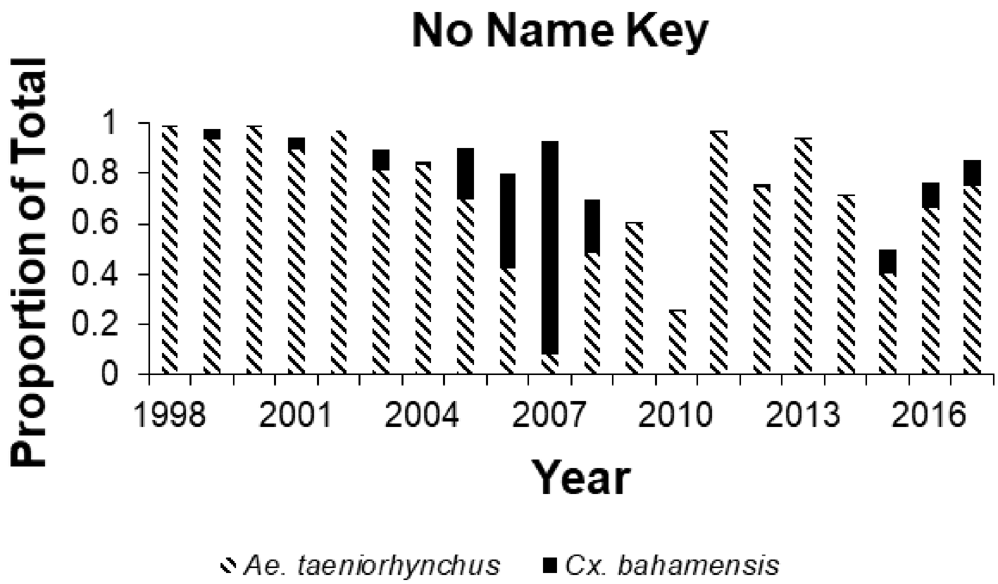

Figure 1. *Aedes taeniorhynchus* and *Culex bahamensis* as proportions of total mosquitoes collected, No Name Key, 1998–2017.

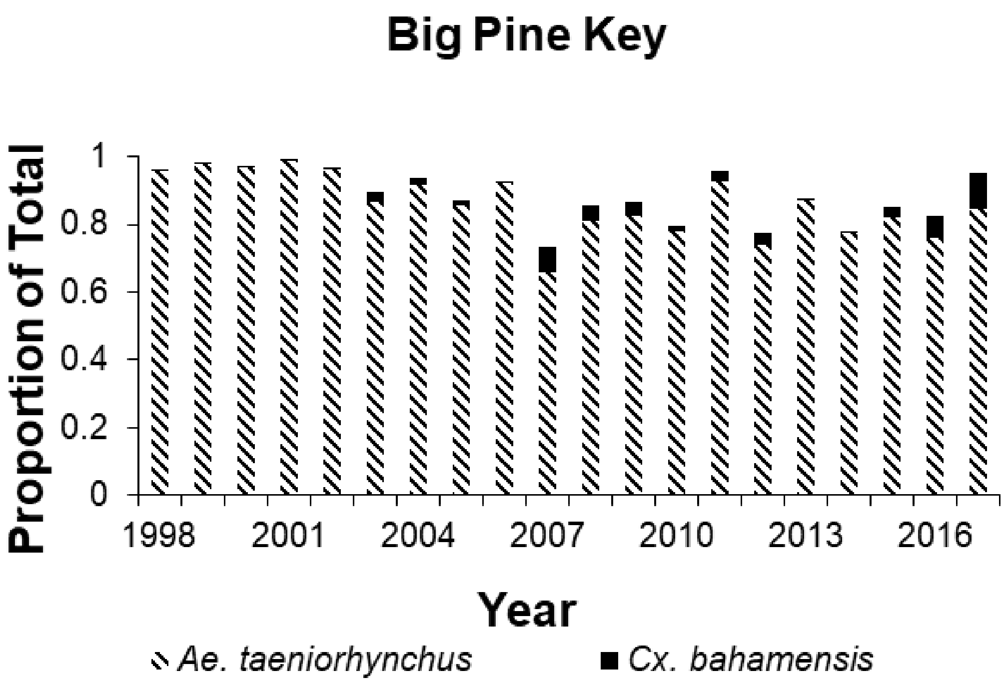

Figure 2. *Aedes taeniorhynchus* and *Culex bahamensis* as proportions of total mosquitoes collected, Big Pine Key, 1998–2017.

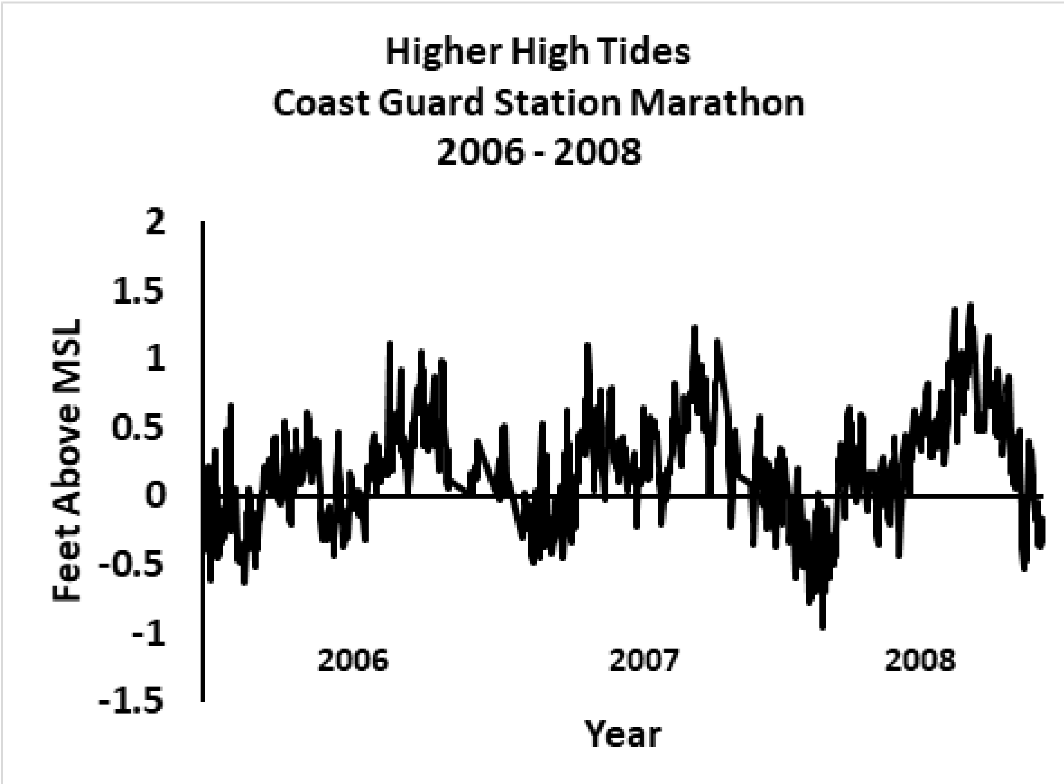

Figure 3. Heights of higher high tides above or below mean sea level, 2006-2008.

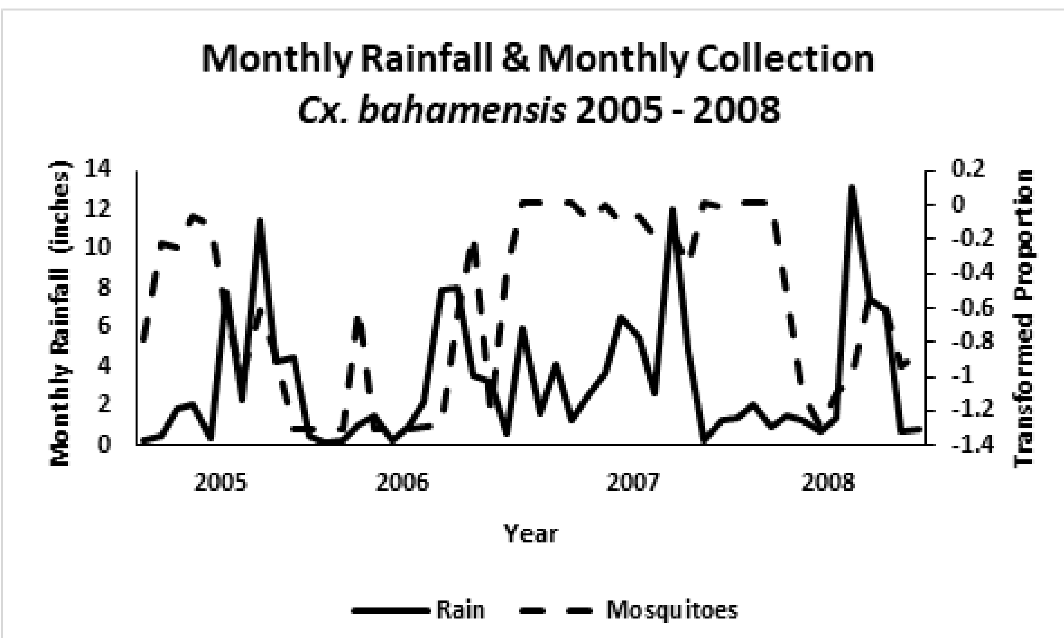

Figure 4. Relationship between rainfall and numbers of *Cx. bahamensis*, No Name Key, 2005–2008.

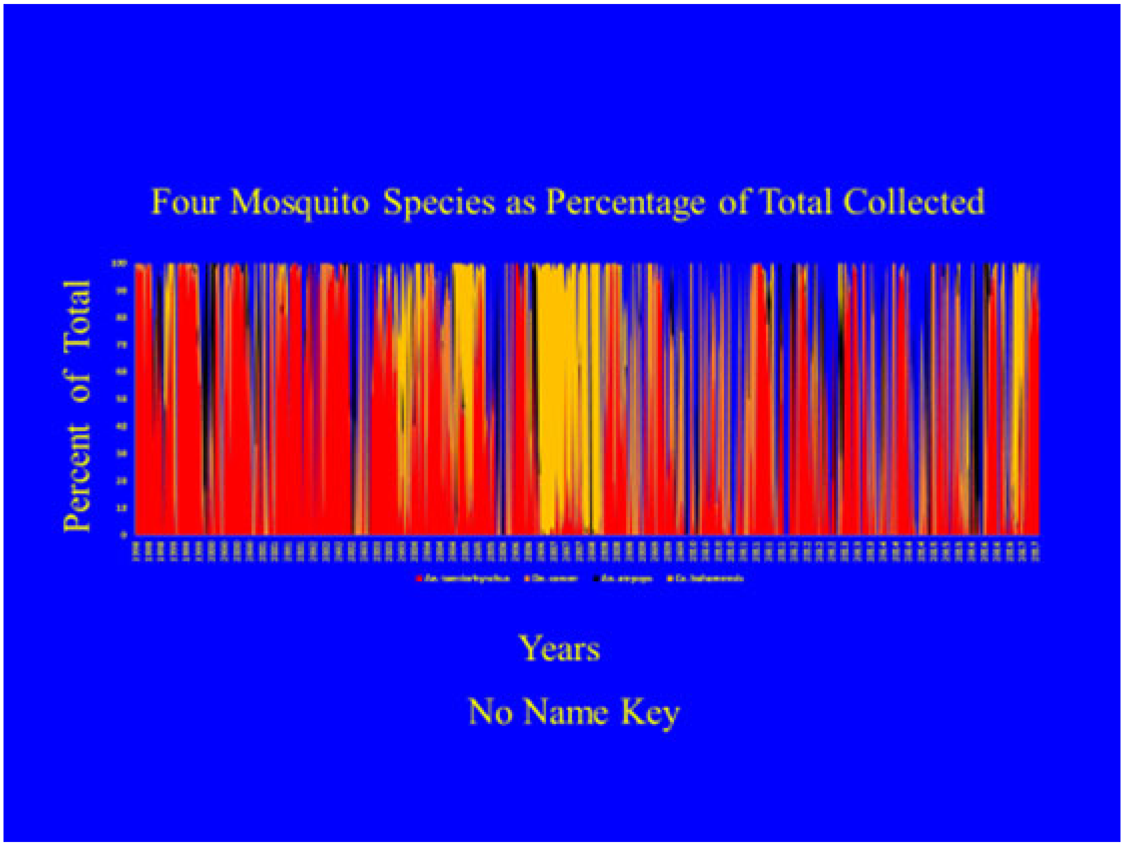

Figure 5. Four most commonly collected mosquito species on No Name Key as a proportion of total mosquitoes collected. Red = *Ae. taeniorhynchus*, orange = *Deinocerites cancer*, black = *Anopheles atropos,* yellow = *Cx. bahamensis*.

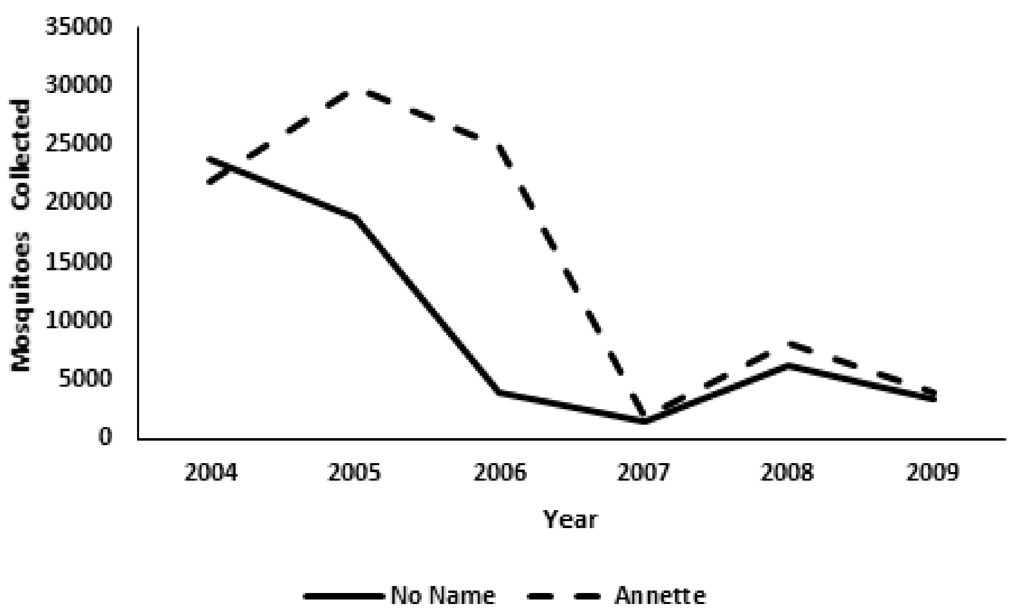

Figure 6. Population fluctuations of *Ae. taeniorhynchus* on No Name and Annette Keys, 2004–2009.

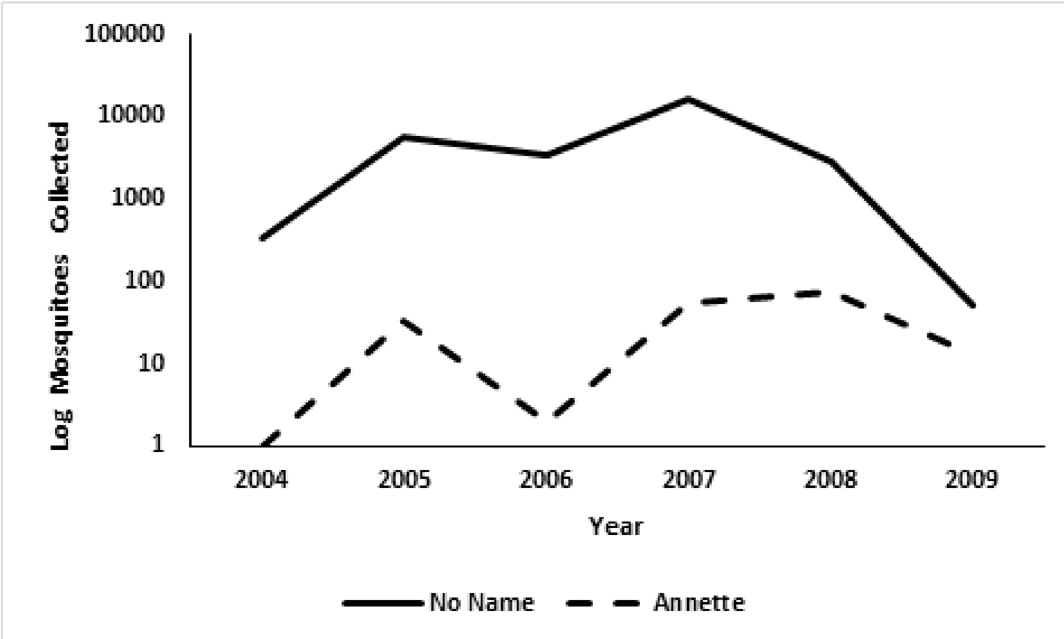

Figure 7. Population fluctuations of Cx. *bahamensis* on No Name and Annette Keys, 2004–2009.

References

Abtew, W. & P. Trimble. 2010. El Niño–Southern Oscillation link to South Florida hydrology and water management applications. *Water Resources Management* 24: 4255–4271.

Beckage, B., W. Platt, M. Slocum, & B. Panko. 2003. Influence of the El Niño Southern Oscillation on fire regimes in the Florida Everglades. *Ecology* 84: 3124–3130.

Carlson, D.B. 1982. Mosquitoes associated with evaporation-percolation ponds in Indian River County, Florida. *Mosquito News* 42: 244–250.

Clark, T.M., B.J. Flis, & S.K. Remold. 2004. pH tolerances and regulatory abilities of freshwater and euryhaline Aedine mosquito larvae. *Journal of Experimental Biology* 207: 2297–2304.

DeSantis, L., F.E. Putz, & K. Williams. 2007. Sea-level rise and drought interactions accelerate forest decline on the Gulf Coast of Florida, USA. *Global Change Biology* 13: 2349–2360.

Frankie, G.W., M. Rizzardi, S.B Vinson, T.I. Griswold, & P. Ronchi. 2005. Changing bee composition and frequency on a flowering legume, *Andira inermis* (Wright) Kunth ex DC. during El Nino and La Nina years (1997–1999) in northwestern Costa Rica. *Journal of the Kansas Entomological Society* 78: 100–117.

Hribar, L.J. 2002a. Mosquito (Diptera: Culicidae) collections in the Florida Keys, Monroe County, Florida, USA. *Studia Dipterologica* 9: 679–691.

Hribar, L.J. 2002b. Relation of *Culex bahamensis* Dyar & Knab, 1906 (Diptera: Culicidae) to rainfall and tides on Grassy Key, Florida, USA. The Annals of Medical Entomology, 11: 11-16.

Hribar, L.J. 2010. Water quality of two larval habitats of *Culex bahamensis* in the Florida Keys. *Notulae Naturae* 483: 1–4.

Miley, K.M., J. Downs, S.P. Beeman, & T.R. Unnasch. 2020. Impact of the Southern Oscillation Index, temperature, and precipitation on Eastern equine encephalitis virus activity in Florida. *Journal of Medical Entomology* 57: 1604–1613.

Molles Jr, M.C. & C.N. Dahm. 1990. A perspective on El Nino and La Nina: global implications for stream ecology. *Journal of the North American Benthological Society* 9: 68–76.

Nayar, J.K. 1969. Effects of larval and pupal environmental factors on biological status of adults at emergence in *Aedes taeniorhynchus* (Wiedemann). *Bulletin of Entomological Research* 58: 811–827.

Nayar, J.K. & Sauerman Jr, M., 1970. A comparative study of growth and development in Florida mosquitoes: Part 2: Effects of larval nurture on adult characteristics at emergence. *Journal of Medical Entomology* 7: 235–241.

Okumura, Y.M. & C. Deser. 2010. Asymmetry in the duration of El Niño and La Niña. *Journal of Climate* 23: 5826–5843.

Petersen, J. J. & H. C. Chapman. 1970. Chemical characteristics of habitats producing larvae of *Aedes sollicitans*, *Aedes taeniorhynchus*, and *Psorophora confinnis* in Louisiana. *Mosquito News* 30: 156–161.

Pierce, W. D., W.E. Duclus, & Y.M. Longacre. 1945. Mosquitoes of Los Angeles and vicinity. *The Sanitarian* 7: 718–726.

Reisen, W.K, D. Cayan, M. Tyree, C.M. Barker, B.F. Eldridge, & M. Dettinger. 2008. Impact of climate variation on mosquito abundance in California. *Journal of Vector Ecology* 33: 89–98.

Schmidt, N. & M.E. Luther. 2002. ENSO impacts on salinity in Tampa Bay, Florida. *Estuaries* 25: 976–984.

Seal, J.N. & W.R. Tschinkel. 2010. Distribution of the fungus-gardening ant (*Trachymyrmex septentrionalis*) during and after a record drought. *Insect Conservation and Diversity* 3: 134–142.

Shaman, J., J.F Day, & M. Stieglitz. 2002. Drought-induced amplification of Saint Louis encephalitis virus, Florida. *Emerging Infectious Diseases* 8: 575–580.

Szyniszewska, A.M., N.C. Leppla, N.C. Manoukis, T.C. Collier, J.M. Hastings, D.J. Kriticos, & K.M. Bigsby. 2020. CLIMEX and MED-FOES models for predicting the variability in growth potential and persistence of Mediterranean fruit fly (Diptera: Tephritidae) populations. *Annals of the Entomological Society of America* 113: 114–124.

van der Kuyp, E. 1954. Mosquitoes of the Netherlands Antilles and their hygienic importance. *Studies on the Fauna of Curaçao and other Caribbean Islands* 5: 37–114.

Vlach, J.J., K.J. Hall, J.F. Day, G.A. Curtis, L.J. Hribar, & E.M. Fussell. 2006. Interisland dispersal of the black salt marsh mosquito, *Ochlerotatus taeniorhynchus* (Diptera: Culicidae) in the Florida Keys. *Journal of the American Mosquito Control Association* 22: 615–621.

Woli, P., B.V. Ortiz, D. Buntin, & K. Flanders. 2014. El Niño-Southern Oscillation (ENSO) effects on Hessian fly (Diptera: Cecidomyiidae) infestation in the southeastern United States. *Environmental Entomology* 43: 1641–1649.

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Systema Dipterorum
The BioSystematic Database of World Diptera

Systema Dipterorum **Update – Spring 2022**

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The latest version of *Systema Dipterorum* (SD) posted online (ver. 3.8, 21 May 2022) totals 240,000+ records and includes 166,859 living species of flies in 12,690 genera. Data entry continues, and as of this writing we have broken through the 167,000 level of living species (167,155). We have finally caught up with all of the 14-year literature backlog we were left with when we took over the database in 2018, and we are now entering papers as they are published – while still finding a few papers that have been missed. Please send any newly published papers or those you know we may have missed to neale@bishopmuseum.org so we can enter the information into SD in a timely manner.

As always, SD is a community effort and a number of people have assisted in correcting entries, noting missing species or literature, and provided copies of hard-to-get literature. Shout outs to the following for their help in various ways since the last update in *Fly Times*: Adrian Pont, Anatoly Barkalov, Andrew Whittington, Art Borkent, Arthur Frost, Benny Chan, Bill Murphy, Brad Sinclair, Brian Brown, Carlo Monari, Chris Angell, Chris Cohen, Daniel Sommaggio, Daniel Whitmore, David Nicholson, Elisabeth Stur, Emily Hartop, Florian Mongin, Gabriele Miksch, Jean-Sébastian Girard, Jeff Skevington, Jere Kahanpää, Jeroen van Steenis, Jim O’Hara, Libor Mazanek, Lisa Fisler, Lorenzo Munari, María Eugenia Cano, Mark Mitchell, Mathias Jaschhof, Mélanie Herbert, Menno Reemer, Michael von Tschirnhaus, Mihaly Földvari, Morgan Jackson, Oliver Keller, Paul Beuk, Peter Cranston, Pjotr Oosterbroek, R. Holden Appler, Ralph Peters, Ray Gagné, Rob Oudejans, Sander Bot, Shannon Henderson, Stephen Smith, Steve Gaimari, Tamara Tóth, Torbjørn Ekrem, Verner Michelsen, Vlad Blagoderov, Ximo Mengual, and Zachary Dankowicz. Special thanks to Zach Dankowicz, who volunteered to enter the last few hundred (!!) or so articles in the references database, which helped us finally catch up on the literature backlog. Also, many thanks to the Catalogue of Life folks (Yury Roskov, Geoff Ower) for illuminating issues needing correction and/or resolution; and Rich Pyle for administering the database on our server and posting the updates.

We have been doing a bit of number crunching on authors, and who among them are in the top 10/50/100 etc. of those describing new species. We hope to present a deep-dive into the statistics in the next issue of *Fly Times*, but we can say at this point in our analysis that the top living Diptera author with ~2,400 species is Ding Yang. Zoya Fedotova follows with ~1,100. Next in line include Steve Marshall, Heikki Hippa, Brian Brown and Henry Disney. Will turbo taxonomy allow taxonomists to surpass Alexander’s ~11,000 species named, and will the cecids and phorids pass the tipuloids? ¯_(ツ)_/¯

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Call for Mariobezziinae (Bombyliidae) specimens

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I am a PhD candidate from Stellenbosch University, South Africa. My research overall aims to better understand the phylogenetic and evolutionary relationships among the different genera of Afrotropical Bombyliidae of the subfamily Mariobezziinae specifically within the Greater Cape Floristic Region of South Africa. My work is part of a larger research project funded by the Belgian Directorate-General for Development Cooperation (DGD) entitled DIPoDIP (Diversity of Pollinating Diptera in South African Biodiversity Hotspots) and coordinated by Kurt Jordaens of the Royal Museum for Central Africa (RMCA) in Belgium. The project website can be found at https://www.pindip.org/.

To better understand the phylogenetic relationships of South African Mariobezziinae to other Afrotropical genera, I aim to reconstruct a genus-level phylogeny of Mariobezziinae that will hopefully include genera that are found outside South Africa. I have had great success collecting Mariobezziinae within South Africa from the genera *Corsomyza* (with exception of *C. ochrostoma*), *Callynthrophora, Megapalpus* and one species of *Hyperusia.* However, I am struggling to find specimens from other Afrotropical regions. Thus, I ask that if anyone has any Mariobezziinae specimens from recent Afrotropical collections (2015 to present) that they are willing to donate to this research, to please contact me.

Species and historical distributions are listed below:

Genus	**Species**	**Author**	**Distribution**
Gnumyia	*brevirostris*	Bezzi, 1921	Namibia, South Africa
	fuscipennis	Hesse, 1938	South Africa, Zimbabwe
Hyperusia	*apiformis*	Greathead and Evenhuis, 2001	Tanzania (50–70 miles north Dodoma)
Mariobezzia	*ebneri*	Becker, 1922	Sudan
Pusilla	*longirostris*	Paramonov, 1954	DR Congo
Zyxmyia	*megachile*	Bowden, 1960	Kenya, Tanzania

Persons who donate specimens to this project will be properly acknowledged and will be updated on the progress of the phylogeny reconstruction.

If you are interested in my research and would like to know more, feel free to email me, and if you would like more information on the DIPoDIP project, feel free to email Kurt Jordaens (kurt.jordaens@africamuseum.be). Many thanks and all the best!

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How not to lose legs, or my experience of collecting and preserving crane flies (Diptera: Tipuloidea)

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Dedicated to the memory of my teacher Oleg Pavlovich Negrobov, and my darling mother Elena Nikolavena Lantsova (*Metneva*)

Abstract. An overview of crane fly collecting methods is given, and the advantages and disadvantages of various preservation techniques are discussed. It is suggested that specimens be kept alive and then killed just before they are prepared, and that they be prepared and pinned the day they are collected. It is recommended that the preparing be done using a binocular microscope, and that the specimens be arranged on blocks of polyethylene foam with minuten pins used to position the legs, and later the specimens are mounted on card points.

Legs of crane flies and their importance in identifying species.

First, let me quote two characteristic statements. “To most entomologists, the words ‘crane flies’ at once suggest the legless condition in which these insects are all too often found in collections” (Byers, 1961). “Joke at the CNC: a key character for Tipuloidea is ‘less than six legs’”. (Fenja Brodo personal communication – the letter from 1.05.2021).

Crane flies are members of the following families: Tipulidae, Limoniidae, Pediciidae and Cylindrotomidae (Oosterbroek, 2022). In comparison with other groups in the Order Diptera, they have extremely long and brittle legs (Alexander, 1927; Stubbs, 1972; Savchenko, 1983, 1986) which are very often lost at different stages of collecting and working on the specimens. If improperly preserved and prepared, loss of legs occurs when the material is being worked on; in addition, space in the entomological boxes and drawers is limited.

The loss of crane fly legs is to be avoided because several leg structures have taxonomic significance. In Tipulidae, for example, “the number of spurs on tibiae, the nature of pubescence, color of trochanters, shape of femora, presence or absence of snow-white bands on femora and (or) tibiae, as well as the structure of male claws are diagnostic, and these features are used to characterize species and subgenera, less often genera and other higher taxa” (Savchenko, 1983). Limoniidae also have long and brittle legs. Like Tipulidae, their legs usually separate at the trochanter / femur contact. Presence or absence, and size and number of spines on tibiae are of higher taxonomic significance in the Limoniidae (Savchenko, 1986).

Often, especially for inexperienced researchers, the loss of legs in crane flies becomes a problem that can create psychological obstacles for further work with this group of Diptera. Savchenko (1983) rightly notes that “the fragility of the legs is one of the reasons why long-legged insects are usually collected less willingly than other, more ‘durable’ insects.” In addition, there is no information on methods for preserving legs of crane flies in the numerous available general guides for the collection and preservation of insects (e.g., Oldroyd, 1958; Peterson, 1959; Schauff, 1986; Walker et al., 1988; Millar et al., 2000; McGavin, 2007; Grootaert et al., 2010; Krogmann & Holstein, 2010; Golub et al., 2012; Upton & Mantle, 2010; Ferro & Summerlin, 2019).

The author, therefore, decided to summarize and share his methods, as well as those of colleagues, developed over many years of collecting expeditions of crane flies in the Caucasus (since 1994).

General remarks on collection of crane flies.

Sweeping. As was rightly noted earlier (Byers, 1961), the preservation of tipuloids begins from the moment of their collection in the field. Sweeping with an entomological net is the main method for collecting crane flies (Alexander, 1927; Byers, 1961; Stubbs, 1972a, and others), however, "some groups of flies (Bombyliidae, Tipulidae, midges) rarely survive sweeping in good condition" (Martin, 1977). To mitigate this, the structure of the net and how the sweeping is carried out is very important. The author uses a net with a wide rounded bottom, with a folding hoop diameter of 40 cm and a bag length of 1 m. A net with such a large volume ensures better preservation of specimens, prevents injury of larger specimens of tipulids, for example, the subgenus *Acutipula*, and makes it possible to collect significant numbers of specimens at a time.

An entomological net with an elongated telescopic handle, varying from 62 to 151.5 cm, is very convenient, and is indispensable in difficult to access habitats for collecting tipuloids such as *Helius* (Limoniidae) on the banks of stagnant water bodies, and *Dicranomyia* and *Molophilus* (Limoniidae) swarming over plants on wet rocky walls in narrow gorges.

Routine sweeping with a net for *Dolichopeza* or *Limonia*, crane flies which have very long and brittle legs, especially in thickets of butterbur or reeds, can lead to leg loss (Byers, 1961). A better tactic is to move slowly, touching the plants with a net, to startle the crane flies and catch them on take off.

Collecting crane flies using artificial light. This "is one of the most productive ways of collecting nocturnal flying insects…" (Martin, 1977), a group to which many crane flies belong. Some not so abundant species that are not easily swept during the day, can be better collected this way. For example, the alpine species *Tipula* (*Vestiplex*) *pallidicosta pullata* Savchenko, 1960 was collected at lights, in the foothills of the North Caucasus on the northern slopes of the Dzhinal Ridge (about 1000 m a. s. l.) in the Zolka Yuzhnaya River valley, where it is not numerous at this altitude. The obvious minus of this method, according to Y.I. Chernov (personal communication), is its "non-ecological nature" – in other words, the true habitats of species that are caught by lights remains unclear. This method is not applicable in the Far North because of the long polar day, and is not practical in the mountains above the tree line due to low temperatures in the evening and nighttime.

Malaise traps. This method for collecting crane flies was used by a number of researchers (Townes, 1962, 1972; Matthews & Matthews, 1971; Ol'schvang, 1978, 1992; Dufour, 1980, 1986; Tereshkin & Shlyakhtenok, 1989). The use of Malaise traps for collecting crane flies is not always successful, for example, in the Canadian northern boreal/subarctic region of Labrador and Quebec (Brodo, 1987). The disadvantages of these traps for faunistic research (insects other than crane flies) were noted after long-term use in Sweden (Karlsson et al., 2020) and in Costa Rica (Borkent et al., 2018). These included incomplete assessment of the fauna and poor preservation of fragile insects making them difficult to identify. Evans (2016) noted: "Catch rates [of Malaise traps in New Zealand] are highly susceptible to small changes in location and are highly site dependent".

The author had a very short-term use of Malaise traps in the Caucasus – on the Abago Plateau (in 2018) and in the Yew-boxwood grove (in 2019) (Caucasian Nature reserve). This experience showed that there are difficulties associated with the use of such traps. It is not easy, and sometimes impossible, to revisit collection sites to remove specimens from the traps, and one can lose these traps due to vandalism.

Barber's pitfall traps. Pitfall traps gave good results for collecting and assessing crane flies, which move on the surface of the soil, such as arctic species *Tipula* (*Pterelachisus*) *carinifrons carinifrons* Holmgren, 1883, its daily and seasonal adult population patterns of abundance in the arctic tundra subzone (near village Dikson, Taimyr Peninsula) (Lantsov & Chernov, 1987). Barber's pitfall trap was useful for capturing cave insects (Barber, 1931; Skvarla et al, 2014), particularly wingless limoniids of genus *Chionea* (Novak, 2005; Novak e al., 2007). Whereas a pitfall trap may not be great for crane flies in general.

Adhesive tapes. In the Far North (near Cape Barrow, Alaska), adhesive tapes "masonite strips 1 m. x 0.1 m., covered by a sticky resin" were used for studying seasonal patterns of abundance of adult crane flies *Tricyphona (Tricyphona) hannai antennata* (Alexander, 1956) (as *Pedicia*), *T. c. carinifrons,* and *Prionocera recta* Tjeder, 1948 (as *Prionocera gracilistyla* Alexander, 1956) (MacLean & Pitelka, 1971).

These two last methods are used both for faunistic and ecological research (study of daily and seasonal dynamics of abundance, establishment of dominant species, etc.). However, these methods result in poor specimens of crane flies especially in mid season when many other invertebrates fall into these traps.

Advantages and disadvantages of traditional preservation methods of crane flies.
Paper envelopes are used for long-term storage of crane flies or until they can be mounted (Alexander, 1927; Byers, 1961; Martin, 1977; Gelhaus, 2005). Alexander (1927) noted that specimens placed in paper envelopes are not damaged when mailed. According to Byers (1961), crane flies extracted from envelopes are all in one plane and gluing them onto stiff paper points prevents loss of legs. The disadvantages of this method is the difficulty of properly placing the legs and wings of the crane flies in the envelope, and the probability of losing legs when removing specimens from these paper envelopes.

Layering. This is another widely used method of storing various insects, including crane flies. It is recommended to use soft materials such as facial tissue or glazed cotton, but not absorbent cotton "because appendages may catch in it and break" (Martin, 1977). One of the dubious advantages of this method is that if legs are broken or separated from the thorax, they remain close to the damaged specimen. To prevent damage to the specimen when it is removed from the layer, work very carefully under the binoculars, using flexible tweezers and scissors for cutting fibers of cotton, if necessary.

Alcohol (70 or 100%). The legs of crane flies are well preserved, especially when specimens are dropped into alcohol immediately after capture, following the rule: one tube – one specimen, although the latter is not always possible. The obvious disadvantage of this method is the discoloration of the specimens. The silvery coating on the head, thorax and abdomen, as well as the color of the elongated stripes on the thorax and the interval between, are often lost depending upon the duration and storage conditions (Vockeroth, 1966; Martin, 1977; Walker et al., 1988). In addition, skills in preparing wet material are required.

Proposed key steps for the preservation of specimens.
These rules are very close to those for mounting of Microlepidoptera (Landry & Landry, 1994).

1. Catching crane flies by a few sweeps, and very carefully removing specimens from net.
2. Placing individual specimens into separate tubes.
3. Keeping specimens alive until they are prepared.

4. Killing crane flies with ethyl acetate when ready to be prepared. (some prefer to use ethylene dichloride as a killing fluid (F. Brodo, personal communication))
5. Pinning and preparing specimens using minuten pins on blocks of polyethylene foam under a binocular microscope. The antennae and wings should be positioned so that they do not obscure each other, and the legs should be prepared around the main pin.
6. Subsequent removal of minuten pins from the polyethylene foam, also under binocular microscope, and gluing on card points with a standard insect pin holding the card

Removing collected crane flies from the net.
Before removing crane flies, it is advisable to swiftly shake the net to concentrate the flies at the bottom, and then to bend the fabric over the raised rim of the net to prevent specimens from flying out. Then select them one by one, using an aspirator and placing each in a separate vial (Stubbs, 1972a). The author uses an aspirator with a removable container, is made from 112 mm of a centrifuge tube with and inner diameter of 27 mm (Fig. 1). It is easier to do this while sitting with the net in one's lap (Fig. 2). For this, it is convenient to have a lightweight folding chair. Removable aspirator tubes can hold 2-5 specimens of small limoniids. It is suggested to put into the tube a folded strip of filter paper. Crane flies should remain alive until they are killed with ethyl acetate, or they may be immediately placed in alcohol. When collecting a mass of specimens, it is better to kill them while they are in the net, by placing the net in a plastic bag with a piece of cotton wool soaked in ethyl acetate, and then put in alcohol.

Figure 1 (left). Aspirator with replaceable container - centrifuge tubes. **Figure 2** (right). Removing crane flies from the net in the field. Dagestan reserve, Kizlyarskiy region, 24.05.2017. Collection of *Erioptera* (*Mesocyphona*) *bivittata* (Loew, 1873) and *Idiocera* (*Idiocera*) *pulchripennis* (Loew, 1856) in a sedge-reed-tussock community near an artesian water source.

For preliminary storage of living specimens, 5 ml (14 x 50 mm), 7 ml (15 x 62 mm) or 10 ml (16 x 80 mm) polypropylene round-bottom cylindrical tubes with snap cap may be used, as well as glass tubes with cotton stoppers 65-80 mm high and 18 mm inner diameter. It is better to pack these tubes in a cartridge belt holding 5 to 8 tubes.

For larger species, especially of the subgenera *Lunatipula* and *Acutipula*, glass vials 95–105 x 31–36 mm, closed with cotton stoppers are more convenient. These are usually used for rearing adults from larvae and pupae. It is very convenient to use such large vials and wider tubes for collecting from wet rock outcrops, common habitats for *Pseudolimnophila (Pseudolimnophila) sepium* (Verrall, 1886) and *Tipula (Emodotipula) obscuriventris* Strobl, 1900, as well as for collecting mating pairs that move slowly.

Killing and subsequent preparing of specimens of crane flies.
The most important rule is not only just processing the material on the day of its collection, which, of course, is not something new and which is common for most entomologists, but keeping the collected specimens in individual containers alive until one is ready to prepare them. The same advice can be found in Martin (1977): "Pin specimens within a few hours of capture, while their internal parts are still soft and their appendages pliable."

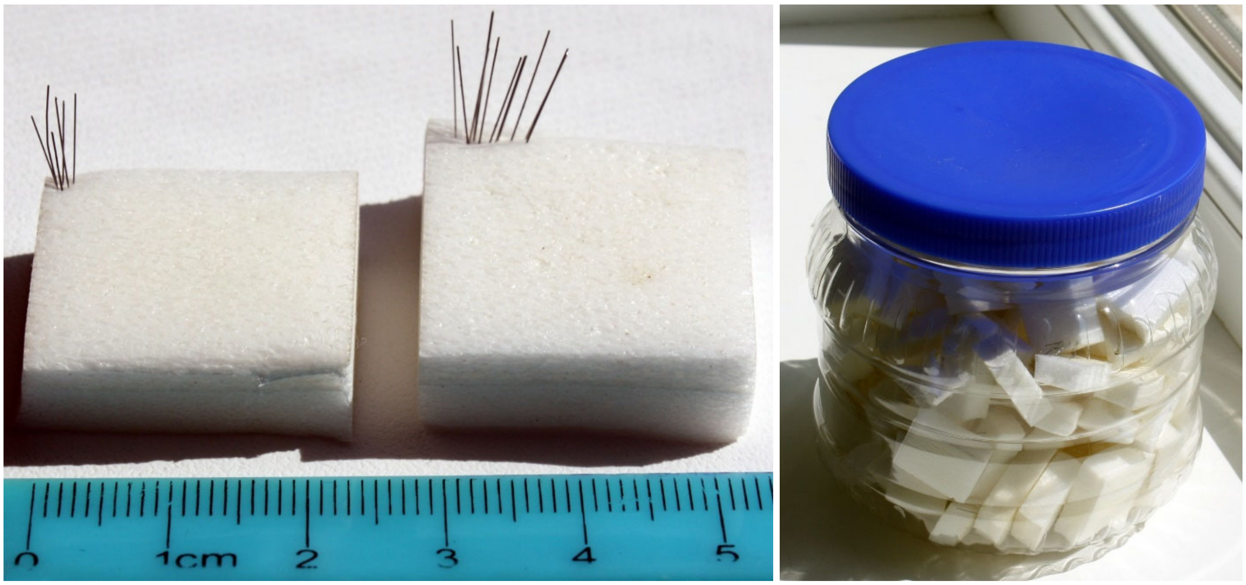
If crane flies are killed as soon as they are removed from the net, then they can dry out so much that their legs will break off. Long hours in the field followed by a long trip to the laboratory, necessitates that special attention be paid to ensure that the material does not dry out during transportation. Therefore, vials should be kept away from direct sunlight, preferably in special boxes or thermos bags made of foam. When not possible to prepare specimens on the day they were collected, vials with live crane flies may be placed in the refrigerator overnight for subsequent work the next day.

Over exposure of crane flies to ethyl acetate vapor is to be avoided as this causes increased fragility of their legs. The author uses polyethylene foam, "Izolon" brand, which is a white, rather dense substance, 8 mm thick, and is better material for the pinning base in entomological boxes.

Minuten pins are pierced into a corner of the polyethylene foam (Fig. 3). It is better to use 15 x 0.29–0.38 mm minutens. These polyethylene foam blocks with the minutens are conveniently stored in a light transparent plastic jar (Fig. 4). Usually 15–20 minuten pins are used for preparing one specimen. For transferring specimens from vials, use soft, flexible tweezers with rounded and smooth ends. For handling the minutens used for preparing, it is better to use steel rigid short entomological tweezers with thin ends.

Before preparing, a minuten pin is pushed through the side of the crane fly thorax approximately at the border of the anepisternum and anepimeron and is fixed to the foam block. Then the antennae and wings are carefully prepared out and staked with minutens, care being taken so that they do not obscure each other and the venation can be clearly seen on both sides of the wing.

Each specimen is braced with extra pins in order to hold the antennae, legs and wings in position until they dry. Special attention should be paid to leg positioning. When preparing crane flies with rather long abdomens and relatively short legs, pins may be laid according to Gelhaus (2005) (Fig. 5). However, when preparing crane flies with relatively short bodies and very long legs, it is better to place the legs around the central minuten or entomological pin, so that they do not stick out to the side and do not protrude beyond the label. (Fig. 6-9). This achieves two goals – the preservation of specimens when working with the collection and the saving of space in entomological boxes. Pieces of polyethylene foam are pierced by a standard entomological pin (Fig. 10, 11), with a temporary label attached below, and placed in boxes. Later the pieces of polyethylene foam with dried specimens are placed on the stage of a binocular stereomicroscope, and the minutens are removed by strong entomological tweezers. One should be very careful removing the minutens, and do it slowly to avoid bending and straightening them abruptly, which could damage the specimen. Then the specimen is mounted (pinned) on a card point, which in turn is mounted to a standard insect pin (double mounted). The legs are saved because facing the main carrier entomological pin (Fig. 8).



Figures 3–4. Polyethylene foam blocks. **3**. With minutens for preparing crane flies. **4** (right). Container for their storage and transportation.

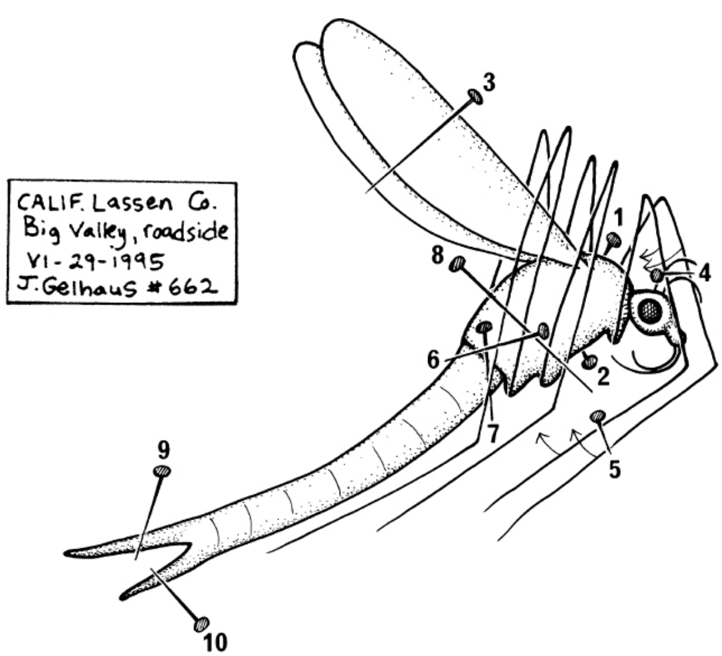
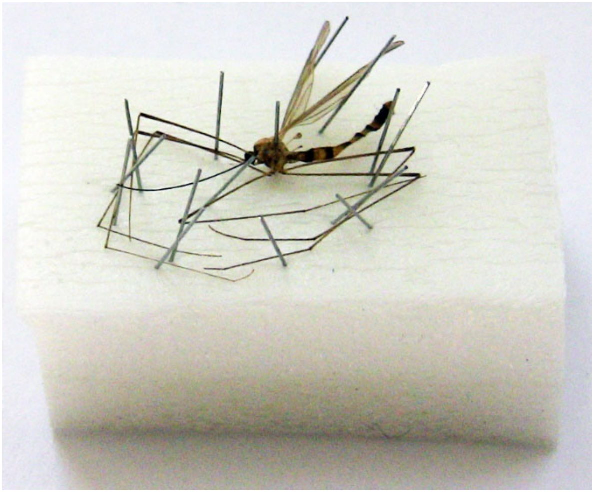
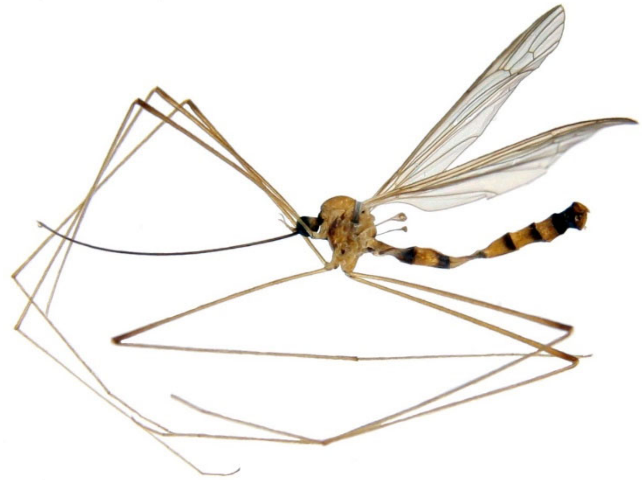


Figure 5. The mode of preparing crane flies of Jon Gelhaus (2005).

The method of preserving crane flies, presented here, is also not without its disadvantages. The legs can be lost at each stage. In addition, it requires a certain amount of time and a lot of patience to work with the material in this manner. However, the result – completely intact specimens in the collection and their subsequent preservation, justifies the efforts expended.

According to Ross (1941) “because of their unwieldy legs these insects [crane flies] should have a double card point mount and the legs should be kept away from the pin to avoid their breakage in handling” (Fig. 12).





Figures 6–7. *Elephantomyia* (*Elephantomyia*) *edwardsi* Lackschewitz, 1932. **6** (left). Prepared with minutens on polyethylene foam. **7** (right). After removal of minuten pins.

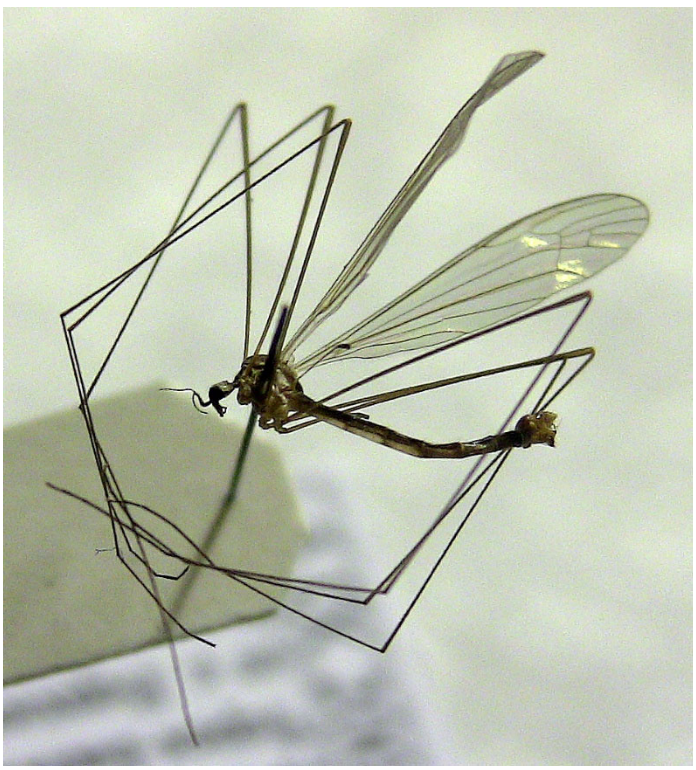


Figure 8 (left). *Dicranomyia* (*Idiopyga*) *halterella* Edwards 1921, double mounted 1♂. **Figure 9** (right). *Tipula (Pterelachisus) trifascingulata* Theowald, 1980, 1♂.

It is clear for everyone who has worked with tipuloids that this method of mounting is definitely unacceptable, since with such an arrangement of legs, specimens will take up a lot of space in entomological boxes and, most importantly, legs will inevitably break when working with material.

Martin (1977) wrote: "Bunch and support drooping legs of Tipulidae during hardening by placing a piece of card about 9 mm (3/8 in.) below them on the pin". Schauff (1986) also suggested: "With long-legged species or those with drooping abdomens, the legs and abdomens may be supported until dry with a piece of stiff paper pushed up on the pin from beneath. Once the specimens are dry, this paper support can be removed." This mounting method can certainly be applied to crane flies, but not to all species. It can be applied to species with relatively short legs, for example, *T. c. carinifrons*. However, it is of little use for species with very long legs in the genera *Dolichopeza*, *Acutipula*, etc.

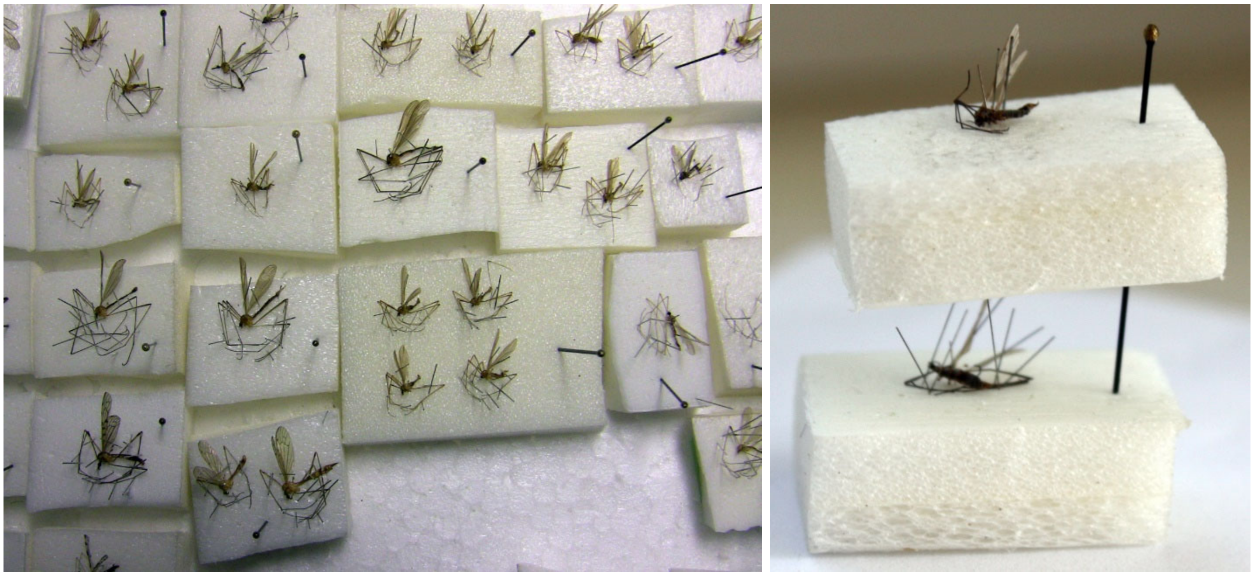


Figure 10–11. Prepared crane flies, **10** (left). Ready for transportation. **11** (right). Temporary placement of two on a single entomological pin.

Another danger is the growth of molds. This can happen in summer if the material was collected at high altitudes with high humidity and low temperatures, and then transported in boxes for a long time at high temperatures. In this case, it is necessary to periodically ventilate the boxes holding the pinned material. "… Tightly closed, impervious containers of metal, glass, or plastic should be avoided. Nothing can be done to restore a moldy specimen" (Schauff, 1986).

Unique method of killing crane flies, or an unusual case during the "hunting" of insects in Dagestan (the North-East Caucasus, Russia).

Crane flies were collected in the Samur liana forest on 05.17.2014 (41°51'05.3"N / 48°32'27.9"E, -16 m) at a slowly flowing stream about 1.5–2 meters wide (Fig. 13).

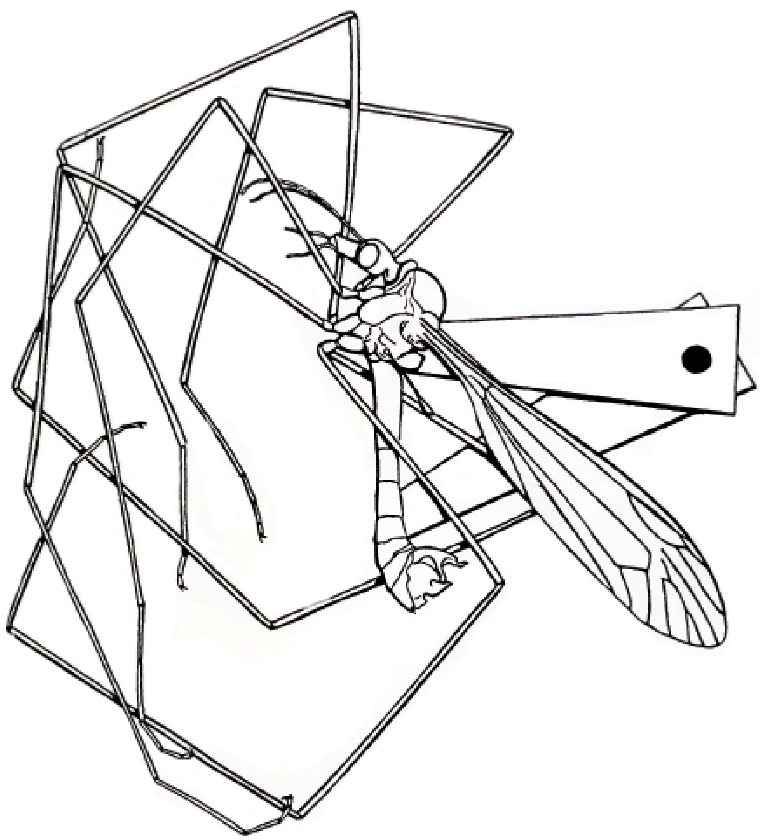


Figure 12. The mode of preparing and mounting of crane flies according to Ross (1941).

The air temperature was in the range of 27–30°C. The vegetation on the banks of the stream was in complete shade. The collection of insects was by sweeping (about 10 sweeps) along the stream over grasses and occasional bushes. It was decided to remove insects from the net about 10–15 m from the place of collection, on a high hornbeam stump brightly lit by the sun. While the net was being transferred to this place, the insects remained in the shade and were active. As soon as the bag of the net was exposed to the bright sun and opened, it turned out that all the insects in it immediately fell asleep as if they had been treated with ethyl acetate. The net contained 2♂♂ and 2♀♀ of *Gnophomyia viridipennis* (Gimmerthal, 1847) and several small representatives of other Diptera families. Repeated moving and transferring of the collected insects to a sunlit area had the same effect. It was obvious that the insects were immobilized by a sharp change in the light and temperature regime. Some of them, including the limoniids, died. The author was unable to find a description of such a phenomenon in the literature.

Figure 13. South-east of Dagestan, Samur liana forest on 05.17.2014 (41° 51'053" N / 48° 32'279"E, ~ -16 m). Shaded biotope at a slowly flowing stream - habitat of *Gnophomyia viridipennis* (Gimmerthal, 1847).

Conclusions.

Achieving the best possible preservation of collected specimens of crane flies, including the preservation of the most vulnerable – the legs – is the most important task (for collectors and for taxonomist as well) in the study of tipuloid dipterans, due to the fact that the morphology of the legs is important in the taxonomy of the group, and cannot be ignored either when compiling keys or describing new species.

Comparison of the known methods of collecting crane flies shows that sweeping and collections at lights are optimal for getting the best specimens for preservation. Due to the fact that the probability of losing legs is constantly very high, it is important to be very attentive and careful at all stages of collecting and preserving material.

The proposed six stages of crane flies preservation – from removing the specimen from the net to preparing the specimen on a block of polyethylene foam under a microscope and then double mounting it – makes it possible to obtain a perfectly prepared and preserved specimen, which, moreover, does not take up much space in the entomological box. The disadvantage of this method is that it requires a certain amount of time and a lot of patience. When preparing, it is optimal to position the legs around the pin.

Acknowledgments.
The author sincerely thanks colleagues for their professional comments and recommendations – Pjotr Oosterbroek (Amsterdam, Nederland), Valentin Pilipenko and Elena Lukashewich (Moscow, Russia), Alexandr Lynov (Voronezh, Russia). The author expresses his deep gratitude to Fenja Brodo (Ottawa, Canada) for her valuable advice and editing of English.

References

Alexander, C.P. 1927. Crane-flies or Daddy Long-legs. *Victorian Naturalist* 44: 4–8.

Barber, H.S. 1931. Traps for cave-inhabiting insects. *Journal of the Elisha Mitchell Scientific Society* 46: 259–266.

Borkent, A., Brown, B.V., Adler, P.H., Amorim D.S., Barber, K., Bickel, D., Boucher, S., Brooks, S.E., Burger, J., Burington, Z.L., Capellari, R.S., Costa, D.N.R., Cumming, J.M., Curler, G., Dick, C.W., Epler, J.H., Fisher, E., Gaimari, S.D., Gelhaus, J., Grimaldi, D.A., Hash, J., Hauser, M., Hippa, H., Ibañez-Bernal, S., Jaschhof, M., Kameneva, E.P., Kerr, P.H., Korneyev, V., Korytkowski, C.A., Kung, G-A., Kvifte, G.M., Lonsdale, O., Marshall, S.A., Mathis, W.N., Michelsen, V., Naglis, S., Norrbom, A.L., Paiero, S., Pape, T., Pereira-Colavite, A., Pollet, M., Rochefort, S., Rung, A., Runyon, J.B., Savage, J., Silva, V.C., Sinclair, B., Skevington, J.H., Stireman III, J.O., Swann, J., Vilkamaa, P., Wheeler, T., Whitworth, T., Wong, M., Wood, D.M., Woodley, N., Yau, T., Zavortink, T.J. & Zumbado, M.A. 2018. Remarkable fly (Diptera) diversity in a patch of Costa Rican cloud forest: Why inventory is a vital science. *Zootaxa* 4402 (1): 053–090 https://doi.org/10.11646/zootaxa.4402.1.3.

Brodo, F. 1987. A revision of the genus *Prionocera* (Diptera: Tipulidae). *Evolutionary Monograph* 8: 93pp.

Byers, G.W. 1961. The crane fly genus *Dolichopeza* in North America. *The University of Kansas Science Bulletin* 42: 665–924.

Dufour, C. 1980. Un nouveau piegelumineux pout la capture des Tipulidae et autres Dipteres Nematoceres: unetente Malaise lumineuse. *Mitteilungen der Schweizerischen Entomologischen Gesellschaft* 53: 313–320.

Dufour, C. 1986. Les Tipulidae de Suisse (Diptera, Nematocera). *Documenta Faunistica Helvetiae* 2: 187 pp., fiches 149 pp.

Evans, A. 2016. Inventory and monitoring toolbox: invertebrates Malaise trapping Version 1.0: 18 pp.

Ferro, M.L. & Summerlin, M. 2019. Developing a standardized list of entomological collection methods for use in databases. *ZooKeys* 861: 145–156. https://doi.org/10.3897/zookeys.861.32347

Gelhaus, J.K. 2005. Systematics and biogeography of the desert crane fly subgenus *Tipula* (*Eremotipula*) Alexander (Diptera: Tipulidae). *Memoirs of the American Entomological Society* 46: 235 pp.

Golub, V.B., Tsurikov, M.N. & Prokin, A.A. 2012. *Insect collections: collecting, preparing and preserving of material*. Moscow. Partnership of scientific publications KMK: 339 pp. [in Russian].

Grootaert, P., Pollet, M., Dekoninck, W. & Achterberg, C. 2010. Sampling insects: general techniques, strategies and remarks. In: Eyman, J., Degreef, J. Häuser, Ch., Monje, J.C., Samyn, Y. & Vandenspiegel, D. Manual on field recording techniques and protocols for All Taxa Biodiversity Inventories and Monitoring. *ABC Taxa* 8(2): 377–399.

Karlsson, D., Hartop, E., Forshage, M., Jaschhof, M. & Ronquist, F. 2020. The Swedish Malaise Trap Project: A 15 Year Retrospective on a Countrywide Insect Inventory. *Biodiversity Data Journal* 8: e47255. https://doi.org/10.3897/BDJ.8.e47255

Krogmann, L. & Holstein, J. 2010. Preserving and Specimen Handling: Insects and other Invertebrates. In Eyman, J., Degreef, J., Häuser, Ch., Monje, J.C., Samyn, Y. & Vandenspiegel, D. Manual on field recording techniques and protocols for All Taxa Biodiversity Inventries and Monitoring. *ABC Taxa* 8(2): 463–481.

Landry, J.-F. & Landry, B. 1994. A technique for setting and mounting Microlepidoptera. *Journal of the Lepidopterists' Society* 48(3): 205–227..

Lantsov, V.I. & Chernov, Y.I. 1987. *Tipuloid crane-flies in the tundra zone*. Moscow. Nauka: 176 pp. [in Russian].

McGavin, G.C. 2007. *Expedition field techniques insects and other terrestrial arthropods*. Published by Geography Outdoors: the centre supporting field research, exploration and outdoor learning. London: 94 pp.

MacLean, S.F. & Pitelka, F.A. 1971. Seasonal patterns of abundance of tundra arthropods near Barrow. *Arctic* 24 (1): 19–40.

Martin, J.E.H. 1977. Collecting, preparing and preserving insects, mites and spiders. In: *The insects and arachnids of Canada*. Part 1. Agriculture, Canada: 182 pp.

Matthews, R.W. & Matthews, J.R. 1971. The malaise trap: its utility and potential for sampling insect populations. *The Michigan Entomologist* 4(4): 117–122.

Millar, I.M, Uys, V.M. & Urban, R.P. 2000. *Collecting and Preserving Insects and Arachnids. A Manual for Entomology and Arachnology (ARC – Plan)*. Pretoria, South Africa: 105pp.

Novak, T. 2005. Terrestrial fauna from cavities in Northern and Central Slovenia, and a review of systematically ecologically investigated cavities. *Acta Carsologica, Ljubljana* 34: 169–210.

Novak, T., Sivec, I., Janzekovic, F. & Christian, E. 2007. *Chionea austriaca* in caves and artificial galleries of Slovenia (Diptera, Limoniidae). *Revue Suisse de Zoologie* 114: 49–57.

Oldroyd, H. 1958. *Collecting, preserving and studying insects*. Hutchinson, London, Macmillan, New York: 327 pp.

Ol'shvang, V.N. 1978. Notes on crane flies of the genus *Tipula* in the zonal and mountain tundra of the Urals and the Yamal Peninsula. Fauna, ecology and variability of animals. *Sverdlovsk*: 15–16. [in Russian].

Ol'shvang, V.N. 1992. The structure and dynamics of the insect population of South Yamal. Ekaterinburg: Science. *Ural'skoe otdelenie RAS*: 104 pp. [in Russian].

Oosterbroek, P. 2022. *Catalogue of the Craneflies of the World*. Available at: https://ccw.naturalis.nl/ (latest update 25 January 2022).

Peterson, A. 1959. *A manual of entomological techniques. How to work with insects*. 9th ed. Edward Brothers Inc., Ann Arbor, Michigan: 435 pp.

Ross, H.H. 1941. How to collect and preserve insects. *Illinois Natural History Survey, Urbana, Circular* 39: 48 pp. http://hdl.handle.net/2027/coo.31924016950622

Savchenko, E.N. 1983. Crane-flies (Fam. Tipulidae), Introduction, Subfam. Dolichopezinae, subfam. Tipulinae (start). *Fauna USSR, N.S.* 127, Nasekomye Dvukrylye [Diptera], 2(1-2): 585 pp. [in Russian].

Savchenko, E.N. 1986. Komary-limoniidy [limoniid-flies].(General description, subfamilies Pediciinae and Hexatominae). *Fauna Ukrainy* 14(2): 380 pp. [in Russian].

Schauff, M. E. (editor), undated. Collecting and preserving insects and mites: techniques and tools. Systematic entomology laboratory. USDA National Museum of Natural History. Washington. 68 pp. [Updated and modified version of the *USDA Misc. Publication* 1443 published by the Agricultural Research service in 1986 and Edited by George C. Steyskal, William L. Murphy and Edna M. Hoover.]

Skvarla, M.J., Larson, J.L. & Dowling, A.P.G. 2014. Pitfalls and preservatives: a review. *Journal of the Entomological Society of Ontario* 145: 15–43.

Stubbs, A.E. 1972. Why do craneflies have long legs? *Entomologists Record and Journal of Variation* 84: 108–111.

Stubbs, A.E. 1972a. Introduction to crane flies [Parts 1 and 2]. *Bulletin of the Amateur Entomologists Society* 31: 46–54, 83–93.

Tereshkin, A.M. & Shlyakhtenok, A.S. 1989. Experience of using the Malaise trap for studying insects. *Zoologicheskiy zhurnal* 68 (2): 290–292. [in Russian].

Townes, H. 1962. Design for a Malaise trap. *Proceedings of the Entomological Society of Washington* 64(4): 253–262.

Townes, H. 1972. A light-weight Malaise trap. *Entomological News* 83: 239–247.

Upton, M.S. & Mantle, B.L. 2010. Methods for Collecting, Preserving and Studying Insects and Other Terrestrial Arthropods. *Australian Entomological Society. Miscellaneous Publications* 3, Fifth Edition, Canberra: 81 pp.

Vockeroth, J.R. 1966. A method of mountain insects from alcohol. *The Canadian Entomologist* 98(1): 69–70.

Walker, A.K., Trevor, K. & Crosby, T.K. 1988. *The preparation and curation of insects*. Revised edition Wellington: 92 pp.

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The crane flies (Tipuloidea) of the Town of Kent, Putnam County, New York

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Epiphragma fasciapenne Say, one of the species found in the Town of Kent and across northeastern North America; each wing ~11.6 mm long.

We are documenting here a year-long investigation of the crane flies as part of a biological inventory undertaken by the town of Kent in 2021. Kent lies in Putnam County, ca. 80 km north of New York City. It is approximately 44 square miles and about half of this terrain is water in the form of ponds, lakes, streams, and wetlands. It lies roughly between 41º26' and 41º30' N latitude, and 73º40' to 73º47' W longitude, and with an elevation ranging between 175 and 385 m with most of the collecting at around 200 m. WRB, a long-time resident of this town, and a professional bryologist, is, of course, inventorying the mosses of this region, but for a personal challenge, he opted to collect crane flies (Tipuloidea) when FB expressed interest in identifying whatever he might collect. EAH is a retired naturalist and assisted WRB in netting crane flies during the spring and summer months. Later in the season, a porch light drew in many more specimens as well as species in the related family of winter crane flies (Trichoceridae). Collecting started in the middle of May and has never really stopped because the Town of Kent has decided to continue this biological inventory for at least another year.

Crane fly taxonomy is in flux. What were once considered to be subfamilies of Tipulidae were elevated to family status, but more recent molecular work indicates that perhaps this is incorrect. Never-the-less, we are opting to use the nomenclature found in the indispensable *Catalogue of the Craneflies of the World* (CCW), created and maintained by Pjotr Oosterbroek (2022). We therefore, recognize the families Tipulidae, Cylindrotomidae, Limoniidae and Pediciidae. The related families Trichoceridae (winter crane flies) and Ptychopteridae (fold-wing crane flies) are included in this survey but are not covered in the CCW. The full list of species collected is in the Appendix.

In 2021, a total of 74 species of crane flies (Tipuloidea) were collected as well as six species of Trichoceridae and two species of Ptychopteridae. Four of the Tipuloidea taxa could not be identified to species because of a lack of male specimens, crucial to the positive identifications in these cases; however, each clearly represented an additional species. For purposes of comparisons, we are restricting this list to the 70 identified species.

The first collection was a hand-netted crane fly on 21 May, a male *Tipula* (*Pterelachisus*) *trivittata* Say, and the last true crane fly collected was *Dicranomyia frontalis* (Staeger), collected surprisingly late in the season, 2 December at a porch light, from 5–9 pm, with a temperature of 50–51ºF (10ºC). To our surprise, that porch light attracted flies, particularly *Trichocera* species, right through December and into January 2022, on evenings when the temperatures crept above freezing. These Trichoceridae should overwinter as adults and reappear again early in the spring (February to April) at which time the females will oviposit eggs that should emerge as adults in the late fall of 2022.

Rarely does an insect inventory such as this, in temperate parts of the Northern Hemisphere, continue through the winter. A notable exception was the All-Taxa Inventory in the Great Smoky Mountains when the crane flies were surveyed using various traps that were emptied continuously from October 2000 to October 2002 (Petersen et al. 2004).

The most intriguing species collected was one that we could only identify as *Atypophthalmus* sp., a genus and species new to North America. We had twenty specimens between 4 September and 9 October, most collected at the porch light. Jon Gelhaus confirmed our identification and volunteered that he had been seeing this species for several years now in the greater Philadelphia area, but only females, and so came to the conclusion that it must be parthenogenetic. Perhaps molecular work will help us identify this species or it will be described as new. One of these females, collected at the light, had about 40 mites clinging to its abdomen (see photo below). How it managed to fly carrying that burden is a mystery. No mites were seen on any of the other specimens.

Species diversity as well as population sizes can differ significantly from year to year. Some years are more favorable to some species than to others for many reasons, including the general weather patterns. Then too, collectors get more proficient with practice, and that may be one of the reasons more specimens were collected as the season progressed. We have a list of species found, but little idea of those that were missed. No Cylindrotomidae were collected; these tend to be rare and in wetlands, so they may be in Kent but not yet found. Generally one expects about 30% more Limoniidae species than Tipulidae, and so far we have 38 Limoniidae compared to 32 Tipulidae. Even so, it is interesting to compare our list for the Town of Kent to collections made elsewhere.

The crane flies of New York have been very well documented by Alexander in several publications. His monograph on the crane flies of New York. Part 1. (1919), dealt with the adults and Part II (1920), the biology and phylogeny. Three additional short supplements (1924, 1929a, 1929b) brought the number of crane flies known from New York to 318 species. More were added in subsequent publications, the most comprehensive being Alexander (1943 and 1965). We have not yet tallied a definitive list for New York but we did note that three of our species are apparently new additions to New York (marked as +NY in the Appendix). *Brachypremna dispellens* (Walker) seems to have migrated north to New York and *Atypophthalmus* sp. and *Achryolimonia neonebulosa* (Alexander) are recent introductions.

Atypophthalmus sp., female laden with mites; each wing ~6 mm long.

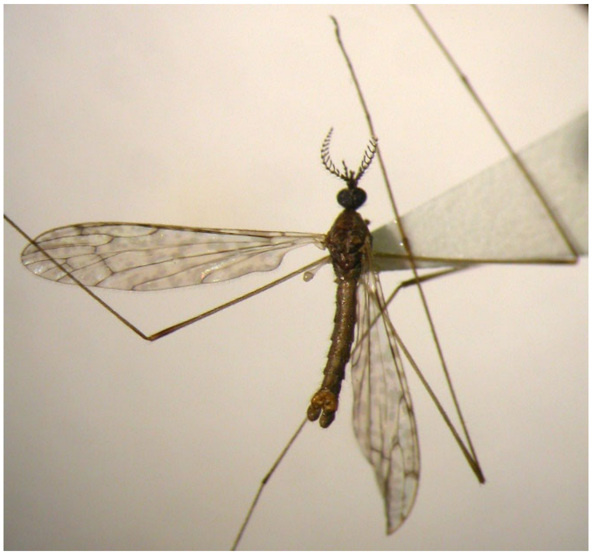
The crane flies of the Ottawa District have been documented by FB for over 50 years (but not so intensely in any one year), and with enormous input by the many entomologists at the Canadian Collection of Insects, past and present (unpublished report). The Ottawa District is a far larger area, approximately 7,854 km^2 compared to the 114 km^2 of the Town of Kent, and includes a distinctly boreal aspect. A total of 242 species have been found to date. Sixteen of the 70 species (23%) found in Kent have never been collected in the Ottawa District (marked with xO in the list in the Appendix). Not surprisingly, these 16 species have a more southern distribution. We share an introduced species with Kent, *Tipula* (*Tipula*) *oleracea*, one of two so-called European crane flies. The larvae eat roots of grasses and other plants and can damage turf.

The crane flies of Pennsylvania have also been well studied and 300 species are known for this state. Illustrated keys and information on habitats, etc. are available online (Young & Fetzner 2014). Only six species (8%) found in Kent do not appear in their list and these six seem to be our generally rare species (marked with xP).

We also compared our list to the 177 species collected during two years of intense inventorying of crane flies in the Smoky Mountains National Park, straddling Tennessee and North Carolina (Petersen et al. 2004). Its 521,000 acres makes it approximately 2,108 km^2. Twenty of our 70 species

(26%) do not occur there. Net collecting as well as a grid of various insect traps were employed for this endeavour. Malaise traps in particular draw in a remarkable number of specimens and these were emptied every two weeks even through the winter. Not all species are attracted to such traps. The terrain here is more mountainous, geologically older, and further west and south and so greater differences in the crane fly fauna would be expected.

Forty-four out of the 70 species (63%) were also found in the Ottawa District, Pennsylvania and in The Smoky Mountains National Park. These are species common to northeastern North America.




(left) *Rhipidia maculata*, one of the smallest crane flies, each wing ~6mm long.
(right) *Pedicia albivitta*, one of the largest crane flies, each wing ~25 mm long. Photo E. A. Herr.

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Literature Cited

Alexander, C.P. 1919. The crane-flies of New York. Part 1. Distribution and taxonomy of the adult flies. *Cornell University Agricultural Experiment Station, Memoir* 25: 767–993. [Reprinted 2015.]

Alexander, C.P. 1920. The crane-flies of New York. Part II. Biology and phylogeny. *Cornell University Agricultural Experiment Station Memoir* 38: 691–1133. [Reprinted 2015.]

Alexander, C.P. 1924. The crane-flies of New York: second supplementary list. *Bulletin of the Brooklyn Entomological Society* 19 (3): 57–64.

Alexander, C.P. 1929a. The crane-flies of New York: third supplementary list. *Bulletin of the Brooklyn Entomological Society* 24 (1): 22–29.

Alexander, C.P. 1929b. The crane-flies of New York: fourth supplementary list. *Bulletin of the Brooklyn Entomological Society* 24: 295–302.

Alexander, C.P. 1943. Family Tipulidae. In: Crampton, G.C. et al., Guide to the insects of Connecticut. Part VI. The Diptera or true flies of Connecticut. First Fascicle. *Bulletin of the Connecticut State Geological and Natural History Survey* 64: 196–486. [Reprinted 1966.]

Alexander, C.P. 1965. Family Tipulidae. *In*: Stone, A. et al., A catalog of the Diptera of America north of Mexico. *United States Department of Agriculture, Agriculture Handbook* 287: 16–90.

Oosterbroek, P. 2022. *Catalogue of the Craneflies of the World (CCW)*. Available from: https://ccw.naturalis.nl/ (accessed 27 April 2022).

Petersen, M.J., Gelhaus, J.K. & Bernard, E.C. 2004. New species and records of crane flies (Diptera, Tipuloidea) from Great Smoky Mountains National Park, Tennessee and North Carolina, U.S.A. *Transactions of the American Entomological Society* 130: 439–455.

Young, C.W. & Fetzner, J.W., Jr. (2014). *The Crane Flies (Diptera: Tipulidae) of Pennsylvania*. Version 2.0. Carnegie Museum of Natural History, Pittsburgh, Pennsylvania, USA. https://www.invertebratezoology.org/cranefly2. [Accessed: 27 April 2022].

Appendix. List of Tipulidae, Limoniidae, Pediciidae, Trichoceridae and Ptychopteridae collected in the Town of Kent, Putnam County, New York, in 2021.
C = common species (10 or more specimens collected; xO = not in Ottawa District; xP = not in Pennsylvania; xSM = not in Smoky Mountains; +NY = additions to New York State. Line separates families in the Tipuloidea from the related families: Trichoceridae and Ptychopteridae.

Superfamily Tipuloidea (70 species + 4 sp.?)

Family Tipulidae the long-palped flies (32 species + 2 sp.?)

Brachypremna dispellens (Walker)		xO			+NY
Dolichopeza (*Oropeza*) *carolus* Alexander	C				
Dolichopeza (*Oropeza*) *obscura* (Johnson)					
Dolichopeza (Oropeza) tridenticulata Alexander	C				
Dolichopeza (*Oropeza*) *venosa* (Johnson)				xSM	
Nephrotoma ferruginea (Fabricius)					
Nephrotoma macrocera (Say)	C				
Nephrotoma virescens (Loew)		xO			
Tanyptera dorsalis (Walker)					
Tipula (*Beringotipula*) *borealis* Walker	C			xSM	
Tipula (*Labiotipula*) *youngi* Alexander			xP	xSM	
Tipula (*Lunatipula*) *bicornis* Forbes				xSM	
Tipula (*Lunatipula*) *disjuncta* Walker				xSM	
Tipula (*Lunatipula*) *duplex* Walker					
Tipula (*Lunatipula*) *fuliginosa* (Say)					
Tipula (*Lunatipula*) *monticola* Alexander					
Tipula (*Lunatipula*) *valida* Loew					
Tipula (*Lunatipula*) sp.					
Tipula (*Nippotipula*) *abdominalis* (Say)					
Tipula (*Platytipula*) *paterifera* Alexander		xO		xSM	
Tipula (*Platytipula*) *ultima* Alexander	C	xO		xSM	
Tipula (*Pterelachisus*) *trivittata* Say					
Tipula (*Schummelia*) *hermannia* Alexander					
Tipula (*Tipula*) *oleracea* Linnaeus		xO		xSM	
Tipula (*Trichotipula*) *algonquin* Alexander			xP		

Tipula (*Trichotipula*) *oropezoides* Johnson					
Tipula (*Trichotipula*) *unimaculata* (Loew)					
Tipula (*Triplicitipula*) *perlongipes* Johnson		xO		xSM	
Tipula (*Triplicitipula*) *triplex* Walker					
Tipula (*Yamatotipula*) *caloptera* Loew				xSM	
Tipula (*Yamatotipula*) *sayi* Alexander				xSM	
Tipula (*Yamatotipula*) *tephrocephala* Loew					
Tipula (*Yamatotipula*) *tricolor* Fabricius					
Tipula sp.					

Family Limoniidae the short-palped crane flies (38 species + 1 sp.?).

Subfamily Chioneinae (12 species + 1 sp.?)

Atarba picticornis OstenSacken		xO			
Cheilotrichia (*Empeda*) *stigmatica* (Osten Sacken)					
Cladura flavoferruginea Osten Sacken	C				
Erioptera (*E.*) *septemtrionis* Osten Sacken				xSM	
Erioptera (*E.*) *straminea* Osten Sacken		xO		xSM	
Erioptera (*Mesocyphona*) *caliptera* Say					
Erioptera (*Mesocyphona) parva* Osten Sacken		xO			
Gnophomyia tristissima Osten Sacken	C				
Gonempeda nyctops (Alexander)		xO	xP		
Gonomyia (*Leiponeura*) *manca* Osten Sacken		xO			
Molophilus sp.					
Ormosia romanovichiana Alexander					
Ormosia rubella (Osten Sacken)				xSM	

Subfamily Limnophilinae (13 species)

Austrolimnophila toxoneura (Osten Sacken)					
Dactylolabis hudsonica Alexander		xO	xP		
Dicranophragma fuscovaria (Osten Sacken)				xSM	
Elephantomyia westwoodi Osten Sacken					
Epiphragma fasciapenne (Say)					
Epiphragma solatrix Osten Sacken		xO			
Pilaria quadrata (Osten Sacken)				xSM	
Pilaria tenuipes (Say)					
Prionolabis rufibasis (Osten Sacken)					
Prolimnophila areolata (Osten Sacken)					
Pseudolimnophlia contempta (Osten Sacken)					
Pseudolimnophila luteipennis (Osten Sacken)	C				
Shannonomyia lenta (Osten Sacken)					

Subfamily Limoniinae (13 species + 1 sp.?)

Achyrolimonia neonebulosa (Alexander)	C		xP	xSM	+NY
Atypophthalmus sp.	C	xO	xP	xSM	+NY
Dicranomyia (*Dicran.*) *divisa* (Alexander)		xO			
Dicranomyia (*Dicran.*) *frontalis* (Staeger)				xSM	
Dicranomyia (*Dicran.*) *stulta* Osten Sacken					
Dicranomyia (*Glochina*) *liberta* Osten Sacken	C				

Dicranomyia (*Idiopyga*) *ponojensis* Lundstrom			xSM	
Discobola annulata (Linnaeus)	C			
Geranomyia rostrata (Say)				
Metalimnobia immatura (Osten Sacken)	C			
Metalimnobia triocellata (Osten Sacken)				
Neolimonia rara (Osten Sacken)	C			
Rhipidia domestica Osten Sacken	C	xO		
Rhipidia maculata Meigen	C			

Family Pediciidae (3 species)
Pedicia albivitta Walker
Tricyphona (*Tricyph.*) *inconstans* (Osten Sacken)
Ula elegans Osten Sacken

Family Trichoceridae (6 species)

Trichocera annulata Meigen	C			
Trichocera bimacula Walker	C			
Trichocera brevicornis Alexander	C			
Trichocera fattigiana Alexander				
Trichocera garretti Alexander				
Trichocera salmani Alexander				+NY

Family Ptychopteridae (2 species)
Bittacomorpha clavipes (Fabricius)
Ptychoptera quadrifasciata Say

The Phantom Crane Fly, *Bittacomorpha clavipes,* Ptychopteridae. Photo by T. Hanrahan.

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Investigations on the Mycetophilidae of North Central Nevada

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Since 2017 I have been studying the Mycetophilidae in North Central Nevada, both to see what species are here, and for whatever I can discover about their biology. This area is ecologically diverse, with habitats ranging from valley floor desert up to nearly 10,000 feet in the higher mountains. During the first few years of my work on this group I put Malaise traps up in a variety of locations, keeping them in place for a few days or a week to see what I might catch. I put emergence traps up over animal burrows and moss, and collected adults that found their way into a rain barrel that I have at my house. In 2021 I put Malaise traps up in six different plant communities in the mountains, and left them there for the entire insect season. This ranged from March and April to early December. I hiked up to each of these traps every other week, went through the catches and pulled out all the mycetophilids, and during the winter identified them all down to genus using the key to mycetophilids in the Manual of Nearctic Diptera. Dr. Jukka Salmela and Dr. Woody Fitzgerald have kindly given me literature with which I can identify some of the specimens I have caught down to species, but with the state of mycetophilid taxonomy as I can see it, many of the specimens I have collected will probably remain unidentified to the specific level. And looking at the data I have so far, I think it will be necessary to identify the material down to species to really gain insight into what the catches have to say about these flies.

In 2022 I am continuing to investigate the mycetophilid fauna of different plant communities in the local mountains. In the Bloody Run Mountains I have three Malaise traps and one emergence trap up thus far. One of them, Aspen Spring (Fig. 1), is up in the same Aspen forest as last year, but higher up near the spring that feeds this forest. It is more open, and more diverse in terms of woody plants, a mix of willows and aspens, and much wetter. It is also on the edge of another section of this forest that leafs-out considerably later than the lower part, and it has a problem with wood boring beetles. The other Aspen grove in the Bloody Runs where I have traps, Dark Sister, is over a ridge from the first one (Fig. 2). It is much denser, the trees are bigger, and it is damper. It too was burned over in the nineties. I debated whether to put a trap up here, but leaf litter samples that I took in this forest and the Aspen Spring forest suggested that they were very different in terms of the arthropod faunas so I began studying this forest as well. The third community, Willow Thicket Spring (Fig. 3), is a dense thicket of willow, wild rose and wild currant surrounding a spring on the mountain side. Immediately surrounding this are large stones with moss. All three of these traps are at roughly the same elevation, about 5500 feet.

So far I have one Malaise trap up in the Santa Rosa Mountains. I wanted to sample a much larger Aspen forest, so I hiked up into an area full of springs and streams. This trap, Upper Singas (Fig. 4), is at an elevation of 7020 feet, and the forest, though made up of Aspens, is very different than the ones in the Bloody Runs. It too has been burnt over, but a decade more recently. The younger trees are heavily infested with the gall producing fly *Euhexomyza schineri* (Fig. 5). This infestation does not kill them, but seems to slow down their leafing out. I set this trap up on April 7, on a warm sunny day – I thought the bad weather was over, but I was wrong. For the next 40 days this area was rendered inaccessible due to snow and high winds. I was not able to return until May 17 – I didn't know if the trap would be there, or if it was, if there was still alcohol in the killing jar. But when I arrived at the site the trap was in good condition and loaded with mycetophilids that I identified as *Boletina* and *Garrettella*. *Garrettella* adults were flying around, I managed to catch a few of these.




Figure 1 (top left). Malaise trap at Aspen Spring – Bloody Run Mountains.
Figure 2 (top right). Malaise and emergence traps in Dark Sister Forest – Bloody Run Mountains.
Figure 3 (bottom left). Malaise trap at Willow Thicket Spring – Bloody Run Mountains.
Figure 4 (bottom right). Malaise trap in Upper Singas Aspen Forest.

Having explored a number of Aspen forests in this part of Nevada I have come to appreciate that they are all different – they have basic similarities, but a lot of variety in their individual character. Once the snow has retreated I will be putting up more traps, all at higher elevations. One will go up on Granite Peak at 9400 feet, one in an extensive Aspen Forest at 8500 feet, which has never been burned in recent times, and one in a *Veratrum californicum* marsh at the same elevation. There are some interesting plant communities on Buckskin Mountain at the northern end of the Santa Rosa's, these may have to wait until 2023. I lost a trap this spring in a wildfire, it was entirely consumed, it would be hard to overstate how unhappy I was about this.

So far in this study since 2017 I have collected 21 genera of mycetophilids. Eighteen of these were taken in the six study locations where I had traps up last year. Table 1 summarizes the genera I found in each of these six plant communities. *Boletina* was the only genus I found everywhere. *Rymosia*, *Garettella*, *Hadroneura* and *Coelosia* were found only in one site each. All the others were found in two or more sites.

In October of 2021 I found *Boletina* larvae and pupae in the leaf litter of one Chinese Elm tree in Winnemucca. I continued to find them in this leaf litter all through November, then they disappeared. I watched this site for adults from the beginning of November through to the beginning of May on a nearly daily basis, and kept track of adult activity and weather conditions. There was adult activity most of these days through the winter until April 15, when the last adults were seen. Numbers were usually small all through this period. I have looked through leaf litter in all the other locations where I have caught *Boletina* adults, but have seen no larvae or pupae in any but the Winnemucca location. When I put the trap up in the Aspen forest in the Santa Rosa Mountains on April 7 this year I took leaf litter back with me and went through it under the scope and ran a portion through the Berlese – no larvae or pupae, yet when I came back on May 17 there were 73 *Boletina* adults in the trap. Large early spring emergences of this genus are common everywhere these flies are found. Obviously they have more than one breeding habitat, and maybe there is more than one species.

Rymosia is rather common in the valleys here but did not show up much in the mountains in 2021. On May 12, 2022 there were mushrooms, as yet unidentified coming up at the edge of the spring in the Bloody Run Aspen forest, not 100 feet from where I had a trap. I collected some of these mushrooms and put them in a rearing chamber. On May 22 I saw adult mycetophilids in the chamber, I collected these and identified them as *Rymosia*. On May 26 when I revisited this site there were no mycetophilids in the trap head. This raises questions in my mind about the behavior of these insects, and questions about how thoroughly the Malaise traps I have out in these sites are really sampling mycetophilid populations. On April 27 a wildfire swept through a large cattail marsh along the Humboldt River where I had a Malaise trap, nothing was left of it except the poles, and the marsh was a blackened moonscape (Fig. 6). But on the night of May 16–17 a dry ice baited EVS trap I had out there to sample adult mosquitoes picked up a female *Rymosia*. Where it came from is a mystery. Despite the fire, I began to pick up insects in this EVS trap right after the fire, and I saw ants out in the burned over area. Did this *Rymosia* somehow survive through the fire and emerge afterward, or did it fly in from somewhere else?

The following are brief descriptions of the sites where I set Malaise traps during 2021:

Aspen – A small Aspen forest with some wild rose and chokecherry. It is located in a narrow gorge. There is a spring at the very top, and a larger one in the midsection. A small stream, which dries up in June, runs from these springs through the forest. An island in miles of sagebrush desert. Bloody Run Mountains.

Chokecherry – A dense stand of pure chokecherry, no streams or open bodies of water. Also an island in miles of sagebrush desert. Bloody Run Mountains.

Serviceberry – An essentially pure stand of serviceberry bushes, many acres, on the east side of an unnamed canyon in the south end of the Santa Rosa Mountains.

Singas – the trap was in a band of riparian vegetation consisting of willows and creek dogwood along Singas Creek in the Santa Rosa Mountains. The creek flows year around even in dry years.

Juniper – the trap was located in an extensive forest of pure juniper, old trees, in the East Range. Very dry.

Desert Peach – a pure stand of desert peach in a dry wash in the East Range. Very dry.

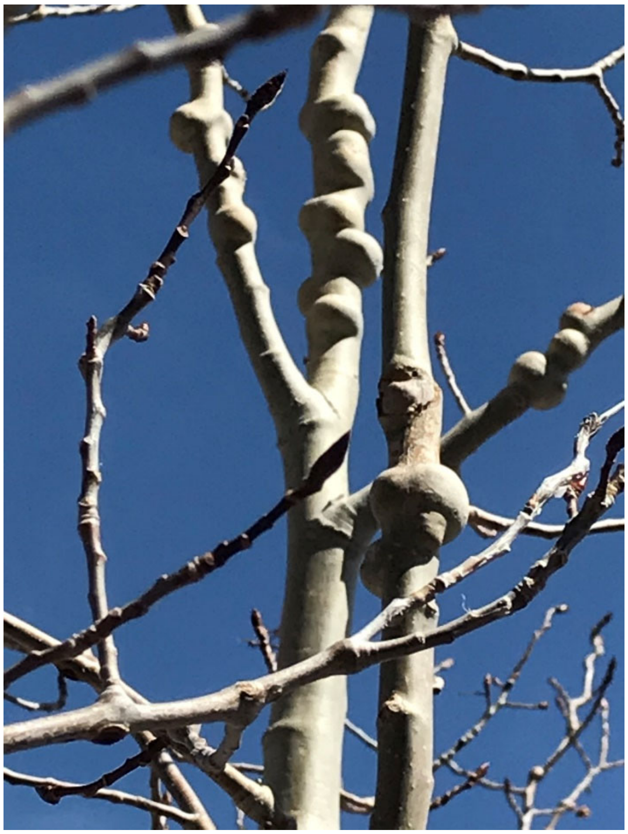

Figure 5 (left). Aspen galls at Upper Singas site.
Figure 6 (right). What is left of my trap near the Humboldt River after the Wildfire.

I have summarized the data I collected in 2021 from the six sites where I had Malaise traps in the tables on the following pages.

Table 1. Mycetophilid genera taken in Malaise traps in different plant communities during 2021.

Genera	**Aspen**	**Chokecherry**	**Serviceberry**	**Singas**	**Juniper**	**Desert Peach**
Anatella	x			x		
Boletina	x	x	x	x	x	x
Brevicornu	x	x		x		
Coelosia	x					
Cordyla	x	x	x	x		
Docosia	x	x	x	x	x	
Epicypta				x		
Exechia	x	x	x	x		
Garrettella			x			
Hadroneura			x			
Leia			x	x		
Megalopelma			x	x	x	
Mycetophila	x	x		x		
Orfelia			x		x	
Phronia				x		
Rymosia				x		
Sciophila			x	x		
Zygomyia	x			x		
TOTALS	9	6	10	14	4	1

Table 2. Seasonal distributions of the genera at various sites.

Anatella

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen			x	x						
Singas									x	

Boletina

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen	x	x								
Chokecherry	x	x								
Serviceberry			x	x						
Singas			x							
Juniper	x	x							x	X
Desert Peach									x	x

Brevicornu

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen		x	x							
Chokecherry		x								
Singas								x	x	x

Coelosia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen		x	x							

Cordyla

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen			x	x	x	x		x		
Chokecherry				x	x					
Serviceberry						x				
Singas				x	x	x			x	x

Docosia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen		x	x	x						
Chokecherry		x	x	x						
Serviceberry			x	x						
Singas		x							x	x
Juniper	x	x								

Epicypta

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Singas								x	x	

Exechia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen								x		
Chokecherry	x	x		x	x	x		x		
Serviceberry								x	x	
Singas							x	x	x	x

Garrettella

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry			x							

Hadroneura

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry			x							

Leia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry						x				
Singas						x				

Megalopelma

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry					x					
Singas							x			
Juniper			x							

Mycetophila

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen		x	x							
Chokecherry	x	x	x							
Singas							x	x	x	x

Orfelia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry			x	x						
Juniper		x								

Phronia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Singas								x	x	

Rymosia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Singas								x	x	

Sciophila

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Serviceberry				x	x					
Singas								x	x	

Zygomyia

Location	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aspen									x	X
Singas							x	x	x	x

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Superiority of the Sante Traps Malaise trap design over the Bugdorm EZ Malaise trap

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In the interest of full disclosure, Sante Traps is a company belonging to the first author's ex-wife.

The design of the Malaise trap has been fiddled with for over almost 100 years, with various improvements and modifications that may or may not increase collecting efficiency. The two most common models are the Townes lightweight style and the larger (definitely not lightweight) Gressitt traps (Gressitt and Gressitt 1962, Townes 1972). Recently, some alternative designs have been proposed (reviewed by van Achterberg 2009), including the "Bugdorm EZ Malaise trap." This trap has the virtue of not needing any poles other than the lightweight flexible tent poles that come with it, but is there any reason to expect better catches from this model, especially when compared to the popular Sante Traps model?

We ran some tests for three weeks, and although we didn't conduct formal statistical analyses, the results were convincing enough for us. We ran the test in Forest Falls (~6,000 meters), San Bernardino County, California, starting in early August, so it was dry and warm. The trap on the left in Figure 1 is in the optimal position because the head is facing south.

We showed Figure 1 (A and B) to a colleague and they suggested that the Sante trap was higher than the Bugdorm trap and potentially casting a shadow on the Bugdorm bottle. We corrected that as follows: the image Figure 3A shows the corrected trap setup at the beginning of week 3. The Bugdorm trap is on the left and the Sante trap on the right. The Bugdorm trap is in the optimal position now with the head facing towards the south. This was the position in which the Sante trap did so well in week 2. The Bugdorm trap also has the bottle in the optimal position, higher than the Sante bottle so the latter does not shade it.

Figure 1. **A**. The setup for week 1 with Bugdorm on the left and Sante on the right. **B**. The traps were rotated 180 degrees, now the Sante trap is on the left. You can see the results of both weeks in Figure 2.

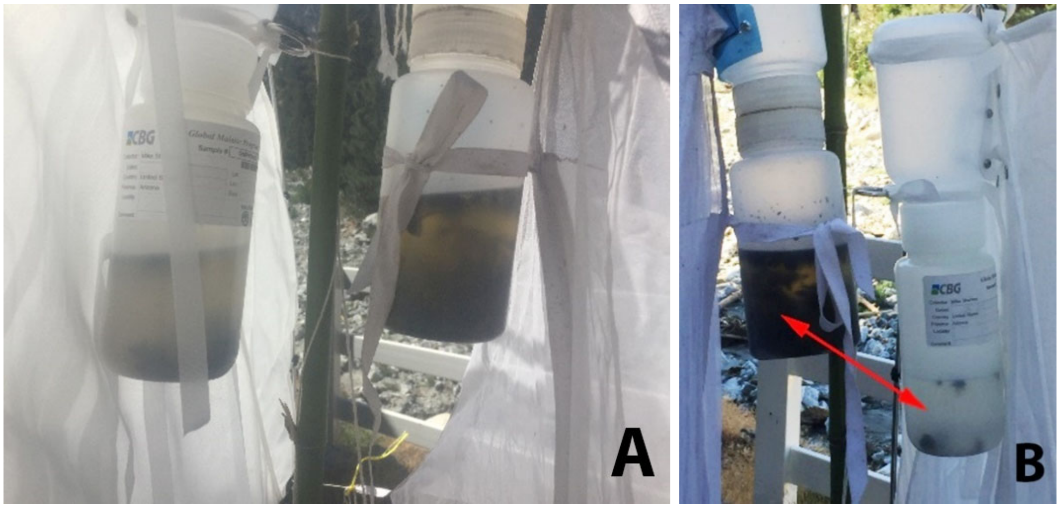

Figure 2. **A**. Week 1 catches. The Sante bottle is on the right. **B**. Week 2 catches. The Sante bottle on the left.

The image in Figure 3B shows the catch for each trap at the end of week 3. Since these are hard to see we emptied them both into white pans (Fig. 4). To see what this means for diversity we pulled the braconids (we know, not flies, but the same principle holds) from both traps. See below (Fig 5).

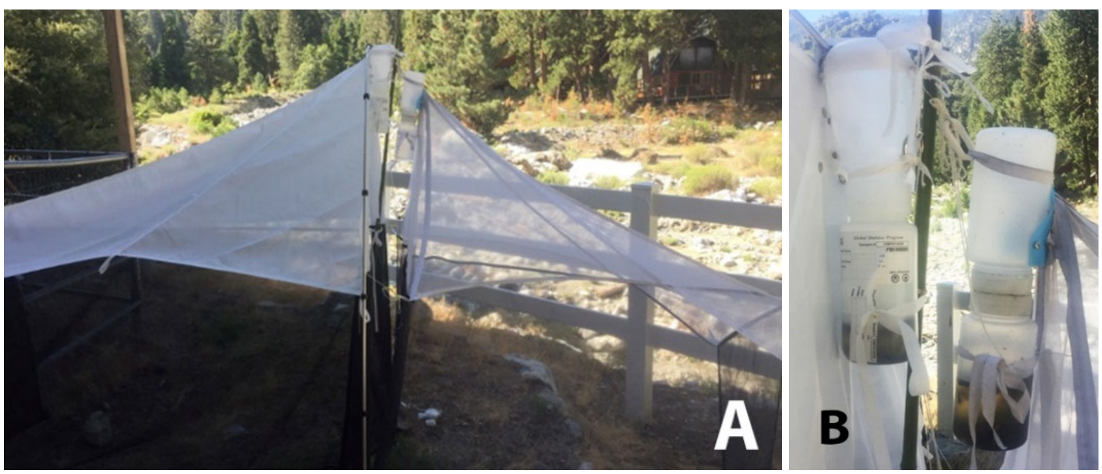

Figure 3. **A**. Week 3 revised setup. **B**. Week three catches. Bugdorm is on the left, Sante on the right.

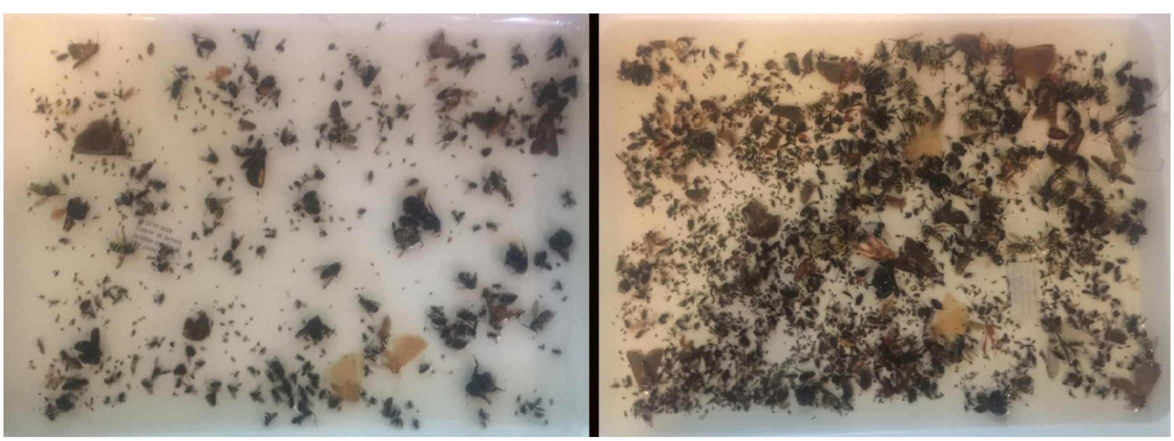

Figure 4. Catch from week 3. Bugdorm is on the left and Sante is on the right, images are the same scale.

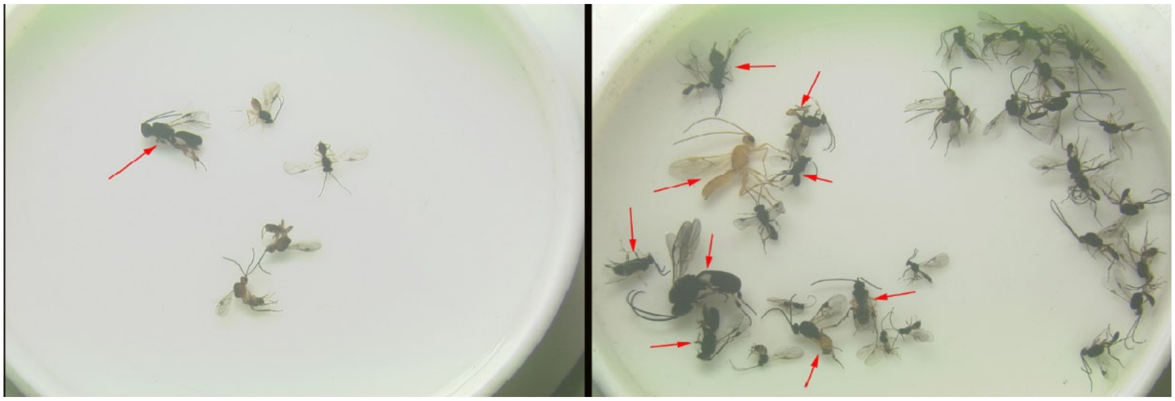

Figure 5. Catch from week 3. Bugdorm is on the left and Sante is on the right, images are the same scale.

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.

References

Gressitt, J.L. & Gressitt, M.K. 1962. An improved Malaise trap. *Pacific Insects* 4: 87–90.
Townes, H. 1972. A light-weight Malaise trap. *Entomological News* 83: 239–247.
van Achterberg, K. 2009. Can Townes type Malaise traps be improved? Some recent developments. *Entomologische Berichten* 69: 129–135.

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HISTORICAL DIPTEROLOGY

Zide Fan (1923–2022)

Dong Zhang[1], Thomas Pape[2], Weibing Zhu[3] & Liping Yan[1]

[1] School of Ecology and Nature Conservation, Beijing Forestry University, China

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Prof. Zide FAN, an Honorary Member of the International Congresses of Dipterology, passed away on 28 February 28, 2022 in Shanghai Sixth People's Hospital, aged 99.

Zide Fan (1923–2022). [Photo from 2008]

Zide Fan was born in Wujiang, Jiangsu Province in 1923. He graduated from the Department of Biology of Central University in 1944. Later, he served as a Professor of Shanghai Institute of Entomology, Chinese Academy of Sciences (CAS). Since 1953, he was engaged in research on flies, mainly focused on Anthomyiidae, Muscidae, Calliphoridae and Sarcophagidae. He has established 5 new tribes (one of which has been regarded as subfamily), 18 new genus-group taxa, and more than 300 new species-group taxa. He has published 147 papers and five monographs, including the influential '*Key to the Common Flies of China*', Fascicle 37 of '*Economic Insect Fauna of China*, *Diptera: Anthomyiidae*', and Vol. 6 of '*Fauna Sinica*, *Diptera: Calliphoridae*'. Prof. Fan's books continue to be important references for dipterists worldwide, and they won the Third Prize of Natural Science Award of Chinese Academy of Sciences. Prof. Fan promoted the systematics of flies and trained a large number of professional personnel for China.

Prof. Fan started his career doing taxonomy of fly larvae and implemented investigations of the breeding habits of flies in residential areas in China. His sound basic research promoted the development of medical and forensic entomology in China, and his contributions to the establishment of comprehensive policies for controlling populations of medically important flies were instrumental for the prevention and control of fly vectors following the devastating Wenchuan earthquake in 2008.

Prof. Fan hosted the symposium '*Medical and Veterinary Entomology*' at the XIX International Congress of Entomology in Beijing in 1992, for which he received an honorary certificate. In 1993, he was elected as a member of the London-based Royal Society of Tropical Medicine and Hygiene.

In August 2010, at the 7th International Congress of Dipterology in Costa Rica, he was elected as an Honorary Congress Member. In 2014, Prof. Fan was among five scientists that were awarded the Lifetime Achievement Award of the Entomological Society of China. He served as a member of the editorial board on *Zoological Systematics*, *Entomotaxonomia*, *Zoological Research*, and *Contributions from Shanghai Institute of Entomology*.

Prof. Fan was a remarkable Chinese scientist, entomologist and dipterist, and his everlasting legacy will continue to motivate and inspire!

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PHILAMYIANY

Diptera on stamps (3): Ephydroidea

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This third contribution to my series “Diptera on stamps” deals with the Ephydroidea, comprising the Ephydridae and Drosophilidae. In addition, one stamp is known showing the so-called “Terrible Hairy Fly” *Mormotomyia hirsuta* Austen, 1936 (Mormotomyiidae) from Kenya (2011) but this was probably only a private issue and therefore is not considered further (see Part 1 for basis of inclusion).

Drosophila melanogaster was depicted twice (KM 346, SE 1572) on stamps celebrating the Nobel Prize winner Thomas Hunt Morgan (1866-1945), who worked extensively with this fly. Remarkably, two species of Drosophilidae (TA 418, TA 535) and one of Ephydridae (TF 882, TF 959) were chosen to represent local endemic species from the species-poor islands of Terres Australes et Antarctiques and Tristan da Cunha. Finally, the obvious and pretty *Drosophila heteroneura* was included on a sheet of 20 insects selected to illustrate the diversity of animals in 1999 (PW 1467).

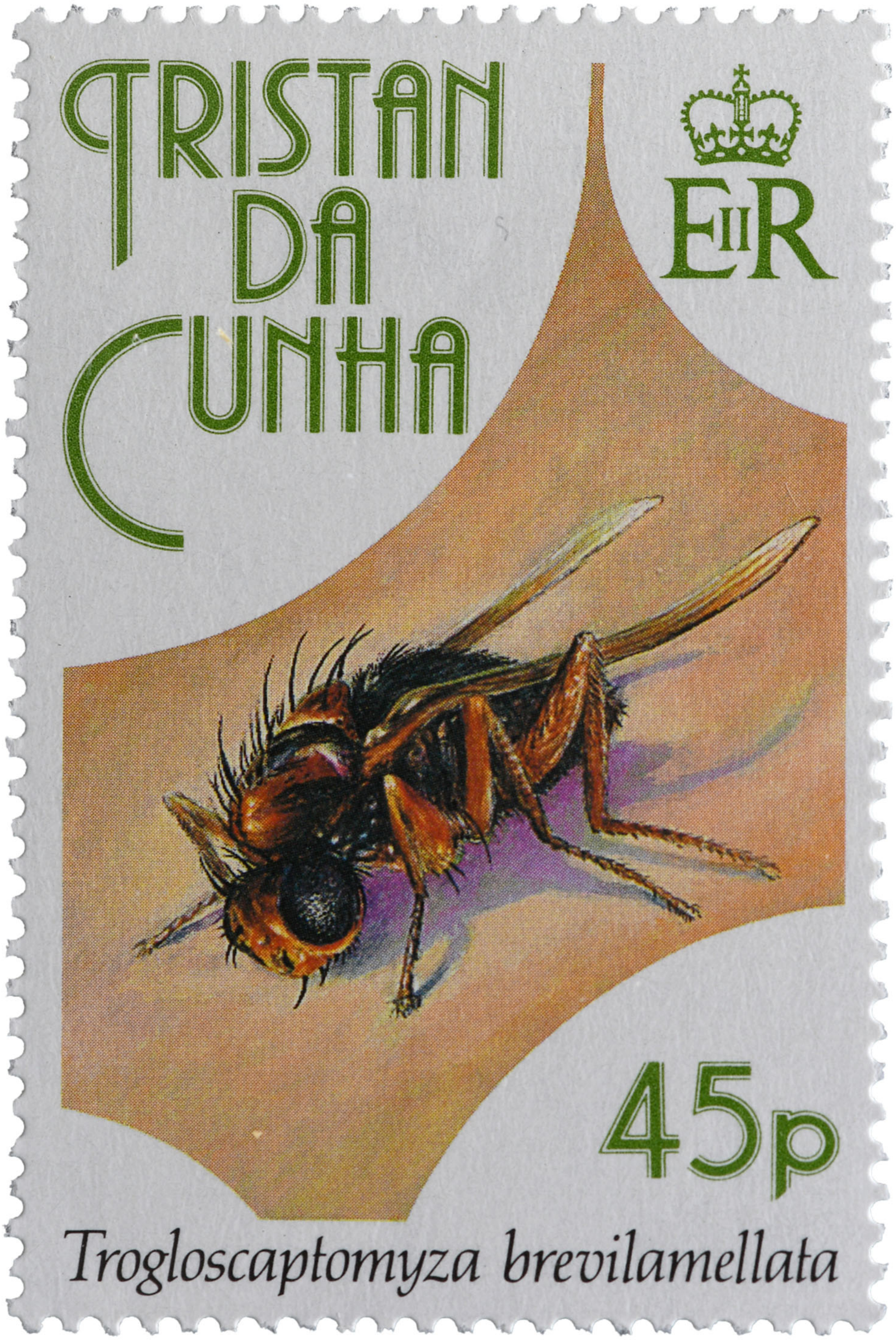
For each stamp I have provided the country and year of issue, title of stamp, title of stamp series (where available/relevant), face value, Michel number and stamp number (the latter both copied from https://colnect.com/).

Ephydridae

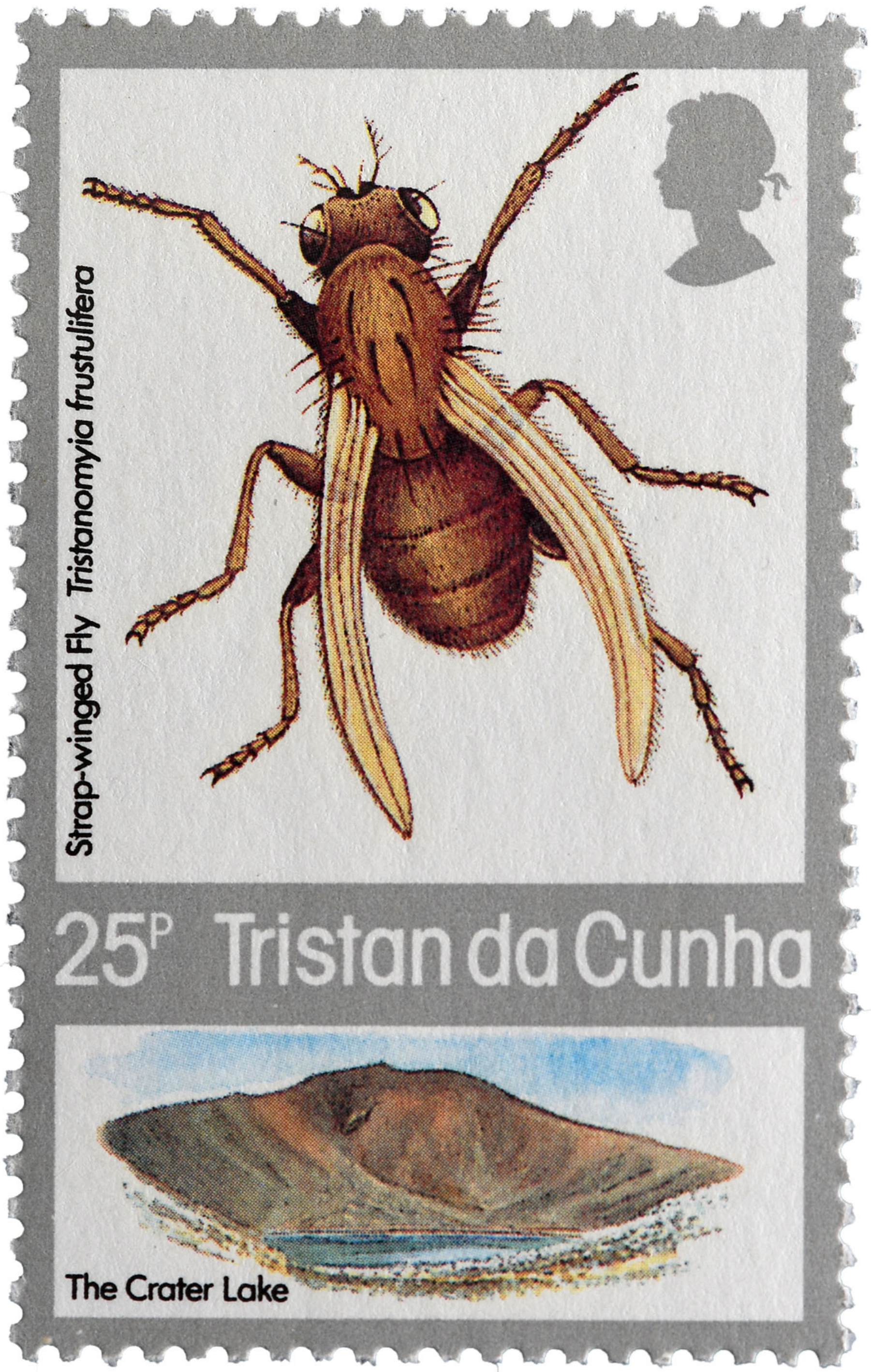
***Amalopteryx maritima* Eaton, 1875 – France [Terres Australes et Antarctiques]**
2017: *Amalopteryx maritima*, 1.55 Euro. – Michel number: TF 959; stamp number: -.

***Amalopteryx maritima* Eaton, 1875 – France [Terres Australes et Antarctiques] 2015**: Reservé Naturelle, Terres Australes Françaises, *Amalopteryx maritima* [Insects and Spiders of Crozet Island], 0.66 Euro. – Michel number: TF 882; stamp number: -.

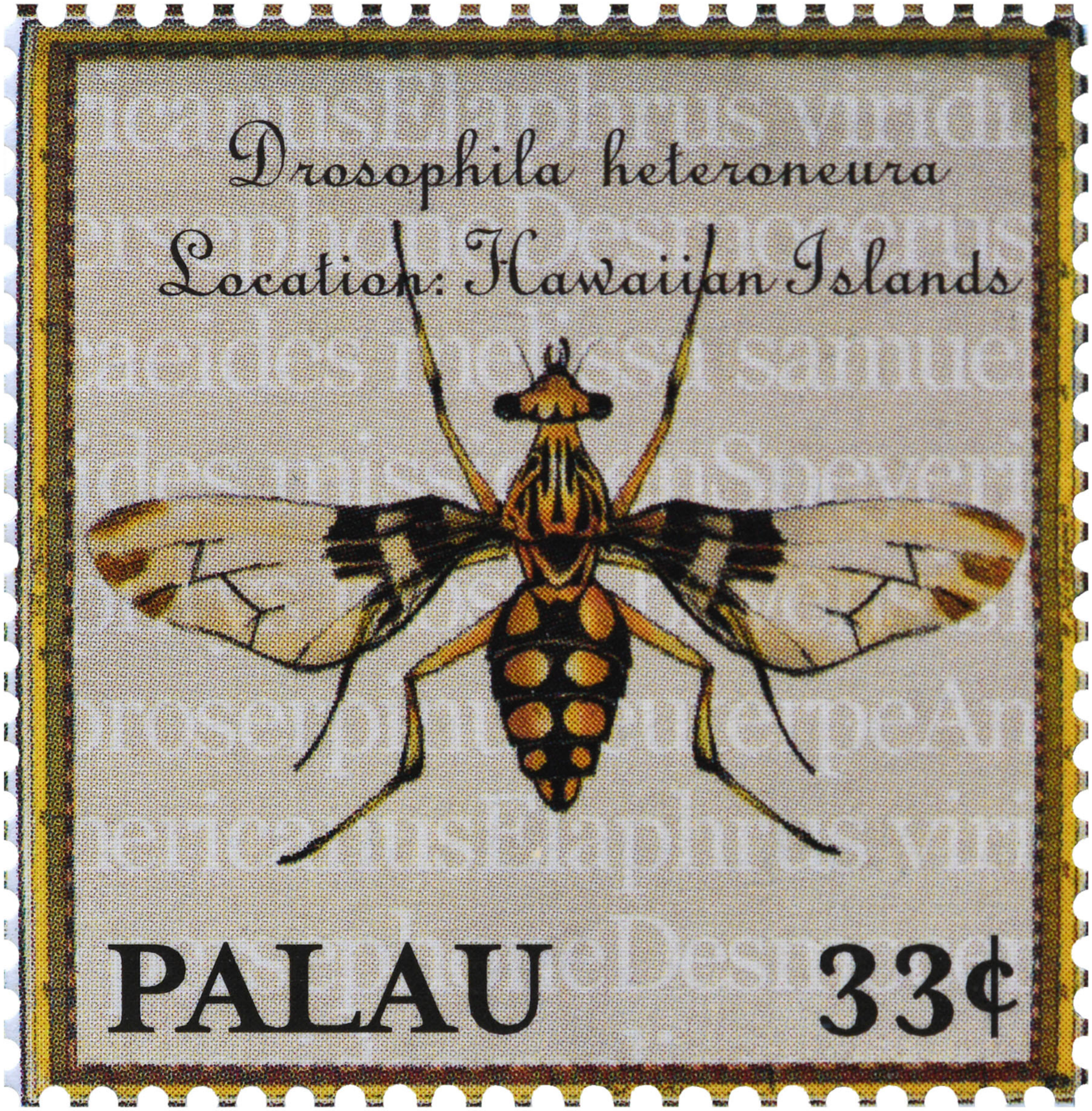
Drosophilidae



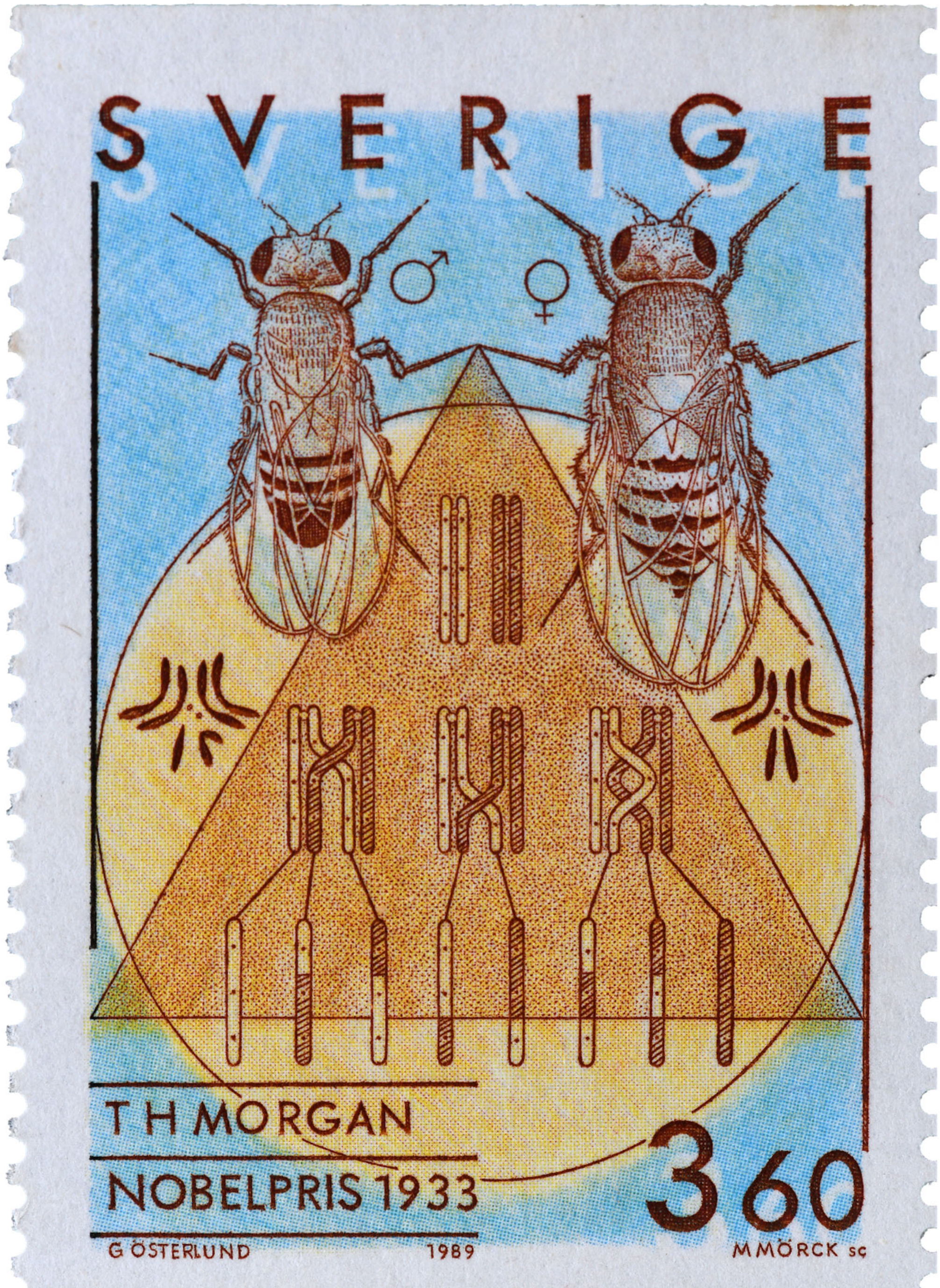
***Scaptomyza brevilamellata* (Frey, 1954) – Britain [Tristan da Cunha] 1993**: *Trogloscaptomyza brevilamellata* [Insects], 45 Saint Helena penny. – Michel number: TA 535; stamp number: TA 522.



***Tristanomyia frustulifera* Frey, 1954 – Britain [Tristan da Cunha] 1987**: Strap-winged Fly *Tristanomyia frustulifera*, The Crater Lake [Indigenous Flightless Species and Habitats], 25 Saint Helena penny. – Michel number: TA 418; stamp number: TA 405.



***Drosophila heteroneura* (Perkins, 1910) – Palau 1999**: *Drosophila heteroneura*, Location: Hawaiian Islands [Earth Day 1999, Pacific Insects], 33 United States cent. – Michel number: PW 1467; stamp number: PW 506b.



***Drosophila melanogaster* Meigen, 1830 – Sweden 1989**: TH Morgan, Nobelpreis 1933 [Nobel Prize Winners - Physiology or Medicine], 3.60 Swedish krona. – Michel number: SE 1572; stamp number: SE 1772.

***Drosophila melanogaster* Meigen, 1830 – Comoros 1977**: R. Koch 1905, T. Morgan 1933, A. Fleming 1945, P. Müller 1948, A. Waksman 1952 [Nobel Prize Winners], 30 Comorian franc. – Michel number: KM 346; stamp number: KM 254.

Acknowledgement
Thanks to David Clements who checked the manuscript! Any comments concerning either the identification of the Diptera shown or references to overlooked stamps would be very welcome!

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Diptera Trading Cards and Trade Cards (II), Anthropomorphism

Stephen D. Gaimari

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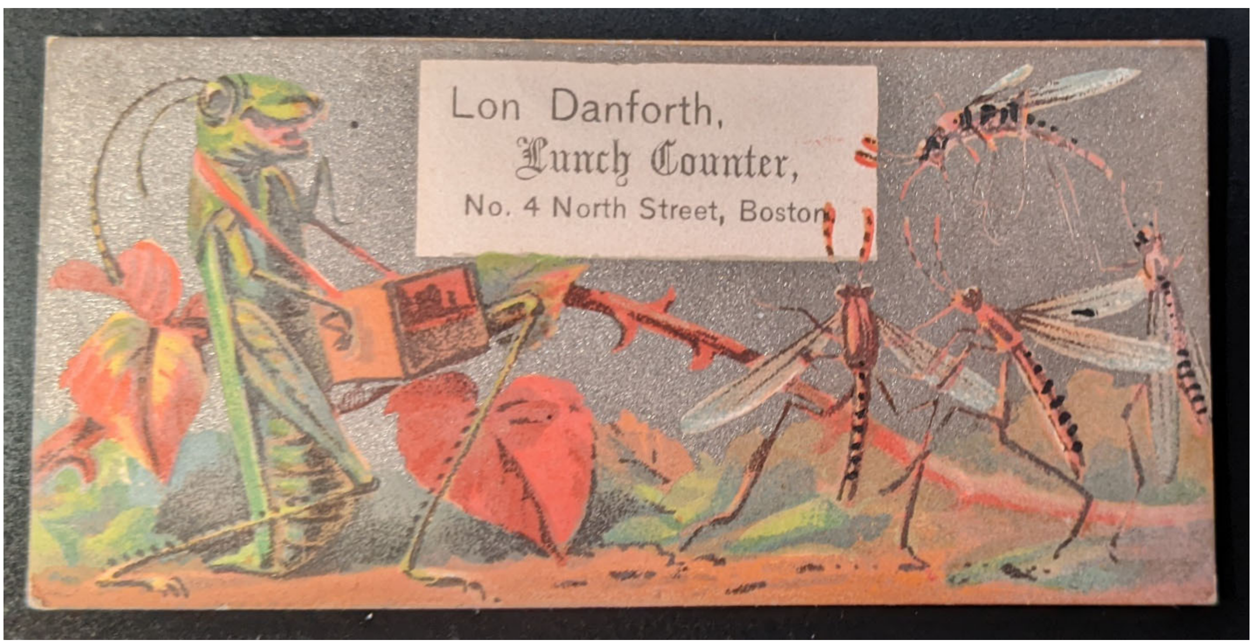
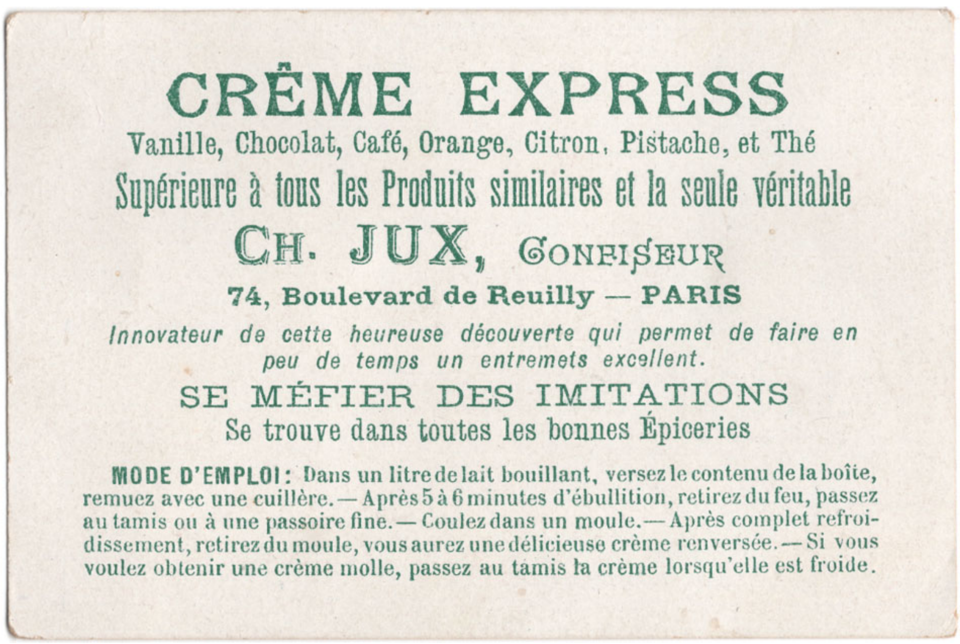
As part of my series on collecting dipterocentric (or at least featuring flies) trade cards, I have two to show in this issue. These are of a card-type for which I have very few representatives – that is, those anthropomorphizing flies. There are two main types of anthropomorphic cards – 1) those with insects dressed in human clothes or engaging in human activities, and 2) those with humans depicted with insect characteristics, such as wings and antennae. There are many such cards for insects as a whole, but I have found very few featuring flies.

The following card (front only, as the back is blank) from the 1880s, measuring 9 x 4 cm, anthropomorphizes the flies by having them acting like humans. The card is an advertisement for Lon Danforth Lunch Counter in Boston, Massachusetts, USA. The chironomid midges (or what I believe to be chironomids, although there appear to be elongated mouthparts like a mosquito) seem to be looking forward to whatever lunch is being brought to them by the grasshopper!



The chromolithographic card on the next page is a Victorian card, measuring 11 x 7.4 cm, with children (who are often the focus of this kind of card) with antennae and wings, and even more significantly with three pairs of legs sticking out of their backs (in addition to the human arms and legs). They do correctly have only one pair of wings, and are identified as being closely related to mosquitoes ("Cousins moustiques") in the Histoire Naturelle Enfantine (children's natural history) series of cards – this being #6 in the series. This is an advertisement for a Parisian confectionary called Crème Express. The more common version of this card advertises for Liebig, which I discussed in the last issue of *Fly Times* as being one of the most famous and prolific companies distributing such trade cards – the odd thing is that Liebig cards are usually proprietary, so it is unusual to find such cross-overs. The Liebig card is dated 1899.



Subsequent articles in this series will cover other types of cards, and maybe we will circle back to anthropomorphic cards at some point, as they are among the more interesting ones.

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Meeting News

10th International Congress of Dipterology (ICDX),
16–21 July 2023 in Reno, Nevada, USA

Shaun L. Winterton, Stephen D. Gaimari & Martin Hauser

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Preparations continue for the ICDX, which is now just a little more than 13 months (or 56 weeks) away! Please visit the website (https://dipterists.org/icdx/) regularly for updates and remember to lodge your interest in attending. The portal for accepting Early Registrations will be ready very soon, along with the portal for abstract submissions after registering.

The Congress website already has plenty of useful resources as you prepare for the meeting, including information on:

- Visa Requirements (https://dipterists.org/icdx/visa.html)
- Flights and travel (https://dipterists.org/icdx/travel.html)
- Competitions (https://dipterists.org/icdx/competitions.html)
- Permits and collecting (https://dipterists.org/icdx/collecting.html)
- Visiting insect collections (https://dipterists.org/icdx/collections.html)
- Tours and tourist information (https://dipterists.org/icdx/tours.html)
- Accommodation* (https://dipterists.org/icdx/accommodation.html)

* A note about Accommodations! We have secured amazing concessionary arrangements with the venue based on our contracted room block. So, to keep costs as low as possible, we are counting on everyone booking their rooms through the venue. Their rates are already significantly reduced and reasonable for the area, and we are strongly discouraging the use of other accommodations!

We are pleased to announce our banquet speaker and plenary speakers for the Congress!
Our banquet speaker will be Dr. Erica McAlister (The Natural History Museum, London, UK), and following is our list of plenary speakers:

- Prof. Rudolf Meier (Museum für Naturkunde, Berlin, Germany)
- Prof. May Berebaum (University of Illinois, Illinois, USA)
- Dr. David Grimaldi (American Museum of Natural History, New York, USA)
- Charley Eiseman (Massachuesetts, USA)
- Prof. Fiona Hunter (Brock University, Ontario, Canada)

We are planning a diverse selection of symposia, and the following are already confirmed. Please contact our Symposium coordinator Martin Hauser if you are interested in contributing to one of the symposia below or if you wish to propose and organize a symposium.

Confirmed symposia: organizer(s)

Advances in Afrotropical Dipterology: Ashley Kirk-Spriggs & Brad Sinclair
Advances in Diptera paleontology: Guilherme Cunha Ribeiro & Vladimir Blagoderov
Advances in lower Brachycera systematics and taxonomy: Xuankung Li & Diego Fachin
Biodiversity surveys and collecting methods: Marc Pollet
Culicomorpha: Brian Wiegmann & John Soghigian
Diptera phylogenomics: Jessica Gillung
Diptera pollinators: Andrew Young
Dipterans as parasites and vectors: Tamara Szentivanyi
Empidoidea: Marija Ivković
Syrphoidea: Jeff Skevington & Ximo Mengual
Systematic & Ecology of Bibionomorpha: Chris Borkent
Taxonomy and phylogeny of Asilidae – honoring Eric Fisher and his impact on understanding the Nearctic & Neotropical fauna: Torsten Dikow
Tephritoidea of economic importance: Severyn Korneyev

We look forward to seeing everyone in Reno in 2023!

To keep up to date, we encourage you to keep an eye on the Congress website, and if you have not done so already, to join the dipterists mailing list https://lists.dipterists.org/mailman/listinfo/dipterists to keep up with all the latest Congress and other dipterological news.

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S.W. Williston Diptera Research Fund – ICDX graduate student travel awards

Torsten Dikow & S.W. Williston Fund committee

Department of Entomology, National Museum of Natural History, Smithsonian Institution,
PO Box 37012, MRC 169, Washington, DC 20013-7012, USA; DikowT@si.edu

S.W. Williston Diptera Research Fund

The S.W. Williston Diptera Research Fund is a small Smithsonian Institution administered endowment fund established for the *increase and diffusion of knowledge about Diptera*. To this day, the fund has supported the travel of graduate students and dipterists to the *International Congresses of Dipterology* and to the USNM for collections-based research as well as attendance at *Fly School* and fieldwork.

This year, the Williston Fund will support the attendance of up to 8 graduate students or recently graduated dipterists (graduation in 2022 or 2023) who are either presenting a poster or oral presentation at ICDX in Reno, Nevada, USA (16–21 July 2023, https://dipterists.org/icdx/) with US$1,500 each. This special competition is made possible through our annual endowment funds.

https://naturalhistory.si.edu/research/entomology/opportunities/williston-diptera-research-fund

The requirements for application are minimal: contact Torsten Dikow as a representative of the Williston Fund committee with a short summary of why you plan to attend ICDX and a research project you plan to present at the congress.

1. summarize your research goals into a short proposal in PDF format (1–2 pages maximum)
2. itemize your budget in the proposal PDF (anticipated transportation costs, per diem costs for lodging and food, and any other items)
3. attach a current CV

Please send the complete application materials in PDF format to Torsten Dikow (DikowT@si.edu) by **1 December 2022**. Please note that every awardee will need to comply with the rules of the Smithsonian Institution regarding travel and reimbursements, which require several forms to be filled out prior to any travel.

Please consider donating to this endowment fund to support the increase and diffusion of knowledge about Diptera and particularly the research and travel of a new generation of dipterists.

The Williston Fund is administered by a committee of at least three members, two of whom (the majority) must be systematists actively working on Diptera, and one who must be a scientist affiliated with, but not necessarily employed by, the Smithsonian Institution (for example, a dipterist of the United States Department of Agriculture Systematic Entomology Laboratory (SEL)). The current committee consists of: Allen Norrbom, Woogie Kim, and Torsten Dikow.

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OPPORTUNITIES

Associate Insect Biosystematist,
CDFA Plant Pest Diagnostics Laboratory,
Examination Bulletin

Stephen D. Gaimari & Shaun L. Winterton

Plant Pest Diagnostics Branch, California Department of Food & Agriculture
3294 Meadowview Road, Sacramento, California 95832, USA;
shaun.winterton@cdfa.ca.gov, stephen.gaimari@cdfa.ca.gov

First, this is not an announcement of a *current vacancy* in the CDFA Plant Pest Diagnostics Center. **However, it IS the critical first step to applying for any of our upcoming vacancies!**

Applying for jobs in California state service has several steps. Among the first steps is to "get on the list" for a classification (in this case, Associate Insect Biosystematist). That is, to make yourself eligible for when the time comes to apply for a vacancy. These eligibility lists have a life of one to two years. We can ONLY hire off of these lists, so it is an all-important first step for any vacancies that arise in that time period. There is no restriction to taxonomic expertise, except for a person being an entomologist. But when vacancies occur, we will be targeting particular taxonomic specialties, possibly including Diptera.

So, we have posted the Official Examination Bulletin for the class Associate Insect Biosystematist at https://www.calcareers.ca.gov/JOBSGEN/2FA15.PDF. As an "Open" examination, any qualified entomologists (including arachnologists) with a specialty in taxonomy and systematics are encouraged to apply.

The class specification is at https://www.calhr.ca.gov/state-hr-professionals/pages/0537.aspx.

The final filing date is 12 August 2022.

Please reach out to Steve or Shaun, or any of our other systematists, for more about our Entomology Lab (https://www.cdfa.ca.gov/plant/ppd/entomology.html) and about the Plant Pest Diagnostics Center as a whole (https://www.cdfa.ca.gov/plant/ppd/).

CALIFORNIA DEPARTMENT OF FOOD & AGRICULTURE

PLANT PEST DIAGNOSTICS CENTER

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Insect Biodiversity Data Postdoctoral Fellowship

Brian V. Brown

Department of Entomology, Natural History Museum of Los Angeles County,
900 Exposition Blvd, Los Angeles, California, 90007, USA

Location: Natural History Museum of Los Angeles County (LACM)
Duration: 2 years
Supervisors: Dr. Brian Brown (LACM)
Dr. Melissa Guzman (University of Southern California)
https://nhm.org/careers-our-museums/careers-natural-history-museum

The Insect Biodiversity Initiative (IBI) is a state funded effort to raise scientific understanding of California's insects, through improving the curation of collections, obtaining DNA barcodes of all California species, obtaining detailed distribution information, and hiring underrepresented groups to participate in the scientific process.

Although some of these goals are aspirational (such as obtaining barcodes of ALL species), we hope to greatly increase our knowledge of insects throughout the state, by sampling existing collections for known species and through making collections of new specimens to obtain many of the smaller, poorly-known, and as of yet uncollected species. These new collections, in particular, are expected to uncover many species new to science, as well as new records of species described from elsewhere. These collections will also provide data on species distributions, endemicity, and potential threatened populations, as well as levels of insect biomass, to inform studies on the worldwide insect decline.

We seek a dynamic leader to head the program of new sampling for IBI. This person would lead a team of three technical staff, in planning, executing, and analyzing the results of this effort. Additionally, he/she will be responsible for coordinating new collections made by other institutions in California, to ensure a cohesive effort. Additionally, this person will lead the LACM portion of the "Existing Collections" group, interacting with other museums and entomology departments to provide specimens for the overall initiative from existing collections.

Primary Duties

- help plan and lead execution of a statewide 2-year survey of California insects, with the intention of discovering new species and undersampled habitats using DNA barcoding.
- analyze results of survey, including climate data, spatial data, taxonomic data
- manage large amounts of DNA sequence data
- help oversee technical staff
- engage in both museum and field entomology associated with this project
- To perform effectively in this position, the incumbent is expected to have:
- PHD in Biology, Zoology, or other natural science related discipline
- Strong data analysis skills, including proficiency in programming, spatial analysis, databases.
- Experience working with natural history collections
- Some experience with, and knowledge of, insects is desired, not required
- Strong organizational and interpersonal skills

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DIPTERA ARE AMAZING!

Following is a short series showing larval activity (and a larva) of Therevidae, shot by Shaun Winterton on K'Gari (a World Heritage-listed island, aka Fraser Island), off the coast of Queensland, Australia. Larvae come near to the surface of the sand at night and at times break the surface. The overnight dew hardens the sand and documents their travel over the course of the night in search of prey, such that an entire night's movements can be traced the following morning. During the daytime, the sand heats up and dries out. Larvae can be found deeper in the sand at this time, often around the bases of plants. Although not reared, the bottom photo, also taken by Shaun, is a species (*Anabarhynchus maritimus*) also found on the beach on this island.




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SOCIETY BUSINESS

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

Three documents are here provided for the record. They are:

1) The approved minutes of the annual meeting of Directors, held 19 December 2021. Prepared by Secretary Shaun Winterton. (5 pages).
2) The financial statement as part of the minutes of the 2021 annual meeting of Directors, updated at year end to reflect the full fiscal year (calendar year). Prepared by Treasurer Chris Borkent. (1 page).
3) The financial statement as part of the minutes of the 2020 annual meeting of Directors, updated at year end to reflect the full fiscal year (calendar year). Prepared by Treasurer Chris Borkent. (1 page).

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

Directors
Stephen D. Gaimari
Martin Hauser
Shaun L. Winterton
Christopher J. Borkent

Officers
Stephen D. Gaimari, President
Martin Hauser, Vice President
Shaun L. Winterton, Secretary
Christopher J. Borkent, Treasurer
Jessica P. Gillung, Meeting Chairperson
Jon K. Gelhaus, Field Meeting Chair
Andrew D. Young, General Meeting Chair

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North American Dipterists Society

DIRECTOR'S MEMO

March 16, 2022

Prepared and filed by:Shaun L. Winterton, Secretary
North American Dipterist Society

Minutes of Directors Meeting
Held Sunday, 19th December, 2021, 2pm at 4304 New York Avenue, Fair Oaks, CA, 95628.
Presiding: Steve Gaimari
Secretary: Shaun Winterton
Attendance: Martin Hauser (MH), Steve Gaimari (SDG), Chris Borkent (CB), Shaun Winterton (SLW)

Item 1. *Call for corrections/emendation of previous meeting minutes* (14th December, 2020):

- After discussion, acceptance as written moved by CB, seconded by SLW. Carried unanimously.

Item 2. *Treasurers report (CB):*
(filed in the main office and Secretary's record; see Appendix 1):

- Total 2021 income was USD$22,977.13 from donations and membership dues. Total 2021 expenses were USD$902.91. The balance of accounts is USD$23,858.58 as of Dec 19, 2021.
- Submitted by CB noted that for reporting purposes, membership values will be reported as well as donations. Moved by SLW, seconded by MH.
- Note, the final 2021 budget summary was provided by CB after the meeting, on 02/28/2022, with total 2021 income of USD$23,271.87 and total 2021 expenses of USD$902.91, and the final 2021 balance of accounts is $24,153.32.

Item 3*. Previous and supplemental business*
1) Review of Unanimous Written Consents executed in 2021:

- Code review for Society website (10th Feb. 2021)
- Purchase of Virtual Private Server (VPS) contract (15th Mar. 2021)
- Entomological Society of America Annual Meeting registration and presentation (30th August 2021)
- Change in date and time for the 2021 Annual Meeting of the Society (8 December 2021)

2) Action items from previous meeting (14th December 2020):

- Continued to use iATS Payments for all incoming Society online financial transactions, including membership, donations and meeting registrations.
- Agreed on continued use of Mark Zivkovic (Pasquesi-Sheppard LLC) for tax preparation for the 2021 tax year. He provided advice for 2020 tax documents at no charge. Tax filing documents provided by SDG.
- Society Zoom account was set up by Secretary for Society business. After verbal discussion the Directors decided to close the account during 2021.
- Review and filing by Secretary of documents regarding non-profit status. SDG provided the following documents submitted to various federal and state entities:

Dr. Shaun L. Winterton • Director & Secretary
North American Dipterists Society • P.O. Box 231113 • Sacramento, California 95823, USA
Telephone: (916) 599-0810 • Email: swinterton@dipterists.org
A 501(c)(3) non-profit organization • https://dipterists.org

Minutes of Directors Meeting
19 December 2021
Page 2

    - US IRS – determination of tax exempt status
    - CA Secretary of State – Corporation Statement of Information
    - CA Attorney General – Registration Annual Renewal
    - FTB – 199N e-postcard confirmation
- Society P.O. Box was renewed through automatic renewal (currently $134 per year).
- ISSN numbers for the journals Fly Times and Fly Times Supplement have been acquired from the library of congress (Provided by SDG).
- Discussion of how to make the Directors annual meeting minutes public. Agreed that they should be published in Fly Times when finalized. Also agreed that unanimous written consents would not be published, but will be listed by title and date in annual meeting minutes.

Item 4. *Reports*:
1) Membership report (SDG):
- The Society has 26 paid members in 2021 (28 as of the final budget report):
    - Six of these paid as new members for the 2022 membership year (eight as of the final budget report)
    - It is hoped that all previous members will renew their membership in 2022; SDG will contact each of them to this effect.
    - Out of the 28 members who paid in 2021 (20 for the 2021 calendar year, 8 for the 2022 calendar year), 16 are Founding Members (with the associated $150 fee). Two of these founding members who joined in 2021 indicated they would continue to pay the full founding member fee ($150) as renewals.
    - Although there may be more students, one member joined (and renewed) in 2021 at the student rate of $20.
- Membership consists of people from five countries: two from Australia (both Founding Members), four from Canada (one Founding Member, three Individual Members), two from Germany (one Founding Member, one Individual Member), two from the UK (one Founding Member, one Individual Member), 18 from the USA (four Founding Directors, eight Founding Members, six Individual Members (one of which was at student member rate)).
- Discussion was had on member recruitment; including what members got from their annual membership. Such benefits presently include the Fly Times and Fly Times Supplement, but this is not exclusively available to members. We could include access to grants in the future, or discounts for merchandise or meeting registrations. Agreement that mechanisms for member recruitment should be part of registration process for ICDX. Merchandising of the Society and ICDX could overlap.

2) Website report (SDG):
- In March 2021 the dipterists.org website went offline due to a fire at the OVHcloud facility serving the site. The details of the website and the outage are provided in Fly Times 66 (note that it is incorrectly reported as March 2020). Through UWC, SDG acquired a new VPS and rebuilt the operating system and website. The website was restored on the 19th March 2021.
- The website continues to serve forms from iATS Payments for Membership and Supporter donations (see previous action items above).
- Details of updated Field Meeting website were provided, including a form to indicate interest in the meeting. Further updates will be provided by SDG in early 2022, including a meeting registration form.

- The International Congresses of Dipterology webpages have been historically served on the nadsdiptera.org website. The dipterists.org website contains all the web information for ICDX, but all content related to previous congresses is retained on the nadsdiptera.org website.
- We continue to serve the entire ICDX website. The ICDX website was built after March 2021, and is continually updated by SDG as needed by the ICDX organizing committee.

3) ICDX report (SLW):
- SDG and SLW will start seeking sponsorships on behalf of the Society for ICDX in 2022, including formal requests from entomological and other societies and businesses.
- Announcements have gone into the Fly Times and this will continue to be the main avenue of communicating updates regarding ICDX preparations; these updates are also sent out via the Dipterists mailing list and website.

4) Dipterists Directory report (SDG):
- 83 people have submitted their information to the directory from 24 countries in the following regions: 1 Africa, 2 Asia, 2 Australasia, 11 Europe, 1 Middle East, 3 North America and 4 South America.

5) Dipterists mailing list report (SDG):
- We continue to use MailmanLists and are very happy with the service.
- 666 people from 87 countries are subscribed to the list from the following regions: 25 Africa, 9 Asia, 2 Australasia, 2 Central America, 33 Europe, 5 Middle East, 3 North America, and 8 South America.
- The first posts to the list were in February and there have been posts in every month except for May, July, August, and September, with a total of 21 different threads.

6) Social media reports:
- Facebook (SDG)
  - 506 people “liked” the page, and 544 “followed” the Society webpage.
  - Most interactions were from the announcement for the Manual of Afrotropical Diptera, volume 3 (5366 people reached, 531 engagements).
  - Most posts have reached approximately 100–200 people. Exceptions were the announcements for the existence of the Society (two posts; about 2000 and 820 reached), our new VPS (700 reached), and the deaths of Chris Thompson (522 reached) and Laszlo Papp (305 reached).
  - Growth is good, but SDG related that it could be better. Discussion centered on how this could be done.
- Twitter (CB):
  - Twitter account has positive developments and will continue to be utilised as a major communication avenue for the Society and ICDX.

7) Publications reports (SDG):
- As reported previously, Fly Times and Fly Times Supplement each have an ISSN.
- Fly Times. The Spring 2021 issue was published online on 19th June; Fall 2021 issue will be published on 20th December.

- No Fly Times Supplements were published in 2021, although at least one issue is in preparation by SDG.
- No issues of Myia were published in 2021.

8) ESA meeting report (CB):
- CB attended the Entomological Society of America conference virtually, and presented a pre-recorded talk on the Society and ICDX at the meeting of the North American Dipterists Society.

Item 5. *New Business:*
1) Annual conflict of interest statements were signed and filed.

2) Tax Preparation for 2021:
- SDG proposed that the Society continue using Mark Zivkovic (Pasquesi-Sheppard) as the tax preparer for the 2021 tax year, although we should expect that it will not be *gratis* as in previous years. All present were in agreement that this should continue.

3) Insurance:
- The legal and fiscal liabilities of insurance for the Society were discussed for the Society in general and for Directors and Officers, noting that meetings should have insurance coverage.
- SDG indicated that the Society will need to provide a certificate of liability for use of the facility for the 2022 field meeting; these certificates of property insurance and of liability insurance would be for $1,000,000 each.
- Any insurance should also be appropriate for the ICDX in 2023, although insurance is included in the contract with the venue, Silver Legacy.
- The directors agreed that options for insurance should be explored as soon as possible as an action item to mitigate any potential insurance liabilities for the Society. It was agreed unanimously that the secretary, with the assistance of the remaining office bearers, would explore options for insurance for the Society and report back to the directors as soon as possible.

4) 2022 Field meeting (SDG):
- The field meeting is scheduled for the 13–17th June 2022 in the Pine Barrens, southern New Jersey, USA. Jon Gelhaus (The Academy of Natural Sciences of Drexel University, Pennsylvania) is the organizer. The meeting will be held at the Lighthouse Center in Waretown, New Jersey, with collecting areas nearby. There is no deposit required, so payment will be at time of use.
- Discussion centered on the proposal by SDG for Jon Gelhaus to be appointed to a formal title in the Society, with appointed authority as Field Meeting Chairperson (under Article 5, Section 1 "and other officers") to act on the behalf of the Directors in matters related to the Field Meeting. This may include signing a contract with the venue and other standard activities of meeting planning and organization. After discussion, SDG proposed Jon be appointed as the Field Meeting Chairperson, with the authority, with consultation, to act on behalf of the Directors with respect to 2022 Field Meeting planning and execution. Moved by SW, seconded by CB. This was agreed unanimously.

- The website will be updated with the newest information after publication of Fly Times 67 with its included update.
- The Academy and Drexel University will provide:
    - vehicles for transportation to the field sites and the Academy to work in the collection
    - microscopes and other equipment
    - the substantial discount on the facilities
- The Society will plan to provide a certificate of property insurance and liability insurance as required, due right before the meeting.
- Registration (which is all inclusive, with lodging, breakfast, lunch supplies, 2 dinners, 2 student helpers (1 being an additional driver), microscopes, a couple of vehicles, use of meeting and lab facilities) would be about $200 per person (based on 25 people). That comes to about $140 for lodging, with $60 for everything else). Lodging may be handled separately, and the $60 is subject to adjustment if necessary.
- Action item: SDG to make a payment page on the Society website for Society meetings using IATS payments. SDG proposed to accept registration plan and action item. Moved by CB, seconded by MH.

5) Proposal that The Society to sponsor part of Fly School:

- SDG proposed that the Society provide a one-off payment to sponsor basic costs of running FlySchool (dipteracourse.com). Another alternative presented was to provide grants to students directly for their participation. Discussion of this, as well as amounts to provide, were tabled to a later date, to be handled through UWC.

Item 6. *Election and/or re-election of Directors:*

- According to the bylaws, the Directors hold their positions through the next annual meeting after election. Thus, the last order of business shall be election/re-election of Directors. Each Director shall cast one vote for up to the number of Directors to be elected and/or re-elected.
    - SDG proposed an up or down vote on all positions and candidates. All presented voted in favor of this proposal.
    - All four current Directors (Steve Gaimari, Martin Hauser, Shaun Winterton, Chris Borkent) were re-elected by verbal vote to continue to fill the four Director positions of the North American Dipterists Society for 2022, and continue to serve in their executive capacity, as currently indicated.

Item 7. *Next meeting:*

- The next meeting of the North American Dipterists Society is scheduled for 12th Dec, 2022 at 1pm.

Item 8. *Meeting Adjournment:*

- CB called for a motion to adjourn the meeting. Seconded by MH. Carried. Meeting adjourned at 17:05pm.

Submitted by:
Shaun L. Winterton
Secretary

North American Dipterists Society 2021 Financial Summary
(January 1, 2021 – December 31, 2021)

Income	
Donation	$ 20,000.00
Memberships	$ 3,271.87
Total	**$ 23,271.87**
Expenses	
Bank fees	$ 81.44
Website fees	$ 177.48
Government fees	$ 45.00
Operating cost	$ 148.99
Conference fees	$ 450.00
Total	**$ 902.91**
Net Gain	**$ 22,368.96**

Beginning balance	**Ending balance**	**Net Change**
1784.36	24153.32	22368.96

North American Dipterists Society 2020 Financial Summary (final)	
Income	
Donation	$5,000.00
Total	**$5,000.00**
Expenses	
Bank fees	$32.44
Website fees	$162.21
Legal fees	$2,275.00
Government fees	$625.00
Operating cost	$120.99
Total	**$3,215.64**
Net Gain	**$1,784.36**

EXPENSES BREAKDOWN

Bank fees		
Check order	$22.95	19-May
foreign transaction	$3.49	29-Jun
paper statement fee	$3.00	31-Jul
paper statement fee	$3.00	31-Aug
Total	**$32.44**	
Web		
Dream host fee	$45.93	2-Jun
OVH.com	$116.28	29-Jun
Total	**$162.21**	
Legal fees		
Check 152	$280.00	28-Aug
Check 151	$1,995.00	5-Jun
Total	**$2,275.00**	
Government fees		
CA registration fee	$25.00	24-Sep
IRS user application fee	$600.00	30-Jul
Total	**$625.00**	
Operating costs		
Zoom registration	$14.99	14-Dec
USPS P.O. Box	$106.00	16-Dec
Total	**$120.99**	

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