FLY TIMES

ISSUE 73

FALL 2024

FLY TIMES

Issue 73, Fall 2024

Editor-in-Chief Stephen D. Gaimari

PUBLISHER
Dipterists Society
P.O. Box 231113
Sacramento, California, 95823, USA
https://dipterists.org

JOURNAL WEBSITE
https://dipterists.org/fly_times.html

PRINTED BY
TheBookPatch, LLC, a service of Wilshire Press, Inc., Scottsdale. Hard copy available for purchase at https://app.thebookpatch.com/BookStore.aspx.

DISTRIBUTION
Fly Times is simultaneously produced 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.

SUGGESTED CITATION
Cranston,P.S. & Farrow, R. 2024. An unusual case-making larval tipuloid from Sydney Sandstone, New South Wales, eastern Australia. *Fly Times* 73: 1–6.

INSTRUCTIONS TO AUTHORS

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

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

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

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

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

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ISSN 2769-6073 (Print)
ISSN 2769-6081 (Online)

Available 20 December 2024

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> *Our Mission is to advance the scientific study, understanding and appreciation of the insect order Diptera, or true flies. To accomplish this, we aim to foster communication, cooperation, and collaboration among dipterists, and to promote the dissemination and exchange of scientific and popular knowledge concerning dipterology.*

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

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

From the Editor – Welcome to the latest issue of *Fly Times*! As usual, I am very impressed with the variety of excellent submissions, and I hope they are enjoyable to the readers. Please consider writing an article or two for the next issue, which is slated for spring of 2025. And for larger works, please consider the *Fly Times Supplement* series, found at https://dipterists.org/fly_times_supplement.html.

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

Cover photo – *Coenosia tigrina* feeding on a male of the *Condylostylus caudatus* species group. Spotted at a park near the CNC in Ottawa. Photograph by Zachary Dankowicz.

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CONTENTS

NEWS AND RESEARCH

HISTORICAL DIPTEROLOGY

PHILAMYIANY

MEETING NEWS

SOCIETY BUSINESS

****

NEWS AND RESEARCH

An unusual case-making larval tipuloid from Sydney Sandstone, New South Wales, eastern Australia

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While leading an Australian native plant group foray to the southern margin of the huge Sydney (or Hawkesbury) sandstone geological formation, Roger Farrow revisited a site where 3 years before, in June 2021(austral mid-winter) he found a vertical rock face on which were scattered many small circular cases sealed onto the stone surface (Fig. 1A–D). Prizing off several cases revealed a live but torpid maggot-like insect larva, which was relatively featureless under modest magnification (Fig. 1E, F). Roger had pondered their identity to no avail, so in July 2024 he made a more detailed examination resulting in some photographs and a short descriptive text for the readership of the *News Bulletin* of the Entomological Society of Queensland (Farrow 2024). Supported by the editor, Roger asked whether anyone could put a name on this enigmatic insect.

Peter Cranston was not the only reader puzzled by the insect: emails flowed between local and international dipterists. Some questioned an allocation to Diptera, raising the possibility that the larvae belonged to another order for which some immature stages had a reduced head and lacked appendages. Clearly we needed to view finer details and make appropriate dissections. Following a subsequent native plant foray, Roger passed some live larvae to Peter. The larvae indeed were ‘maggot-like’ and when removed from their encircled positions in the case, only wriggled feebly, lacking any appendages or creeping welts. The head was hemicephalic, incompletely fixed in a semi-retracted position within the anterior thorax (Figs 1G, 2A). The posterior body appeared simple with a rounded spiracular plate bearing two spiracles (the metaspiraculate condition), with only very modest 'anal lobes’ (Fig. 2H). The densely spinose cuticle (Fig. 2I) seemed typical for a tipuloid and all-in-all the larvae must belong to the superfamily Tipuloidea.

The hunt for an identification.

The literature in Australia concerning Tipuloidea (now comprising the family-level taxa Tipulidae (s.s.), Limoniidae, Cylindrotominae and Eriopterinae) has been adult focused, with no synthesis of meagre immature data. Lacking adults, despite rearing attempts, we consulted global literature to progress an identification. All keys consulted implied that a very reduced posterior spiracular plate and anal lobes indicated our larva belonged to “Limoniinae”, likely a paraphyletic assemblage (Petersen *et al*., 2010), although with a possibility of belonging with some taxa allocated to Pediciinae. Further advance was made using a valuable larval key to aquatic Tipuloidea larvae in the Mediterranean region of Europe (Podeniene, 2023). In this work, the larvae keyed unambiguously to *Thaumastoptera* Mik, although some key characteristics (lacking body appendages or creeping welts, with minimal lobes on a dorsally-orientated spiracular disc), might be convergent with certain larval Pediciidae. Apparently conclusive of identity is the constructed larval case, reported only in the genus *Thaumastoptera* and in no other larval Tipuloidea. Otherwise, case-bearing in lower Diptera is known only in some distant Chironomidae.

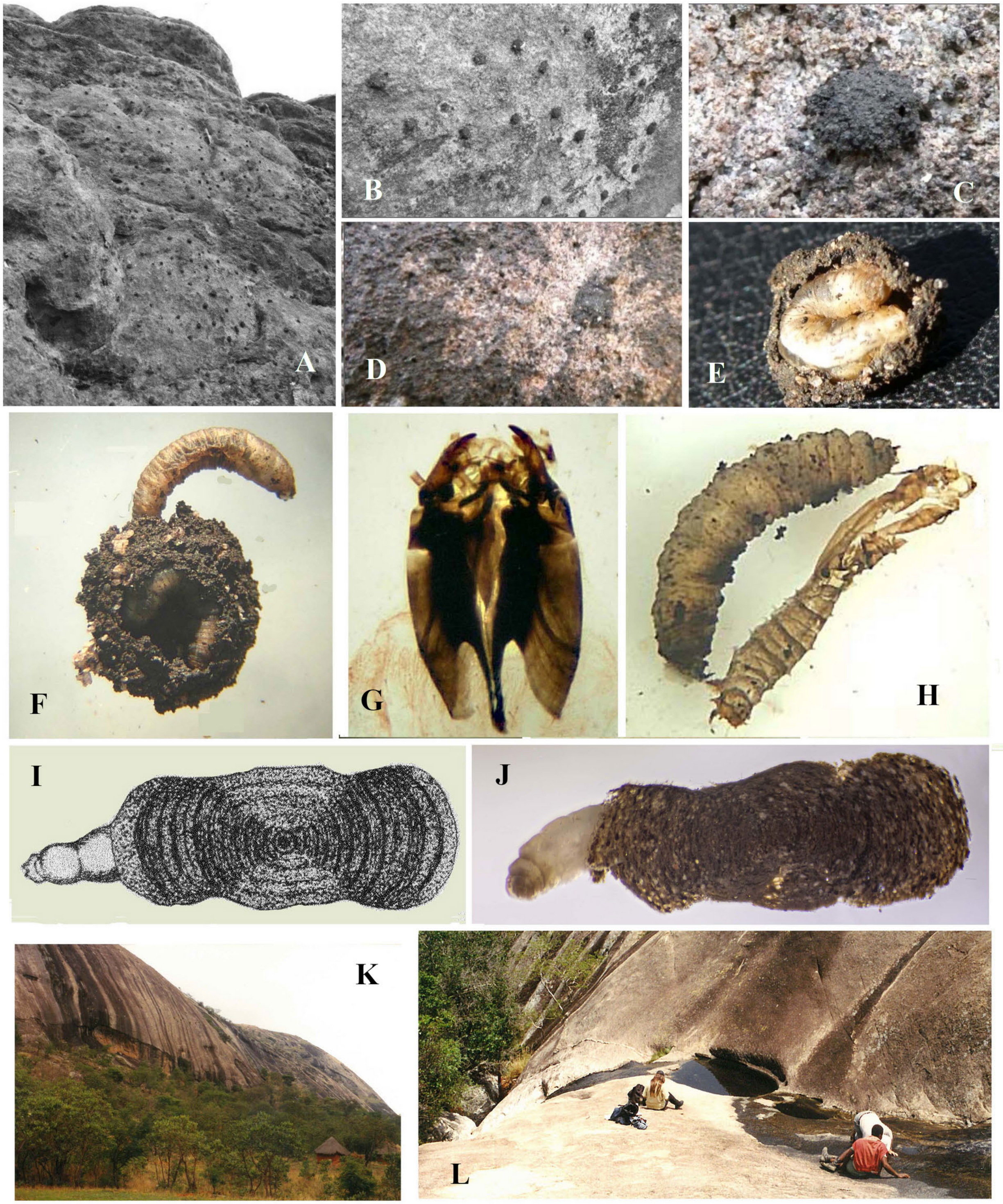

Figure 1. **A.** Sandstone rock face, Morton National Park, NSW, height c. 10–12 metres. **B.** Tipuloid larval cases on rock face. **C.** Dark biofilm with 'grazed' area around case. **D.** Single Tipuloid larval case surrounded by 'grazed' biofilm. **E.** Inverted case containing late stage larva. **F.** Extracted larva and Inverted case with enclosed larva. **G.** Tipuloid larva, cleared head capsule, 1.1 mm long. **H.** Tipuloid larva and pupal exuviae, length 8 mm. **I.** *Thaumastoptera* case and protruding anterior larval body. **J.** *Thaumastoptera* case and protruding anterior larval body. **K.** Rock face (height ca. 100 metres), Ngoma Kuriru, Zimbabwe. **L.** Headwater stream and dry black biofilm / seeps, Ngoma Kuriru, Zimbabwe. Photographs 1A–E, R. Farrow; 1F–H, P.J. Gullan; 1J, H. Mauch; 1K–L, P.S. Cranston; 1I, drawing from Lenz (1920).

Thaumastoptera contains a gradually expanding diversity of species, based on the European type-species *Thaumastoptera calceata* Mik 1866. Recently, Starý & Oboňa (2018) reviewed four Palaearctic species and Boardman (2020) added a species from Cameroon to the known Madagascan and southern African taxa. Thus Oosterbroek (2024) reports 11 species of *Thaumastoptera* presently known, distributed in Europe, the North Caucasus, Saudi Arabia, Cameroon, Madagascar, South Africa, California*,* India and the Philippines. Amber fossil records, supported by possibly diagnostic wing venation, have been reported from Jordanian (Podenas 2000) and Baltic (Kania 2015) ambers. These distributional data include no reports from the Australasia and Pacific regions or Southeast Asia. All records are from adult flies, excepting the type-species which is known in the immature stages. Current understanding of larval morphology, ecology and behaviour still derives almost exclusively from Lenz (1920). Initially he studied material from his professor, August Thienemann, subsequently enhanced by his own collections following recognition of the semi-aquatic locations of the larval cases found near woodland springs in German forested headwaters (Lenz 1920).

However, this generic identity is by no means a *fait accompli*: morphological and eco-behavioural differences between the Australian tipuloid and *T. calceata* seem quite profound, commencing with the case structure, natural history and extending to likely diagnostic cephalic structures.

Ecology and case-structure.

The case of *T. calceata* is described and figured by Lenz (1920) and photographed by Mauch (2017) (Fig. 1I, J). The case-structure and larval biology of *T. calceata* and that observed or deduced for the Australian taxon differ significantly. The German forested spring habitats of the case-bearing immature stages of *T. calceata* seem to be persistently damp in a 'semi-aquatic' condition (Lenz, 1920). Recent studies show beaver dams sustain cased *T. calceata* (Schloemer, 2023). Larval activity around the sand-mud case is easily observed as larvae are active, with head and anterior body usually protruding from the case (Mauch, 2017) (Fig. 1J), even reversing within the case. It is not clear if this larva seals itself into the mobile case.

This behaviour contrasts with our Australian larva, which has yet to be observed outside its case that is firmly affixed to the rock surface. Roger argues that a distinctive area 'bare' of cyanobacterial biofilm around each sedentary case derives from larval grazing around its domicile (Fig. 1C, D). Examination of larval gut contents under a microscope shows it comprises exclusively dark microparticles of cyanobacteria. This flocculent faecal material is extruded if the larval body is squeezed. The case appears to comprise the same material, 'cemented' into a rigid, moulded, circular cover that lacks any opening other than the ventral surface contacting the substrate. Under the microscope, comminuted biofilm and faecal material appear identical – either or both could be the source material for case construction. We do not know the nature of the adhesive (perhaps from digestion ?), nor how and when an active larva exits and re-enters its sealed case.

At the only Australian location at which our larva is known, during several visits we have not experienced wet conditions (which are intermittent, following major rain events). At best the site location may become sporadically semi-aquatic (on a rock face that shows evidence of sporadic flow of rainwater) yet is definitely terrestrial for most of the time. Roger observed that the location of densest cases on a rock surface was stained black from a cyanobacterial biofilm. Globally, inevitably this is associated with ephemeral water flow over rock slabs. Peter has explored this interesting ecosystem in which a vegetational 'cap' holds water and seepage follows adequate rainfall. His long-time favourite chironomid midge larvae develop in such ephemeral waters in western and central Australia and southern Africa (Fig. 1K, L).

Further disparities concern head capsule morphology, perhaps associated with construction and use of a dissimilar case, but more likely linked to different feeding modes in an only apparently similar substrate (wet versus temporarily moist biofilm). From Lenz's (1920) detailed descriptions and drawings, the mandible of *T. calceata* is multi-toothed across the inner and molar surfaces, poorly aligned (Lenz, 1920, Fig. 2F). The mandible of the Australian taxon substantially resembles that seen in many Culicomorpha, with a dominant apical tooth, one or two aligned inner teeth and a substantial plumose seta interna associated with a 'toothed' mola (Fig. 2G).

The toothed mentum (labium) of *T. calceata* is rounded with teeth subequal in size and shape (Fig. 2C), whereas in the Australian taxon the median tooth is quite elongate (Fig. 2B). In *T. calceata*, the antenna is bipartite (Fig. 2D) with apical sensilla, whereas the antenna of the Australian taxon has a single segment surmounted by a rounded sensillum (Fig. 2E).

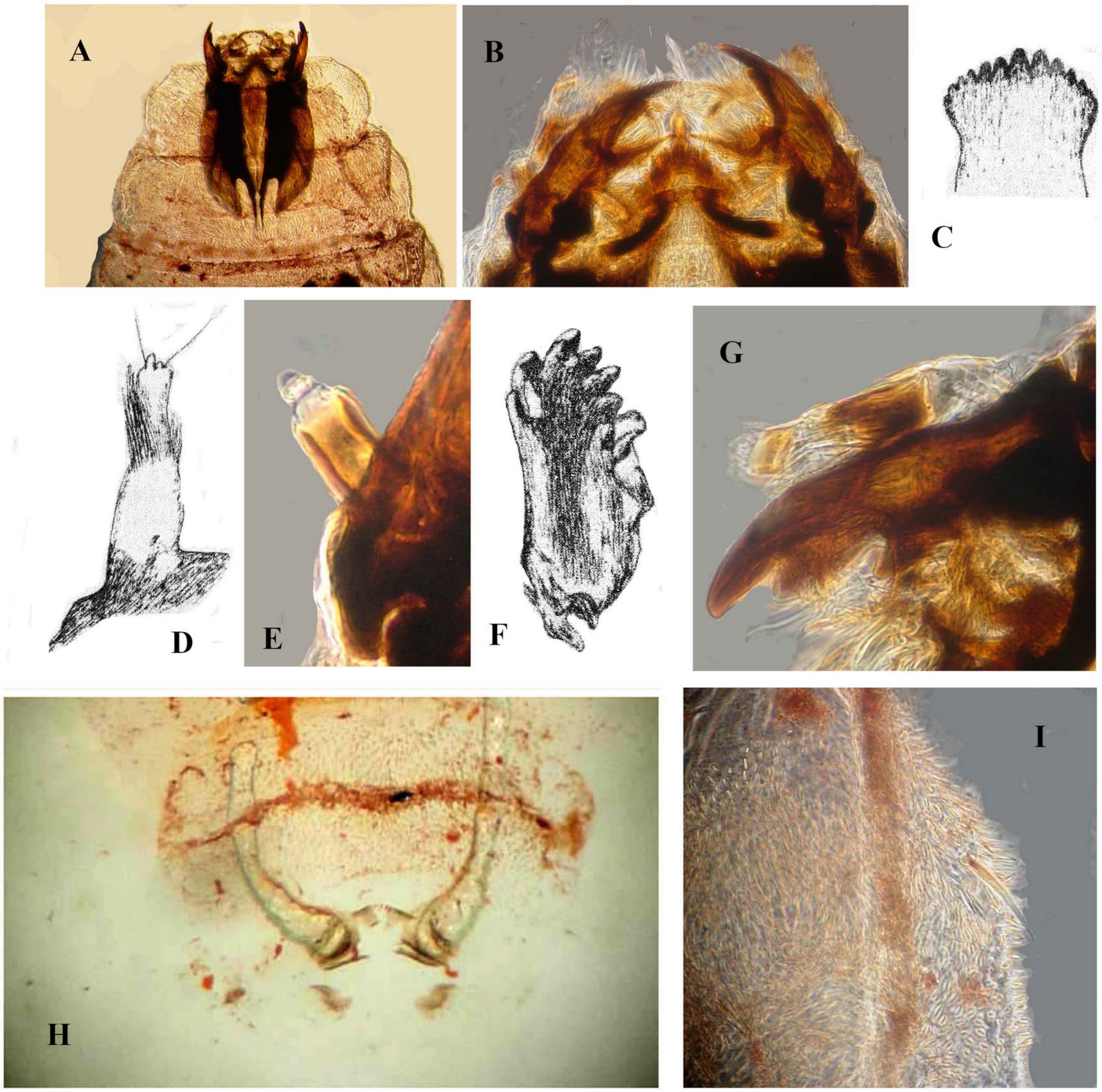

Figure 2. **A.** Head capsule and anterior thorax. **B.** Anterior head, ventral view, mentum, mandibles. **C.** *Thaumastoptera*, mentum (labium sensu Lenz). **D.** *Thaumastoptera*, antenna. **E.** Tipuloid larva, antenna, 80 µm. **F.** *Thaumastoptera* mandible. **G.** Tipuloid larva, mandible, 400 µm. **H.** Tipuloid larva, posterior segments, spiracular disk. **I.** Tipuloid larva, thoracic cuticle, spinules 25 µm, long spine 125 µm. Photographs 2A–B, 2E, 2G–I, P.J. Gullan; 2C–D, 2F, Drawings, from Lenz (1920).

Conclusion

An early suspicion was that this Australian larval taxon was congeneric with the sole documented case-bearing tipuloid, *Thaumastoptera calceata*. However, this must be rejected in the light of very different morphology of important head capsule features. In other dipterans in which the larvae are well known, such different morphologies would be incompatible with belonging to the same high taxon, let alone congeners. We cannot even suggest a family within Tipuloidea for our larva. Most likely it would be allocated to Limoniidae, but almost certainly this is a paraphyletic assemblage of taxa that do not fit Tipulidae s.str. (Petersen *et al*, 2010). Furthermore, larval diagnostic features for comparative analysis have not been proposed. Clearly a detailed study of the larva and its ecology and life-history would be fascinating, but the location is somewhat remote from Canberra and Sydney and is not conducive even to irregular visits. Ex-situ rearing has failed thus far. While future bar-coding our larva against local adult tipuloids may be possible to allow linkage, it is premature to hope for identification via CO1 barcode or even multi-gene DNA study, due to lack of a comprehensive library of related taxa. The mystery continues …..

Acknowledgments

Many thanks to Erik Mauch, for correspondence and access to photographs of larval *Thaumastoptera* in Germany from his 2017 book. Harald Mauch photographed a larva collected by Stefan Zaenker in the German Hessian Midland Mountains. Greg Courtney (Iowa State University) provided advice and access to an important but elusive reference. Lenz's publication was accessed through the Biodiversity Heritage Library, an immensely valuable source for 'older literature'. An Adobe product helped make an OCR of the original 'old' German Lenz publication, and Google Translate made life a lot easier. We thank Penny Gullan for photography at the site and subsequent photomicroscopy. Finally, thanks to all who attempted to identify the Australian larva from often sub-optimal photographs!

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Communal oviposition by *Tabanus fairchildi* Stone, 1938 and comments on parental care in Diptera

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Tabanus fairchildi is a widespread North American tabanid: Wisconsin to Nova Scotia, south to Oklahoma and Florida (Burger, 1995). It is one of only three North American species whose larvae live among stones and gravel in swift-flowing streams (Pechuman, 1972, 1981; Teskey, 1990). Pechuman (1972, 1981) stated "*The eggs are placed on projecting stones or logs in riffles, and often many females deposit their eggs on the same object, resulting in the accumulation of several hundred egg masses.*" This report adds further information on the egg-laying habits of *Tabanus fairchildi*. Our observations were on the Southwest Miramichi River in New Brunswick, Canada. Rather than being a "*swift-flowing stream*" it is a large wide river draining 7,600 Km^2 in central NB. The oviposition site was a metal bar about six inches wide in 24 inches of water (at the time of the photograph) with a scale on the bank-facing north side to measure water depth (Fig. 1).

Figure1. Southwest Miramichi River, July 8 2024 7pm Atlantic Daylight Time; arrow points to the communal oviposition site, a metal bar.

On 8.50 pm Atlantic Daylight Time (ADT) on July 1 2024, KJC photographed dozens of *T. fairchildi* ovipositing on the southern face of the metal bar (Fig. 2) with far fewer ovipositing on the north-facing face above the "8" (Fig. 3); white recently-laid egg masses were present on both surfaces. This presence of multiple eggs masses on the same substrate is in agreement with Pechuman (1982). However, the observation that multiple females oviposited together as opposed to one female laying one batch of eggs then leaving before a second female repeats the process is a new finding.

Figures 2–3. Communal oviposition by *T. fairchildi*. **2** (left). On the southern face of the metal bar; note the recently-laid egg masses (white); 8.50 pm Atlantic Daylight Time July 1 2024. **3** (right). On the the north-facing face of the metal bar. There are also recently laid egg masses on the eastern (downriver) narrow edge of the metal bar; 8.50 pm Atlantic Daylight Time July 1 2024. Compare with July 8 when eggs were being laid below “7” and with a gap between the two masses (Fig. 4).

On our visit on July 8 2024 at 7 pm ADT the river was high and prevented us from seeing the south-facing side of the stake. On the north-facing side the mass of females at the top of the metal bar had expanded to cover the “8” mark and a second mass of females began just above the “6” mark and extended to the river surface (Fig. 4). Worth noting is a gap between the two masses of females where there were neither egg masses nor females. Figure 5 is a crop from the lower mass of females on July 8 2024 showing females, freshly-laid egg masses, and black hatched egg masses.

Discussion

We spent about 30 minutes observing on July 8 and saw no females arrive or leave. Activity on the metal bar was slight; some females walking slowing over the mass. A nagging question is why the gap between masses (Fig. 4)? Perhaps the first female to arrive landed and oviposited on a bare piece of the metal bar. Later-arriving females were attracted to this female and the mass grew larger. If this scenario is correct it doesn’t explain why one female would start a cluster at some distance from the first. We can’t image there being a different surface on the metal bar at mark “7” where no females oviposited.

Communal oviposition is well known for the athericid *Atherix lantha* Webb (Infrorder: Tabanomorpha) (Loiselle and Giroux, 2019). In this species the females aggregate in large clusters on objects over streams. After oviposition the females remain with their egg mass until death, newly-arriving females land, oviposit, and die on the cadavers of previous females. The larvae hatch in the midst of the dead and dying females and drop into the water; the entire sequence regarded as “*something approaching parental care*” (Smith, 1989).

Loiselle and Giroux (2019) quoted Madsen (2012) who described similar behaviour of a European species and who speculated:

> *The first attraction to a good spot for the females seems to be visual; afterwards pheromones are used. A fresh bulk of flies would have a characteristic odour. The gathering of females has probably a negative impact on egg predation. And perhaps their size creates confusion with wasps?*

Figures 4–5. Communal oviposition by *T. fairchildi*. **4** (left). On the north-facing face of a metal bar; 7 pm Atlantic Daylight Time July 8 2024. Note that the top cluster of females and eggs has expanded downward to cover the "8" mark which was clearly visible on July 1 (Fig. 3). **5** (right). On the north-facing face of a metal bar; July 8 2024. A crop of Fig. 4.

Although not communal oviposition, egg-laying in the North American tabanid *Goniops chrysocoma* (Osten Sacken) resembles that of the Athericids. A female lays a batch of eggs on the underside of leaves above damp ground in wooded areas and remains with the eggs until they hatch; when disturbed she makes a loud buzzing sound. Such behaviour would fit into Smith's (1989) "*approaching parental care*." From Pechuman's observations (1972, 1981) of "*the accumulation of several hundred egg masses* [on the same object]" it would appear that the females of *Tabanus fairchildi* do not remain with the eggs until they hatch.

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Observations on a minute aquatic empidoid from a waterfall in New Zealand

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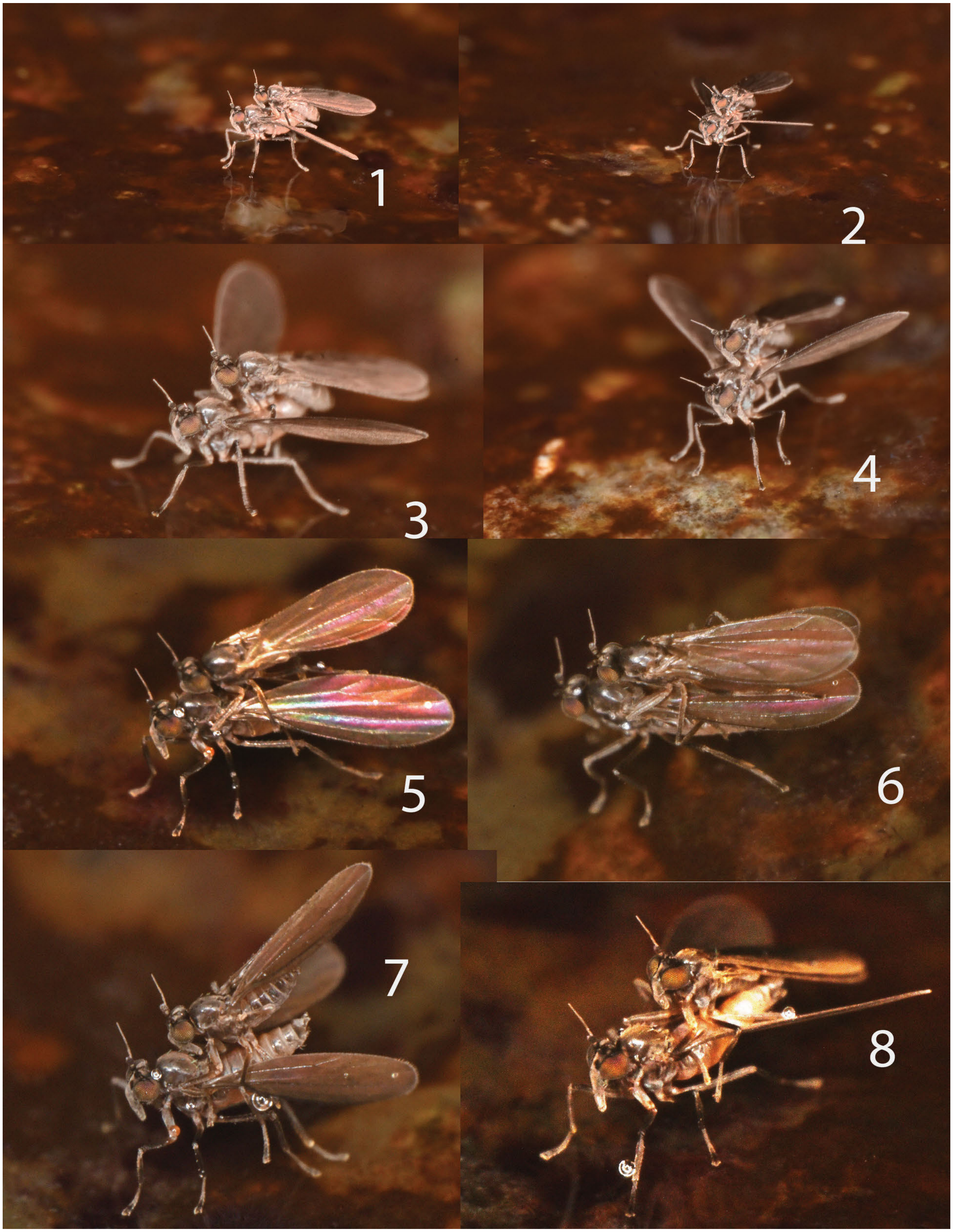
On March 18, 2023 one of us (SAM) noticed multiple pairs of an exceptionally tiny empidoid in the spray zone of a small waterfall in an old growth podocarp forest in Whirinake National Forest Park near Minginui, New Zealand (Figs 1–6). They were mating in the fine spray while standing on a thin film of water. These minute flies had an estimated body length of one mm excluding the long wing (based on frame size of the photographs). A first few photos were taken using a 105 macro and extension tubes, then further shots were taken the following day with higher magnification equipment (a Laowa 85 mm macro with extension), a bit of plastic wrap to protect the lens and camera in the spray, and a headlight to get a clearer picture of what was happening. The same site again had multiple mating pairs, allowing for the appended photos showing most external structures, including the remarkable proboscis and wing. Beyond recognizing it as a "doid", SAM could not identify the fly but BJS recognized it as a species of *Asymphyloptera* Collin (Empididae: Clinocerinae). Four undescribed species have been identified from New Zealand.

There is only a single species of *Asymphyloptera* described from the Australasian Region, but numerous undescribed species have been identified [Australia, New Caledonia, New Zealand and Philip Island (Norfolk)] (Sinclair 1995, 2015). In the New World, 10 species have been described (Chile, Colombia, Costa Rica, Dominica, Ecuador, Mexico, Peru, Venezuela, and USA) (Sinclair 2015; Ramos-Pastrama et al. 2023). Species occur in small streams to rocky cascading creeks where adults are found on wet rocks as described above. The flies sometimes run in a zig-zag fashion and rather than take flight to avoid capture will run along the rock face. Although currently assigned to the subfamily Clinocerinae, a recent molecular phylogeny assigned *Asymphyloptera* outside of the subfamily (Vojvoda Zeljsko et al. 2024).

We are drawing this remarkable undescribed fly to the attention of the dipterological community with a note in Fly Times rather than a formal description because it is thus far known only from photographs. Although SAM had written permission to collect on private land near the park where the flies were found, he did not have a National Park research permit. The Department of Conservation (DOC) in New Zealand is supportive of research and there is an accessible online system for applying for permits, but the system does not lend itself to general collecting or "opportunistic" discovery ... you have to know in advance exactly where and what you are going to collect, and you have to provide precise coordinates and maps. This was not a research trip, and SAM had no expectation of making a dipterological discovery while on a hike in the park. With no research intent, does opportunistic photography of an organism of interest to specialists constitute illegal research? We hope not, or else thousands of iNaturalist photographers are breaking the law too.



Figures 1–6. An undescribed species of *Asymphyloptera* (Empididae: Clinocerinae) mating in an old growth podocarp forest in Whirinake National Forest Park near Minginui, New Zealand.

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Trapping Drosophilidae with prune juice

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I am continuing to try different baits to attract Drosophilidae from my composter. So far, I have found Vienna lager beer to be a better bait than American Adjunct Lager (Hribar 2020a), red wines to be better baits that white wines (Hribar 2024), and apple cider vinegar to be a better bait than distilled white vinegar (Hribar 2024). I attempted to determine whether Vienna lager beer or red wine was the better bait but that trial was inconclusive (Hribar 2020b).

One potential bait that I tested was prune juice, purchased from a local grocery store. I used the new trap that I constructed (Hribar 2024b), baited it, and set it out overnight (23–24 June 2024). I collected 15 *Drosophila* sp. flies, all of which resembled *D. melanogaster*. Not very impressive, but better than zero. I wondered whether the new trap would perform better or worse than the water bottle trap I had used previously (Hribar 2024a). I baited both traps with prune juice and set them out for four days (5–9 July 2024). There was a definite difference between the two traps (Table 1).

Table 1. Drosophilidae collected in two traps baited with prune juice.

	Water bottle trap	Cosmetic jar trap
Z. indianus (African fig fly)	25	0
D. repleta Group	4	0
Other Drosophilidae	218	3
Phoridae	2	0
Lepidoptera	1	0
Psocoptera	1	0

The water bottle trap definitely collected more flies than the cosmetic jar trap. One of two reasons may explain this: 1, the holes in the cosmetic jar trap are not big enough; 2, there are not enough holes in the cosmetic jar trap. Piñero & Foley (2018) used many more than I did; I drilled only four holes.

Next, I wondered how prune juice would perform versus Concord grape juice. Two water bottle traps were baited (one with each bait) and deployed from 22–26 July 2024. The difference in catch was remarkable (Table 2).

Table 2. Drosophilidae collected in traps baited with Concord grape or prune juices.

	Concord grape	Prune
Z. indianus (African fig fly)	17	0
D. repleta Group	4	0
Other Drosophilidae	81	0

The trap baited with Concord grape juice also collected one Calliphoridae (*Lucilia sericata*), one bark beetle, and six other Coleoptera. The trap baited with prune juice attracted no Drosophilidae at all. Only two insects were collected, one Scatopsidae and one weevil. A cursory examination of the scatopsid suggests *Holoplagia guamensis* (genitalia and yellow-white tarsi). I looked at photos online and the weevil appears not to be either the plum curculio or the plum gouger. This is not surprising, as the plum curculio does not occur in southern Florida (Lampasona et al. 2020). The plum gouger is recorded from Florida, but I have found no locality records (Ayers 1923, O'Brien and Wibmer 1982). In any case, plum trees will not flower and hence not set fruit south of Lake Okeechobee (Sarkhosh et al. 2018).

Prune juice and grape juice differ in chemical composition. Prune juice contains different acids than does grape juice. Prune juice has no citric acid or tartaric acid, and only a trace of ascorbic acid. Prune juice also contains high levels of quinic acid whereas grape juice has none. Prune juice contains only about a third the amount of malic acid that grape juice does. Grape juice contains only traces of citric and ascorbic acids, but does contain tartaric acid (Van Gorsel et al. 1992).

Another consideration to take into account is that the product sold as prune "juice" is not actually a juice; rather, it is a water extract of prunes (Luh 1980). Prunes are dried plums (Stacewicz-Sapuntzakis et al. 2001). There are two plum species of worldwide economic significance. The Japanese plum (*Prunus salicina* Lindl.) is adapted to grow from temperate to subtropical regions. The European plum (*Prunus domestica* L.) is grown in cooler temperate climates. Fruit of both species is used to produce juice. A third *Prunus* species, *P. mume* Sieb. et Zucc., produces a fruit that is called a plum in Asia; it is actually more closely related to apricot. The fruit, also called "ume", is used to produce juices (Fanning et al. 2016). In Europe and the United States, prunes are made from *P. domestica* fruit, which has been cultivated and propagated since ancient times. The most commonly used cultivar in California is d'Agen, a cultivar of French origin (Stacewicz-Sapuntzakis et al. 2001).

Plums grown for prunes may not be suitable hosts for *D. suzukii* (Wilson et al. 2013). Does this hold true for other Drosophilidae? Alawamleh et al. (2016) reported *Z. indianus* from plum in Jordan, as did Çatal et al. (2019) from plum in Turkey. Çatal et al. (2021) also collected *Z. indianus* in apple, cherry, peach, blackberry, fig, and plum orchards. Obviously, *Z. indianus* is attracted to plum and to prune juice. The stark difference between the prune juice and Concord grape juice baits suggests that *Z. indianus* prefers grape to plum as an oviposition medium, but will utilize other substrates when grape is not available.

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An unusual case of trapped eggs in the male terminalia of the bot fly *Cuterebra patagona* Guérin-Méneville, 1844 (Oestridae: Diptera) from Argentina

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While conducting a series of bot fly terminalia dissections, two cuterebrine eggs were discovered in the sacculiform conjunctiva (membranous sac connecting the hypandrium, anteroventral epandrium, and sternite 6) of a *Cuterebra patagona* male specimen (Fig. 1). This is the first report of this extraordinary circumstance, and only the fourth documented Neotropical cuterebrine species with scanning electron micrographs of eggs (Leite 1988; Leite & Williams 1989; Colwell *et al.* 1999, Colwell 2006). It would be premature to directly associate this finding to this species, and further investigation is required for confirmation.

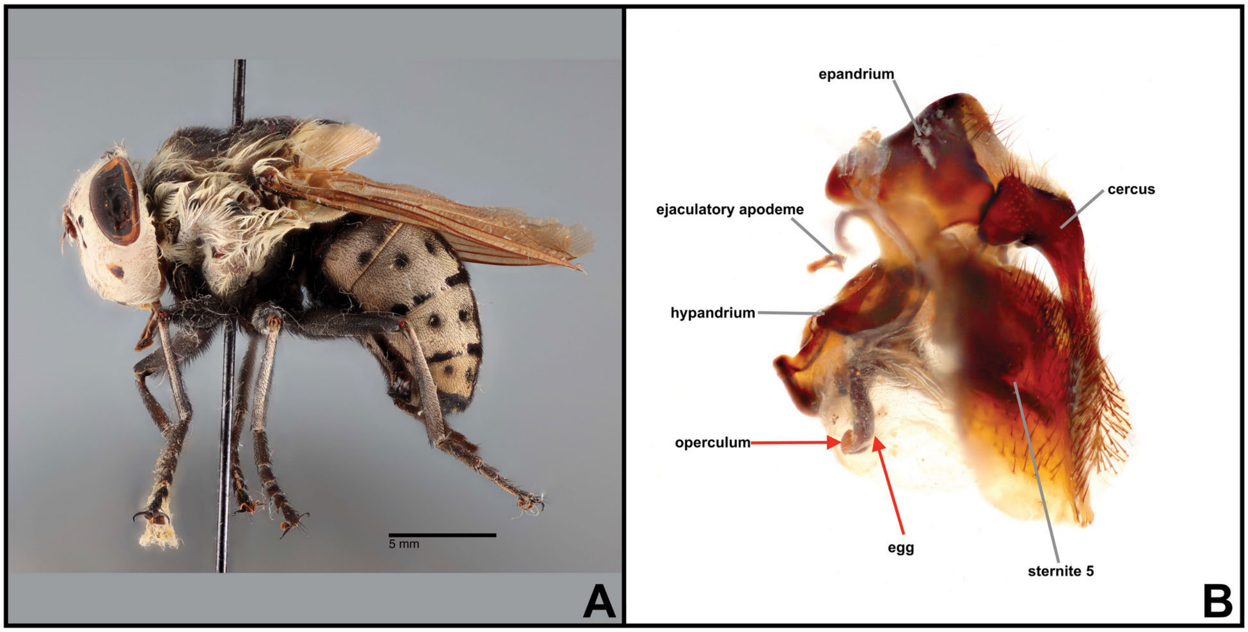

Figure 1. *Cuterebra patagona* Guérin-Méneville, 1844, ♂: **A.** Habitus, lateral view; **B.** Dissected terminalia, lateral view (egg pointed by red arrows).

Some brachyceran flies have been observed to oviposit on other flies as transport hosts. Eggs of a parasitic fly in the genus *Stylogaster* were found on the abdomen of a calliphorid in the genus *Tricyclea* (Jensen *et al.* 2020). The monotypic human bot fly (*Dermatobia hominis*) uses mosquitos and other zoophilic and synanthropic flies as paratenic porters of their eggs to their definitive hosts. Other bot flies of the subfamily Cuterebrinae, restricted to the Western Hemisphere, have females known to lay their eggs indirectly on, along the runs of, or on areas frequented by their mammal hosts (Ferrar 1987; Marshall 2012).

The biology of *Cuterebra patagona* remains to be investigated, as it is a poorly-studied South American species, compounded by their paucity in research museum collections despite the distinctive appearance and size of the adults. Since its formal description, the literature has been limited to the female adult morphology and the species' distribution record. For this intriguing observation, one plausible scenario is that the male bot fly's attempt to mate with a gravid female may result in accidental egg deposition inside the membranous sac.

Description of eggs (Figs 2–4): Length: 0.526 mm (n=2). Rufous, elongated digitiform profile, anteriorly recurved, tapering on both blunt ends, wider surface ventrally appearing bilobed forming furrow posteriorly; anterodorsal fingernail-like operculum; exochorion lacunose with thick struts on the dorsal side giving an appearance like narrow cells; struts on lateral sides more widely spaced than dorsal region, aeropyles more apparent; ventral side of the exochorion with relatively smooth, shallow, and wide mesh cobbled sculpting. Micropylar plate at the anterior pole, dorsomedial pit cluster (of five) near the posterior end.

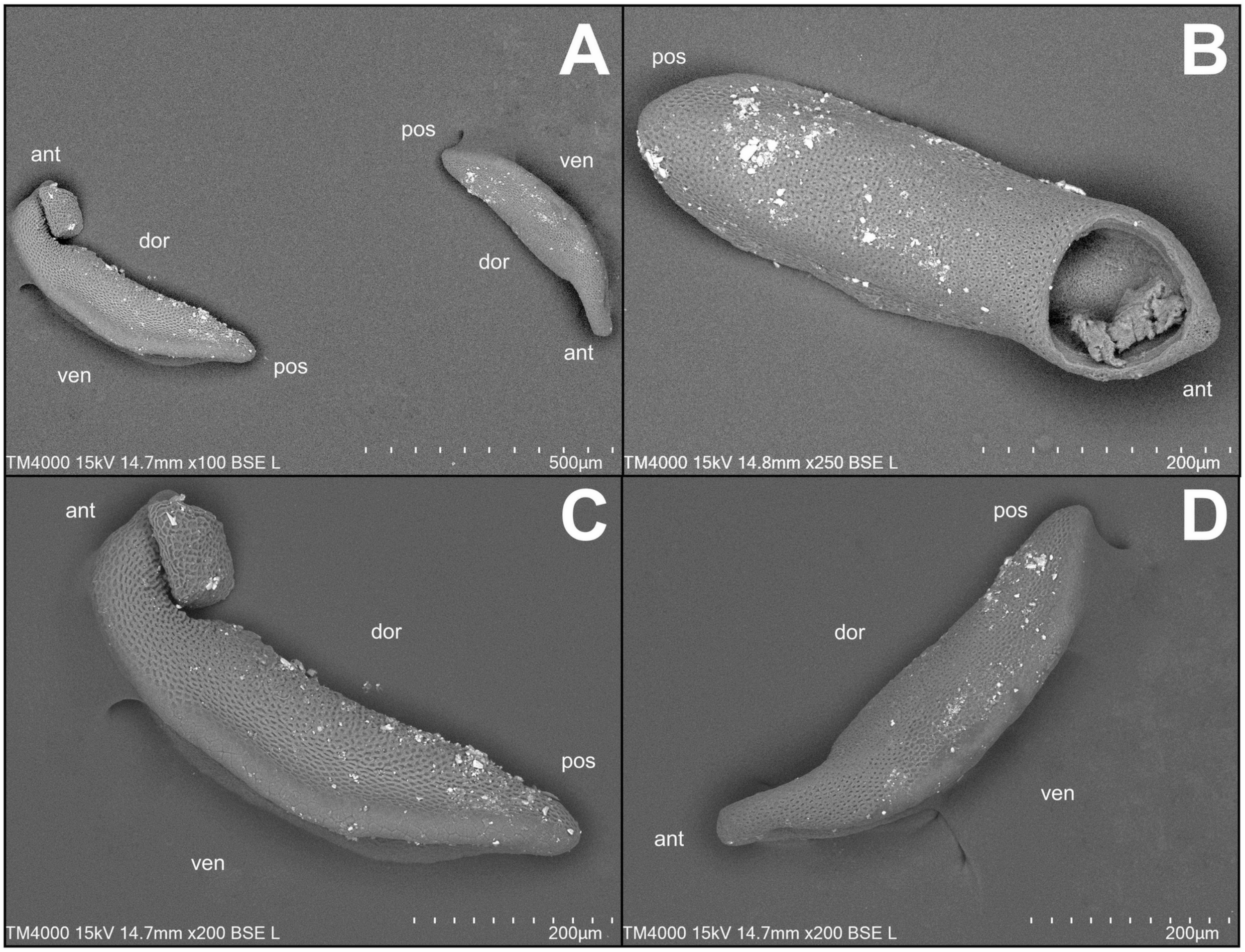

Figure 2. Scanning electron micrographs of eggs found in *Cuterebra patagona* male terminalia: **A.** Lateral aspect; **B.** Dorsal aspect; **C** and **D.** Eggs magnified 200X, lateral aspect. Abbreviations: *ant*, anterior end; *dor*, dorsal side; *pos*, posterior end; *ven*, ventral side.

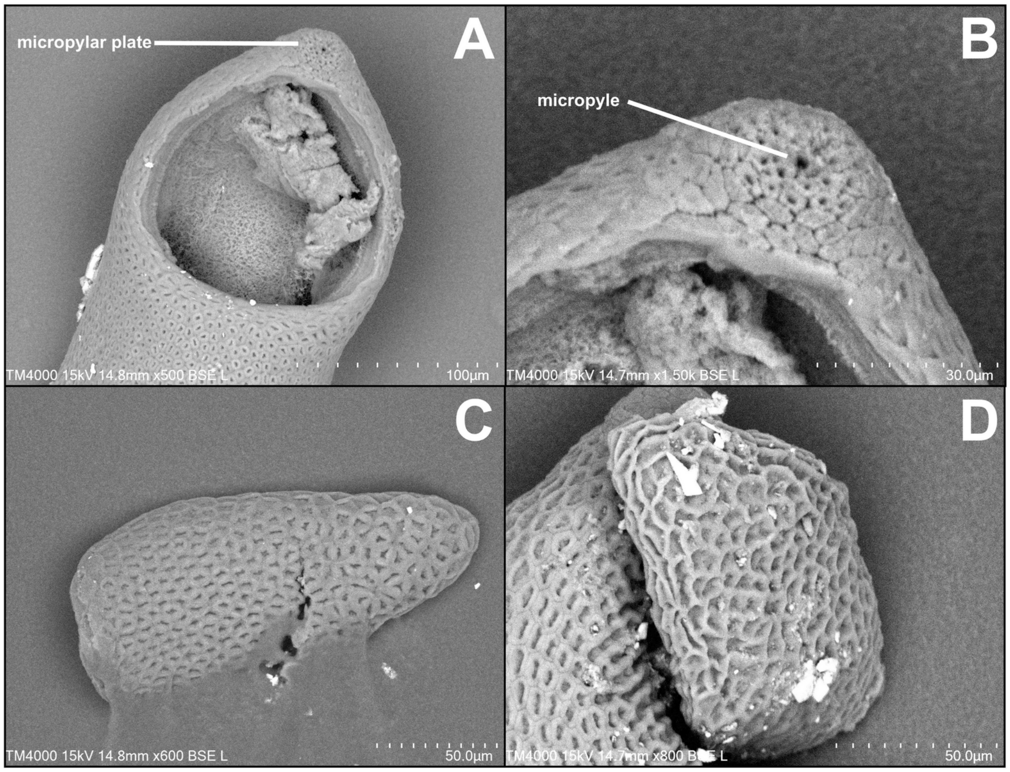

Figure 3. Scanning electron micrographs of eggs (anterior end) found in *Cuterebra patagona* male terminalia: **A.** Anterior end, dorsal aspect; **B.** Micropylar plate at anterior end, anterodorsal view; **C** and **D.** Egg operculum, dorsal aspect.

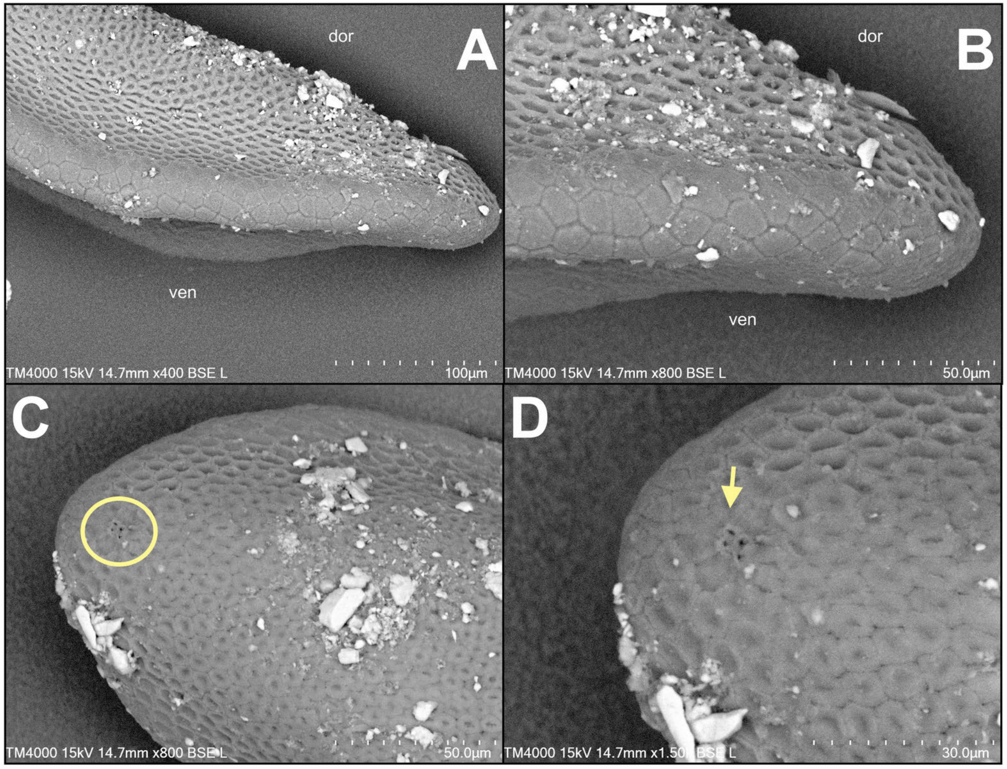

Figure 4. Scanning electron micrographs of eggs (posterior end) found in *Cuterebra patagona* male terminalia: **A.** Posterior half, lateral aspect; **B.** Posterior end (magnified 800X), lateral aspect; **C** and **D.** Dorsomedial posterior pits, encircled and pointed by arrow, respectively. Abbreviations: *dor*, dorsal side; *ven*, ventral side.

Acknowledgements

Thanks are due to Jason Bond and Emma Jochim (UC Davis), Sandy Shanks, Thomas Pape, and the Laidlaw Smith Arthropod Morphology Grant for the generous support.

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Fly Times 73 (2024)

Composition of soldier flies (Diptera, Stratiomyidae) and hoverflies (Diptera, Syrphidae) in Malaise traps in two nature reserves in the Netherlands. Two research periods compared

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This paper is a translated and slightly revised version of the following original article:

> Van Eck, A. 2024. De samenstelling van wapenvliegen en zweefvliegen in malaisevallen in De Brand en De Kaaistoep. Twee onderzoeksperiodes naast elkaar gezet. De Vliegenmepper 33 (1): 16–28.

Key words: insect decline, De Brand, De Kaaistoep, Hallmann

Abstract

Two separate Malaise trap projects from the past (1990 and 1998 respectively), in the Netherlands, have been repeated in 2020, in order to provide data to compare the species composition and numbers of insects, then and now. This paper shows and discusses some results concerning the Diptera families Syrphidae and Stratiomyidae.

Introduction

In 1990, more than thirty years ago, a Malaise trap project was carried out in “De Brand” nature reserve near Udenhout (Netherlands, province of Noord-Brabant). This was an initiative of the insect work group of the KNNV branche in Tilburg. A booklet about this project was published in 1996 (Van Zuijlen et al. 1996). In 1991 it was decided to continue the research project at another location in “De Brand”, year-round, from which no results have been published yet.

Initiated by the Hallmann publication that caused quite a stir internationally (Hallmann et al. 2017), where it was concluded that – to say the least – things are not going well for our flying insects, the same work group has repeated its Malaise trap project in 2020 exactly thirty years later and at the same place as in 1990. Moreover, a Malaise trap study in De Kaaistoep, the water extraction area west of the city of Tilburg, was repeated that same year, being 22 years after the first study from 1998. In this article I would like to briefly highlight the results concerning two fly families from those studies, the Stratiomyidae and the Syrphidae. I will also briefly discuss the results concerning these families of the Malaise trap study carried out in De Brand in 1991.

Methods

Figure 1 indicates the locations in “De Brand” nature reserve where the Malaise traps were placed in 1990, 1991 and 2020 respectively.

The same Malaise trap, of the Townes type and all black, thus with exactly the same dimensions, was used in 1990 and 2020. Moreover, the direction in which the traps were placed was the same in both years: the 'back' of the trap was placed in a meadow against a brook forest, with the trap facing southwest. However, due to succession, it was not possible to place the trap at exactly the same spot as in 1990. During the first three trapping weeks of 2020, the trap also was located a little further from the intended spot, since, due to high surface water levels in the meadow, that was still inaccessible. The trap was placed at its intended spot on March 28, 2020 (after three samples taken);

in the same corner of the same meadow and as near as possible to the same brook forest, as in 1990 (Fig. 2). In 1991, the trap was placed at another location in "De Brand", one kilometre further east (Fig. 2) and approximately two kilometres southeast of the sand dunes of the "Drunense Duinen", and oriented to the south-southeast.

The Malaise trap used in "De Kaaistoep" was also of the Townes type, but white in both years, and of the same dimension. Figure 3 indicates the location in "De Kaaistoep" in 1998 as well as 2020. Table 1 indicates the locations with coordinates and periods the Malaise traps in "De Brand" and "De Kaaistoep" were operational.

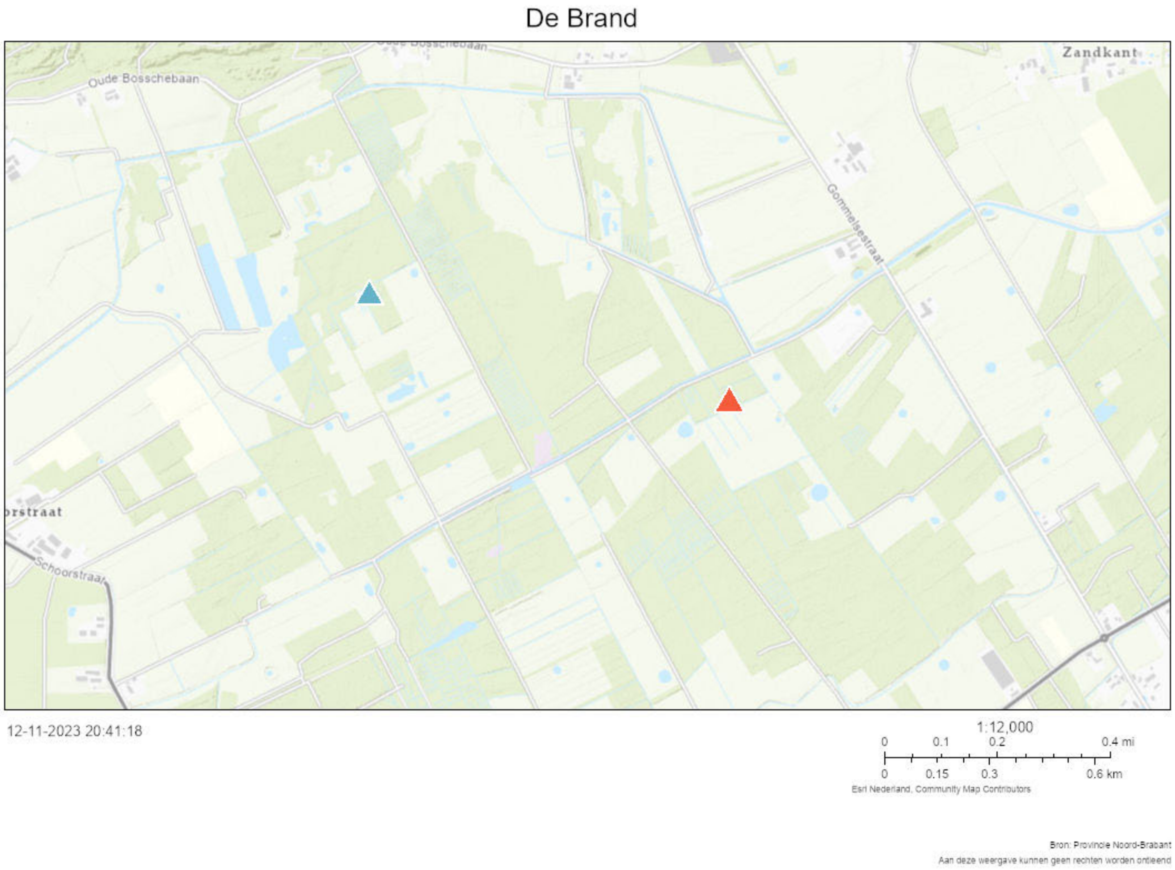

Figure 1: Position of the Malaise traps in De Brand. Turkoise triangle: 1990 and 2020. Red triangle: 1991.

Table 1: Locations with coordinates and periods the Malaise traps in "De Brand" and "De Kaaistoep" were operational, indicating the deadlines between which Stratiomyidae and Syrphidae were captured. Data from the winter period 1991–1992 (De Brand) have not been taken into account.

Study area	**Geo decimal degrees**	**Period**	**Stratiomyidae from/until**	**Hoverflies from/until**
De Brand	51.63222°N, 5.13226°E	1990 – 17-3 until 28-11	28-4 until 01-9	17-3 until 20-10
De Brand	51.62955°N, 5.14961°E	1991 – 16-3 until 14-3-1991	20-4 until 28-9	06-4 until 12-10
De Brand	51.63220°N, 5.13338°E	2020 – 01-3 until 06-12	06-6 until 15-8	21-3 until 31-10
De Kaaistoep	51.54325°N, 5.01489°E	1998 – 25-4 until 11-10	25-4 until 22-8	25-4 until 10-10
De Kaaistoep	51.54325°N, 5.01489°E	2020 – 05-3 until 30-10	18-4 until 17-9	05-3 until 30-10

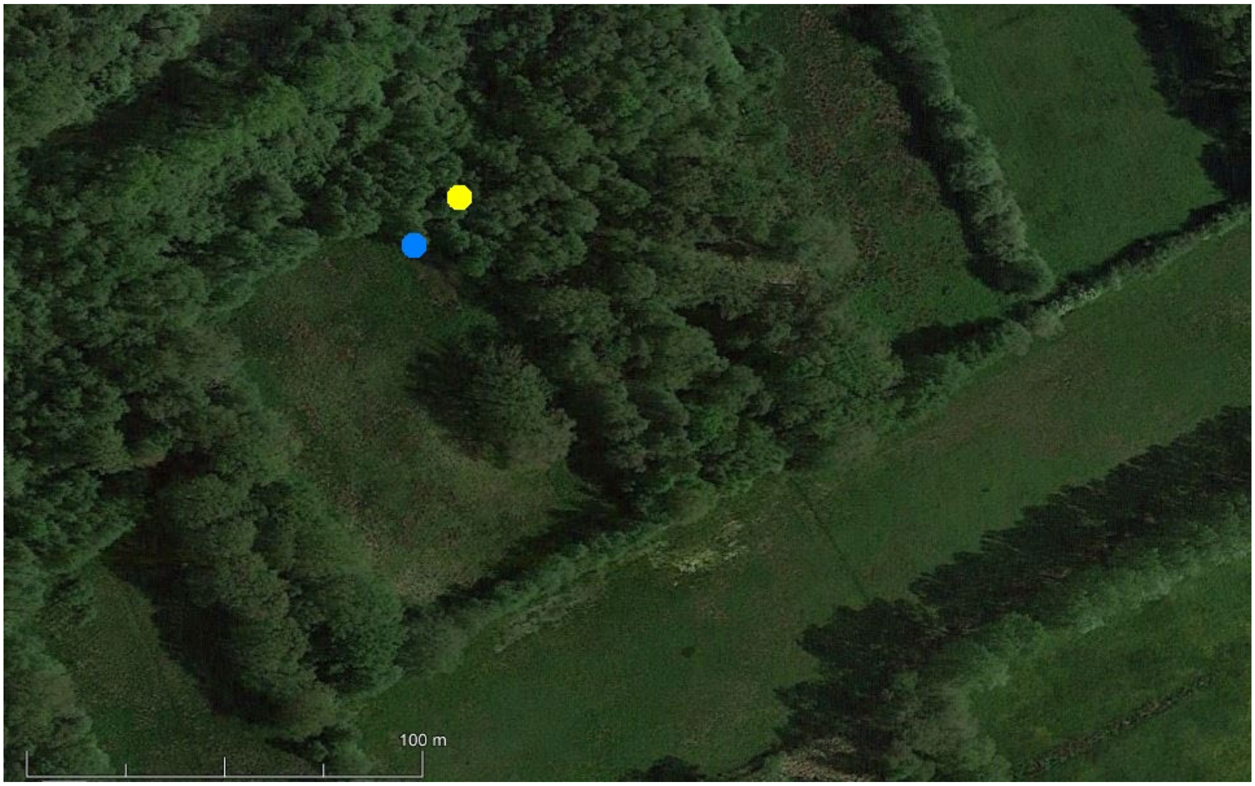

Figure 2: Location of Malaise traps in 1990 (yellow dot) and 2020 (blue dot). Aerial photo from October 2023. As can be seen, the location from 1990 has become completely overgrown with trees.

BRIEF DESCRIPTION OF THE PITCHES

De Brand

De Brand nature reserve is a peaty brook area with deciduous forests and humid meadows (Brinkhof et al 2004). It is an exponent of the small-scale Brabant brook forest landscape, located on a loamy soil alternating with sandy and marshy soils. In the deciduous forests, pedunculate oak (*Quercus rubur*) dominates, with species such as hazel (*Corylus avellana*), bracken (*Pteridium aquilinum*) and blackberry (*Rubus* sp.) in the shrub layer. Many poplars have also been planted traditionally (*Populus* sp.). The forests are characterized by the system of gullies and ridges, an old system for draining the forests. Reed land is present in the wettest places in De Brand. Seepage occurs in various places in the area, while in other places the surface water stagnates. Willow and alder brook forest (*Salix* sp. and *Alnus* sp.) develop locally. As a result of succession and desiccation, some reedland areas gradually develop into these types of brook forest. Agriculture has been established in the parts where the subsoil consists of sandy soil.

For a description of the location in 1990, I refer to Van Zuijlen et al. 1996 (in Dutch).

In 2020 Paul van Wielink and Milly Verpraet state (unpublished):
In the immediate vicinity of the Malaise trap (< 10 metres southwest) is a grassland (Fig. 4) with massive black alder (*Alnus glutinosa*) behind. On the southeast side we find an area with reeds (*Phragmites australis*) and a ditch. Nearby lies a ditch with abundant water mint (*Mentha aquatica*). Cuckoo flower (*Cardamine pratensis*) is blooming near the trap. Marsh-bedstraw (*Galium palustre*) is present in large numbers and blooms in the immediate vicinity.

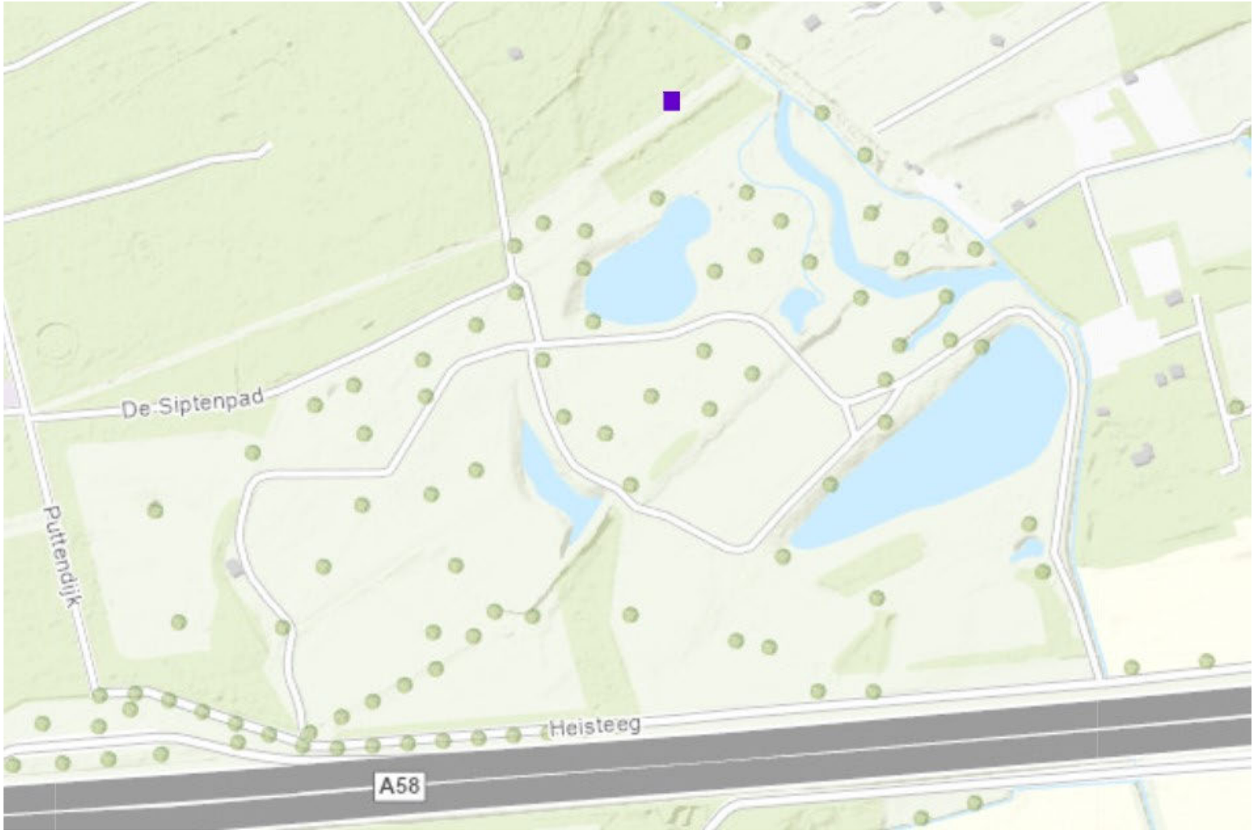

Figure 3: Placement of the Malaise traps in “De Kaaistoep”. Blue square: 1998 and 2020. The light green strip (forest) between Malaise trap and the lake was not yet there in 1998 (see text).

The grassland is still very wet at the end of April. Common rush (*Juncus effusus*) is the predominant species with >90% surface coverage, but there are also many marsh thistles (*Cirsium palustre*) and Cuckoo-flower. In many places nettle (*Urtica dioica*) accumulates. The grasses yorkchire-fog (*Holcus lanatus*) and sweet vernal grass (*Anthoxanthum odoratum*) are present everywhere. Here and there grow meadow buttercup (*Ranuculus acris*), dandelion (*Taraxacum vulgare*), sorrel (*Rumex acetosa*) and marsh-bedstraw. In the grassland, about fifty metres from the trap, are four pedunculate oaks (*Quercus robur*) of more than twenty metres high. At the beginning of August the marsh thistle has almost finished blooming. There also appears to be a lot of scattered sharp flowered rush (*Juncus acutiflorus*). Marsh-bedstraw is present and blooming everywhere, also close to the trap. Here and there purple loosestrife (*Lythrum salicaria*), meadow buttercup and great spearwort (*Ranunculus lingua*) as well as big trefoil (*Lotus pedunculatus*) and willow herb (*Epilobium* sp.). In some places the nettle (*Urtica dioica*) is large and tall, especially under the oaks east of a ditch running along the forest east of the trap. About forty metres west of the trap, a species of bentgrass (*Agrostis* sp.) is now present in abundance in the grassland.

About the location in 1991 (based on unpublished inventories by Paul van Wielink) the following can be said:
A Malaise trap was placed at a forest edge bordering an approximately 250 metres wide extensively grazed pasture (meadowgrass-ryegrass vegetation (*Poa-Lolium* spp.)). On either side of the trap (towards ENE and WSW), the border between forest and meadow extended for more than a hundred metres. There were ditches in the meadow, most of which dried up during the summer of 1991, except one about 25 metres from the trap. Vegetation in the ditches consisted of water manna grass

(*Glyceria flautans*) with marsh plants, changing to exclusively water manna grass on the southeast side. About thirty metres from the trap stood a solitary white willow (*Salix alba*) hanging over the water in the edge of the forest. The meadow contained a lot of common rush (*Juncus effusus*) and cow pies. The forest behind the trap (alder-oak forest with hazel) changes into alder with remote sedge (*Carex remota*) and bird cherry-ash tree forest (*Pruno-Fraxinetum*) with hemp-agrimony (*Eupatorium cannabinum*). Blackberries and nettles (*Urtica dioica*) grew over a large span bordering the forest. The most common trees in the forest are pedunculate oak, European ash (*Fraxinus excelsior*) and black alder.

Figure 4: **Left**. Malaise trap March/April 1990. © T.M.J. Peeters. **Right**. Malaise trap March 28, 2020. © A. van Eck

De Kaaistoep

1998

The Malaise trap was placed in a terrain managed by TWM-Gronden B.V., approximately one and a half kilometre west of the city of Tilburg and approximately 500 metres north of the highway A58 (Fig. 3). The trap was positioned with its collecting bottle facing SE on an approximately seven metre wide path (Fig. 5). This path (running from WSW to ENE) marks a water pipe that was laid in the ground in the 1970s. The reprocessed sand is very poor. At approximately ninety metres from the Malaise trap, the path ends (towards ENE) at the lowland stream 'De Oude Leij'(or Donge), which is approximately four metres wide. On the other side of the stream (more than a 100 metres from the trap) stand some old oak trees of more than twenty metres high. About fifty metres to the south, a large shallow fen of approximately 500 m², was realised in the autumn of 1994. On the north side of the path grows a wide hedge of willows, blackberries, black cherry (*Prunus serotina*) and dogwood (*Cornus* sp.). Behind the hedge lies a dry deciduous forest mainly consisting of birch (*Betula* sp.) with oak and black cherry in between, and alder. The dry deciduous forest also contains some rhododendrons (*Rhododendron* sp.) and spruces (*Picea* sp.), but these are at least twenty metres away from the trap. The trap lies in a small recess between blackberry bushes and nettles.

2020

The trap was placed on March 5, 2020 by Ron Felix and Paul van Wielink at approximately the same spot and in the same way as in 1998. As in 1998, it is white and has the same dimensions. On the south side of the path, a strip of deciduous woodland of more than 25 metres wide has developed, separating the path from the large fen and consisting of pedunculate oak, birch, willow, aspen (*Populus tremula*) and a dead white willow (*Salix alba*). Dead deciduous wood can be found everywhere on and in the subsoil, it was laid out in bundles after a major pruning in January 2011.

The path near the trap is overgrown for approximately 90% with maidenhair moss (*Fissidens adianthoides*). Furthermore ground-ivy (*Glechoma hederacea*), trancy ragwort (*Jacobaea vulgaris*), field wood-rush (*Luzula campestris*) and catsear (*Hypochaeris radicata*) is found there. On August 27, 2020, less than ten metres from the trap, the following plants were found: sandbar willow (*Salix exigua*), nettle, bird cherry (*Prunus* spp.), blackberry, poplar (*Populus* spp.), pedunculate oak, elderberry (*Sambucus nigra*), birch, alder, rhododendron, hazel, laurel (*Laurus nobilis*), common yarrow (*Achillea millefolium*) and lots of dead deciduous wood. A little further away grew beech (*Fagus* sp.), aspen and bracken.

Figure 5: **Left**. Malaise trap on July 19, 1998. **Right**. Malaise trap on September 10, 2020. Photos © P. v. Wielink.

RESULTS IN DE BRAND

The 1991 Malaise trap study took place in a different part of the nature reserve. The results of this study have not previously been published. In the context of this article, the study provides some additional information on the community of soldier flies and hoverflies in De Brand at that time; one might also compare the results with those from 1990.

Table 2: Number and species of soldier flies and hoverflies, respectively, in De Brand, during the Malaise trap research in 1990, 1991 and 2020.

Family	Totals 1990	Totals 2020	Species 1990	Species 2020	Totals 1991	Species 1991
Stratiomyidae	456	21	10	6	1985	12
Syrphidae	3358	1384	77	58	1059	48

The soldier flies in De Brand

Changes in the soldier fly community in De Brand

The differences are clear. Species from swampy, herb-rich grasslands are virtually absent in 2020. Numerous species, such as *Chloromyia formosa* and species from the genera *Beris* and *Microchrysa*, were almost not caught in 2020.

Table 3: Species and numbers of soldier flies, caught in the Malaise trap in De Brand in 1990, 1991 and 2020. The years 1990 and 2020 have been placed next to each other for clarity, as they are (almost) from the same spot.

Genus	**Species**	**Author**	**1990**	**2020**	**1991**
Beris	*chalybeata*	(Forster)	126		1046
Beris	*clavipes*	(Linnaeus)	35	2	173
Beris	*vallata*	(Forster)	58	7	142
Chloromyia	*formosa*	(Scopoli)	57		482
Chorisops	*tibialis*	(Meigen)	1	6	21
Microchrysa	*cyaneiventris*	(Zetterstedt)	123		38
Microchrysa	*flavicornis*	(Meigen)	5	1	
Microchrysa	*polita*	(Linnaeus)	15	3	7
Oxycera	*leonina*	(Panzer)			1
Pachygaster	*atra*	(Panzer)			47
Pachygaster	*leachii*	Curtis	1		
Sargus	*bipunctatus*	(Scopoli)			1
Sargus	*flavipes*	Meigen		2	2
Sargus	*iridatus*	(Scopoli)	35		25
Total			456	21	1985
Number of species			10	6	12

The hoverflies in De Brand

(the nomenclature has been adjusted to the most recent insights (Speight 2020)

Changes in hoverfly community in De Brand

In all species groups large differences in catches between 1990 and 2020 can be observed. To keep the graph (Fig. 6) presentable, *Neoascia tenur*, whose larvae live aquatic, has been excluded; 1050 specimens of this species were collected in 1990, and 235 in 2020. In 1991, only 33 specimens of *N. tenur* were collected, so there may be a locality effect. Nevertheless, we see major differences in all species groups between 1990 and 2020. Numbers are at least 50% lower; species with phytophagous larvae (*Cheilosia*) are virtually absent in 2020, and 1991. Some zoophagous species also show large differences between 1990 and 2020, e.g. *Eupeodes corollae*, *Melanostoma scalare* and *Syrphus* species. *Trichopsomyia flavitarsis*, still found in numbers in 1990, is completely absent from the 2020 Malaise trap catches. This also applies to the two species of *Pyrophaena*. The differences in numbers in *Rhingia campestris*, with its terrestrial saprophagous larvae living in cow dung, may be explained by the termination of grazing by cows in the intervening period.

Changes in the surrounding area of De Brand

Overall, little has changed in the areas surrounding the Malaise trap; the place is still very centrally located in relation to the surrounding countryside, the buffer zone to agricultural areas has remained the same or even grown somewhat thanks to extensification of a number of nearby fields. As far as management is concerned, there are some changes. In recent years, measures have been taken to restore hydrology and re-wet the nature reserve and to eliminate eutrophication via surface water. Desiccation and eutrophication had already started in the 1980s and 1990s and continued in the years that followed. Nevertheless, in the period around 1990 there was still nutrient-poor seepage in the surrounding ditches. Measures are now being taken to turn the tide , which is also noticeable at the research location. Where the Malaise trap stood in 1990, it was impossible to keep dry feet in 2020; In 2020, a considerable part of the northeast corner of the meadow was partly submerged until well into springtime (Fig. 7).

Table 4: Species and numbers of hoverflies, caught in the Malaise trap in De Brand in 1990, 1991 and 2020. Species are ordered by larval lifestyle (Reemer et al 2009) and within that alphabetically. The years 1990 and 2020 have been placed next to each other for clarity, as this concerns a repeated research at (almost) the same spot. (see notes after table)

Genus	**Species**	**Author**	**L**	**1990**	**2020**	**1991**
Anasimyia	*interpuncta*	(Harris)	as	50	22	6
Eristalinus	*sepulchralis*	(Linnaeus)	as	10	40	
Eristalis	*arbustorum*	(Linnaeus)	as	5	1	
Eristalis	*horticola*	(DeGeer)	as	4		
Eristalis	*intricaria*	(Linnaeus)	as	2	34	1
Eristalis	*nemorum*	(Linnaeus)	as	2		
Eristalis	*pertinax*	(Scopoli)	as	7	33	2
Eristalis	*tenax*	(Linnaeus)	as	3		1
Eurimyia	*lineata*	(Fabricius)	as	32	12	
Helophilus	*hybridus*	Loew	as	18	13	
Helophilus	*pendulus*	(Linnaeus)	as	324	167	192
Helophilus	*trivittatus*	(Fabricius)	as	1		
Lejogaster	*metallina*	(Fabricius)	as	3		1
Melanogaster	*hirtella*	Loew	as	97	1	
Neoascia	*interrupta*	(Meigen)	as		58	
Neoascia	*meticulosa*	(Scopoli)	as	46		3
Neoascia	*podagrica*	(Fabricius)	as	10		2
Neoascia	*tenur*	(Harris)	as	1050	285	33
Orthonevra	*geniculata*	Meigen	as	3	7	
Parhelophilus	*frutetorum*	(Fabricius)	as	15	1	1
Parhelophilus	*versicolor*	(Fabricius)	as	4	1	
Cheilosia	*albipila*	Meigen	fy	5		
Cheilosia	*albitarsis*	(Meigen)	fy	68	2	8
Cheilosia	*barbata*	Loew	fy	1		
Cheilosia	*fraterna*	(Meigen)	fy	4	3	
Cheilosia	*impressa*	Loew	fy	10		
Cheilosia	*pagana*	(Meigen)	fy	11	1	5
Cheilosia	*vernalis*	(Fallén)	fy		1	
Brachyopa	*pilosa*	Collin	ts	1		
Brachypalpoides	*lentus*	(Meigen)	ts			4
Brachypalpus	*laphriformis*	(Fallén)	ts	1	1	
Chalcosyrphus	*nemorum*	(Fabricius)	ts	36	73	
Ferdinandea	*cuprea*	(Scopoli)	ts	2	2	6
Matsumyia	*berberina*	(Fabricius)	ts			5
Myathropa	*florea*	(Linnaeus)	ts	1	1	
Rhingia	*campestris*	Meigen	ts	114	4	56
Syritta	*pipiens*	(Linnaeus)	ts	16	1	
Temnostoma	*bombylans*	(Fabricius)	ts	1		1
Tropidia	*scita*	(Harris)	ts	25	10	9
Volucella	*bombylans*	(Linnaeus)	ts	5	1	3
Volucella	*pellucens*	(Linnaeus)	ts	1		1
Xylota	*abiens*	Meigen	ts		2	2
Xylota	*florum*	(Fabricius)	ts	1		
Xylota	*meigeniana*	Stackelberg	ts	6		
Xylota	*segnis*	(Linnaeus)	ts	13	8	20
Xylota	*sylvarum*	(Linnaeus)	ts	4	1	11
Xylota	*tarda*	Meigen	ts			1
Chrysotoxum	*bicinctum*	(Linnaeus)	zoö	5		13
Chrysotoxum	*cautum*	(Harris)	zoö		1	

Table 4: (continued)

Genus	**Species**	**Author**	**L**	**1990**	**2020**	**1991**
Dasysyrphus	_albostriatus_	(Fallén)	zoö	7	1	
Dasysyrphus	_tricinctus_	(Fallén)	zoö	3		
Dasysyrphus	_venustus_*	(Meigen)	zoö	1		4
Epistrophe	_eligans_	(Harris)	zoö		1	
Epistrophe	_nitidicollis_**	(Meigen)	zoö	3	1	
Episyrphus	_balteatus_	(DeGeer)	zoö	37	50	212
Eupeodes	_bucculatus_	(Collin)	zoö	2		
Eupeodes	_corollae_	(Fabricius)	zoö	124	12	79
Eupeodes	_latifasciatus_	(Macquart)	zoö	29	3	2
Eupeodes	_luniger_	(Meigen)	zoö	4	4	1
Melanostoma	_mellinum_	(Linnaeus)	zoö	307	202	111
Melanostoma	_scalare_	(Fabricius)	zoö	173	7	4
Meligramma	_triangulifera_	(Zetterstedt)	zoö		6	
Meliscaeva	_auricollis_	(Meigen)	zoö	5		
Neocnemodon	_pubescens_	(Del. & Ps-W)	zoö	1		
Neocnemodon	_sp._		zoö		6	2
Paragus	_haemorrhous_	Meigen	zoö	5	5	2
Paragus	_sp._		zoö			2
Pipiza	_accola_	Violovitsh	zoö		1	
Pipiza	_noctiluca_	(Linnaeus)	zoö	2	2	3
Pipiza	_notata_	Meigen	zoö	1	1	6
Pipizella	_viduata_***	(Linnaeus)	zoö	12		
Pipizella	_virens_	(Fabricius)	zoö		3	
Platycheirus	_albimanus_	(Fabricius)	zoö	33	1	3
Platycheirus	_angustatus_	(Zetterstedt)	zoö	125	121	151
Platycheirus	_clypeatus_	(Meigen)	zoö	79	2	33
Platycheirus	_europaeus_	Goeldlin et al	zoö		5	
Platycheirus	_fulviventris_	(Macquart)	zoö	22	32	
Platycheirus	_occultus_	Goeldlin et al	zoö	15	19	
Platycheirus	_peltatus_	(Meigen)	zoö	5		1
Platycheirus	_scambus_	(Staeger)	zoö	2		10
Platycheirus	_scutatus_	(Meigen)	zoö	9	1	2
Pyrophaena	_granditarsa_	(Foerster)	zoö	122		5
Pyrophaena	_rosarum_	(Fabricius)	zoö	31		
Scaeva	_pyrastri_	(Linnaeus)	zoö		1	1
Scaeva	_selenicita_	(Meigen)	zoö	1		
Sphaerophoria	_batava_	Goeldlin	zoö	2		
Sphaerophoria	_scripta_	(Linnaeus)	zoö	25	92	29
Sphaerophoria	_virgata_	Goeldlin	zoö	1		
Syrphus	_ribesii_	(Linnaeus)	zoö	30	7	4
Syrphus	_torvus_	Osten-Sacken	zoö	13		
Syrphus	_vitripennis_	Meigen	zoö		10	1
Trichopsomyia	_flavitarsis_	(Meigen)	zoö	99		4
Trichopsomyia	_lucida_****	(Meigen)	zoö	11		
Xanthandrus	_comtus_	(Harris)	zoö	1	1	
Total				3358	1384	1059
Number of species				77	58	48

* : This specimen was checked and confirmed. *Dasysyrphus neovenustus* has recently been confirmed from De Brand as well.

** : These specimens have been checked for *Epistrophe olgae* Mutin, recently confirmed from the Netherlands, and the identification is correct.

*** : 6 male and 6 female. The chance that *P. virens* was found among the females can easily be ruled out on the basis of distribution and hand-collecting data from that period (Reemer et al 2009).

****: an inspection in 2018, for the purpose of a review of the genus *Trichopsomyia* (Van Steenis 2018), made clear that these determinations were incorrect. They turned out to be nine males of *T. flavitarsis* and two females of *Pipizella* (pers. comm. Jeroen van Steenis, October 27, 2023). As a consequence *T. lucida* is removed from the list of hoverflies found in De Brand (Van Zuijlen et al 2006).

L: larval lifestyle, species groups following the classification in Reemer et al (2009): ts, terrestrial saprophage; as, aquatic saprophage; phy, phytophage; zoo, zoophagous.

The meadow in front of the trap was lightly grazed by some cows for a very short part of the year before and in 1990. In 1990 it was mowed once and the clippings were removed. It was a relatively open grassland, rich in, for example, buttercups (*Ranunculus* spp.), cuckoo-flower (*Cardamine pratensis*) and sorrel (*Rumex acetosa*) (Fig. 8). In 2020, the situation had changed drastically. Cows no longer grazed, so it had become an unmowed,overgrown,, grassland , in which common rush (*Juncus effusus*), marsh thistle (*Cirsium palustre*) and tall grasses predominate (Fig. 8). All in all, the grassland in front of the trap has become much poorer in species. Common rush and field wood-rush have become dominant. Nettle and marsh thistle have also increased significantly. The diversity of species has become much poorer. Cuckoo-flower and buttercup are still present to a limited extent, sorrel and common comfrey (*Symphytum officinale*) have almost disappeared. Of course thirty years later, the surrounding forest and the oaks in the meadow have aged. Some parts of the ditch have disappeared as a result of hydrosere. Old reed land NW of the meadow greatly reduced and got replaced by willow forest. The adjacent brook forest has undergone a significant transformation, from willow-dominated brook forest to alder brook forest with black alder (*Alnus glutinosa*).

RESULTS IN DE KAAISTOEP

The soldier flies in De Kaaistoep

Changes in the soldier fly community in De Kaaistoep

The differences between the results from 1998 and 2020 are remarkable. The number of soldier fly species has dropped from nine to five. And the total number of specimens in 2020 is less than 4% from that in 1998. Genera present with large numbers in 1998, *Beris* and *Microchrysa*, are completely absent in 2020.

Table 5: Species and numbers of soldier flies, caught in the Malaise trap in "De Kaaistoep" in 1998 and 2020.

Genus	Species	Author	1998	2020
Beris	*chalybata*	(Forster, 1771)	1512	
Beris	*clavipes*	(Linnaeus, 1767)	3	
Beris	*vallata*	(Forster, 1771)	30	
Chloromyia	*formosa*	(Scopoli, 1763)	3	
Chorisops	*tibialis*	(Meigen, 1820)	10	9
Microchrysa	*cyaneiventris*	(Zetterstedt, 1842)	3	
Microchrysa	*polita*	(Linnaeus, 1758)	45	
Oxycera	*leonina*	(Panzer, 1798)	2	2
Pachygaster	*atra*	(Panzer, 1798)	16	45
Pachygaster	*leachii*	Curtis, 1824		1
Sargus	*bipunctatus*	(Scopoli, 1763)		4
Total			**1624**	**61**
Number of species			**9**	**5**

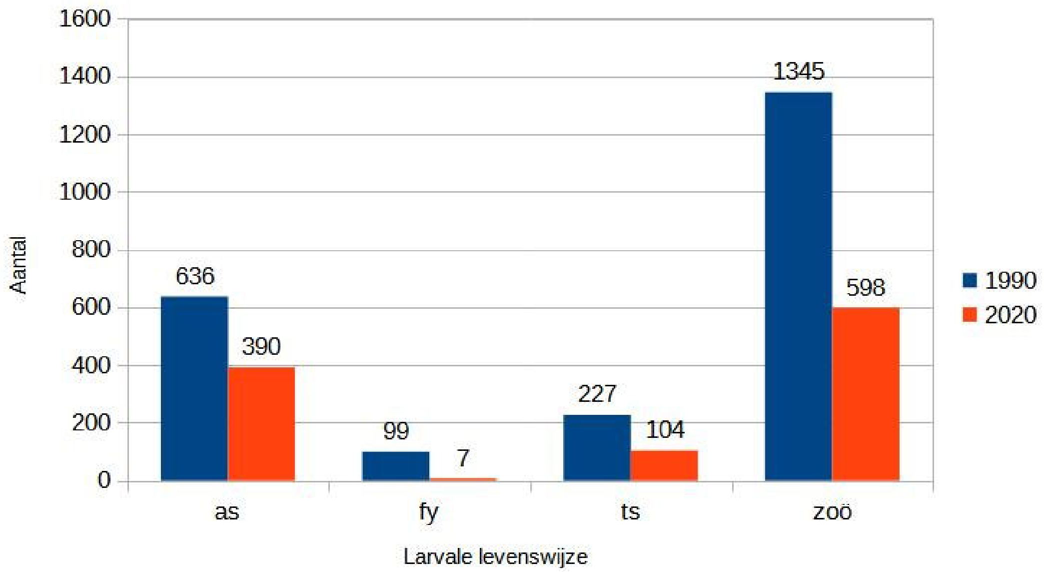

Figure 6: Numbers of hoverflies in the Malaise trap in De Brand, sorted by larval lifestyle, in 1990 and 2020. *Neoascia tenur* not taken into account. See table 4 for the legend. Aantal = Numbers; Larvale levenswijza = Larval lifestyle.

Figure 7: **Left**: Northeast corner of the meadow soaking wet on March 1, 2020. © M. Oosthoek. **Right**: The meadow in front of the Malaise trap on June 20, 2020. © A. van Eck

Figure 8: **Left**. Malaise trap May/June 1990. © T.M.J. Peeters. **Right**. Malaise trap on June 13, 2020. © A. van Eck

The hoverflies in De Kaaistoep

Table 6: Species and numbers of hoverflies, caught in the Malaise trap in De Kaaistoep in 1998 and 2020. Species are ordered by larval lifestyle (Reemer et al 2009) and within that alphabetically. (see notes after table)

Genus	Species	Author	L	1998	2020
Eristalinus	*sepulchralis*	(Linnaeus)	as		2
Eristalis	*arbustorum*	(Linnaeus)	as	1	2
Eristalis	*intricaria*	(Linnaeus)	as	1	1
Eristalis	*nemorum*	(Linnaeus)	as		1
Eristalis	*pertinax*	(Scopoli)	as	10	7
Eristalis	*tenax*	(Linnaeus)	as	6	7
Helophilus	*hybridus*	Loew	as	2	
Helophilus	*pendulus*	(Linnaeus)	as	39	17
Melanogaster	*hirtella*	Loew	as		3
Neoascia	*podagrica*	(Fabricius)	as	1	1
Cheilosia	*albipila*	Meigen	fy		3
Cheilosia	*albitarsis*	(Meigen)	fy	14	
Cheilosia	*bergenstamm*	Becker	fy		3
Cheilosia	*grossa*	(Fallén)	fy		2
Cheilosia	*pagana*	(Meigen)	fy	5	
Cheilosia	*urbana*	(Meigen)	fy		2
Cheilosia	*variabilis*	(Panzer)	fy		1
Eumerus	*funeralis*	Meigen	fy	1	
Eumerus	*sp.*		fy	1	
Eumerus	*strigatus*	(Fallén)	fy	1	2
Merodon	*equestris*	(Fabricius)	fy	4	1
Pelecocera	*tricincta*	Meigen	fy		1
Brachyopa	*bicolor*	(Fallén)	ts	3	
Brachyopa	*pilosa*	Collin	ts		1
Brachypalpoides	*lentus*	(Meigen)	ts	2	
Callicera	*fagesii*	Guérin-Méneville	ts	1	
Ceriana	*conopsoides*	(Linnaeus)	ts	1	3
Chalcosyrphus	*nemorum*	(Fabricius)	ts	3	1
Chalcosyrphus	*piger*	(Fabricius)	ts		2
Ferdinandea	*cuprea*	(Scopoli)	ts	2	1
Myathropa	*florea*	(Linnaeus)	ts	7	2
Rhingia	*campestris*	Meigen	ts	6	
Syritta	*pipiens*	(Linnaeus)	ts	11	
Tropidia	*scita*	(Harris)	ts		17
Xylota	*segnis*	(Linnaeus)	ts		1
Baccha	*elongata*	(Fabricius)	zoö	29	
Chrysotoxum	*bicinctum*	(Linnaeus)	zoö	53	
Chrysotoxum	*cautum*	(Harris)	zoö	22	1
Chrysotoxum	*festivum*	(Linnaeus)	zoö		1
Chrysotoxum	*vernale*	Loew	zoö	17	
Dasysyrphus	*albostriatus*	(Fallén)	zoö	3	2
Dasysyrphus	*venustus**	(Meigen)	zoö	2	
Didea	*alneti*	(Fallén)	zoö	1	
Didea	*fasciata*	Macquart	zoö	2	
Didea	*intermedia*	Loew	zoö	35	9
Epistrophe	*eligans*	(Harris)	zoö	3	4
Epistrophe	*melanostoma*	(Zetterstedt)	zoö	2	
Epistrophe	*nitidicollis***	(Meigen)	zoö	5	41

Table 6 (continued)

Genus	**Species**	**Author**	**L**	**1998**	**2020**
Episyrphus	*balteatus*	(DeGeer)	zoö	221	132
Eupeodes	*corollae*	(Fabricius)	zoö	9	80
Eupeodes	*latifasciatus*	(Macquart)	zoö	9	2
Eupeodes	*luniger*	(Meigen)	zoö	8	54
Eupeodes	*nielseni*	(Dusek & Láska)	zoö		1
Fagisyrphus	*cinctus*	(Fallén)	zoö	8	
Lapposyrphus	*lapponicus*	(Zetterstedt)	zoö		3
Melanostoma	*mellinum*	(Linnaeus)	zoö	97	89
Melanostoma	*scalare*	(Fabricius)	zoö	93	18
Meligramma	*triangulifera*	(Zetterstedt)	zoö	1	1
Meliscaeva	*auricollis*	(Meigen)	zoö	34	12
Microdon	*analis****	(Macquart)	zoö	1	
Neocnemodon	*latitarsis*	(Egger)	zoö		1
Neocnemodon	*sp.*		zoö	7	2
Paragus	*haemorrhous*	Meigen	zoö	37	87
Paragus	*pecchiolii*	Rondani	zoö		1
Paragus	*sp.*		zoö	24	54
Parasyrphus	*punctulatus*	(Verrall)	zoö	6	
Pipiza	*festiva*	Meigen	zoö	1	
Pipiza	*lugubris*	(Fabricius)	zoö	5	
Pipiza	*noctiluca*	(Linnaeus)	zoö		2
Pipizella	*cf. virens*	(Fabricius)	zoö		1
Pipizella	*sp.*		zoö		2
Pipizella	*viduata*	(Linnaeus)	zoö	8	2
Platycheirus	*albimanus*	(Fabricius)	zoö	78	11
Platycheirus	*angustatus*	(Zetterstedt)	zoö	99	10
Platycheirus	*clypeatus*	(Meigen)	zoö	4	
Platycheirus	*europaeus*	Goeldlin et al.	zoö	2	
Platycheirus	*peltatus*	(Meigen)	zoö	4	
Platycheirus	*scutatus*	(Meigen)	zoö	22	3
Scaeva	*pyrastri*	(Linnaeus)	zoö		1
Scaeva	*selenitica*	(Meigen)	zoö	10	12
Sphaerophoria	*batava*	Goeldlin	zoö	30	
Sphaerophoria	*scripta*	(Linnaeus)	zoö	91	13
Sphaerophoria	*sp.*		zoö	38	101
Syrphus	*ribesii*	(Linnaeus)	zoö	59	3
Syrphus	*torvus*	Osten-Sacken	zoö	1	
Syrphus	*vitripennis*	Meigen	zoö	8	7
Trichopsomyia	*joratensis*	Goeldlin	zoö	2	
Triglyphus	*primus*	Loew	zoö	1	
Volucella	*bombylans*	(Linnaeus)	zoö	1	
Xanthandrus	*comtus*	(Harris)	zoö	1	1
Xanthogramma	*pedissequum*	(Harris)	zoö	4	1
Xanthogramma	*stackelbergi*	Violovitsh	zoö		3
Total				1320	852
Number of species				65	57 (58)

*: These specimens have been checked for *Dasysyrphus neovenustus* and the identification is confirmed.

** : These specimens have been checked for *Epistrophe olgae* Mutin and the identification is confirmed.

*** : This probably concerns *Microdon major* Andries, a species that was recently confirmed for De Kaaistoep on the basis of puparia found.

Table 6 (continued)

Kaaistoep MT 2020 Syrph outside data 1998			
Area	**Date**	**Taxon**	**N**
De Kaaistoep	5.iii.2020 until 12.iii.2020	*Cheilosia grossa*	1
De Kaaistoep	12.iii.2020 until 19.iii.2020	*Cheilosia grossa*	1
De Kaaistoep	19.iii.2020 until 26.iii.2020	*Eristalis intricaria*	1
De Kaaistoep	19.iii.2020 until 26.iii.2020	*Eupeodes corollae*	1
De Kaaistoep	26.iii.2020 until 2.iv.2020	*Eristalis pertinax*	2
De Kaaistoep	26.iii.2020 until 2.iv.2020	*Eristalis arbustorum*	1
De Kaaistoep	26.iii.2020 until 2.iv.2020	*Cheilosia albipila*	1
De Kaaistoep	2.iv.2020 until 9.iv.2020	*Cheilosia albipila*	1
De Kaaistoep	2.iv.2020 until 9.iv.2020	*Cheilosia albipila*	1
De Kaaistoep	2.iv.2020 until 9.iv.2020	*Eristalis pertinax*	3
De Kaaistoep	2.iv.2020 until 9.iv.2020	*Melanostoma mellinum*	1
De Kaaistoep	2.iv.2020 until 9.iv.2020	*Platycheirus albimanus*	1
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Cheilosia urbana*	2
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Epistrophe nitidicollis*	1
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Eristalis pertinax*	1
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Helophilus pendulus*	2
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Melanostoma mellinum*	4
De Kaaistoep	9.iv.2020 until 16.iv.2020	*Paragus haemorrhous*	1
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Cheilosia bergenstammi*	2
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Epistrophe nitidicollis*	1
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Epistrophe eligans*	1
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Epistrophe nitidicollis*	2
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Eristalis nemorum*	1
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Melanostoma scalare*	1
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Paragus haemorrhous*	4
De Kaaistoep	16.iv.2020 until 23.iv.2020	*Syrphus vitripennis*	1
De Kaaistoep	8.x.2020 until 15.x.2020	*Melanostoma mellinum*	1
De Kaaistoep	8.x.2020 until 15.x.2020	*Melanostoma scalare*	1
De Kaaistoep	15.x.2020 until 22.x.2020		0
De Kaaistoep	22.x.2020 until 29.x.2020	*Eupeodes corollae*	1
De Kaaistoep	22.x.2020 until 29.x.2020	*Melanostoma mellinum*	3

Changes in the hoverfly community in De Kaaistoep

The number of hoverfly species is somewhat lower in 2020 than in 1998. The number of specimens collected in 2020 is about 65% of the numbers in 1998.

Figure 9 shows that especially the numbers of species whose larvae are zoophagous have dropped. This is clearly visible in the genera *Chrysotoxum* and *Platycheirus. Baccha elongata*, caught in high numbers in 1998, was not found at all in 2020. *Sphaerophoria batava* was no longer found in 2020. On the other hand, some species from this group actually showed higher numbers in 2020, e.g. the genera *Eupeodes* and *Paragus*. And even more striking, *Tropidia scita*, a species with aquatic larvae, made its first appearance in the Malaise trap of 2020.

Changes in the surrounding area of De Kaaistoep

In 22 years, the site has become seriously roughened. The path, originally 7 metres wide, is only 2 to 3 metres wide in 2020, mainly due to an overgrowth of bramble thickets and nettles. The increase in blackberries, nettles, aspen and bracken indicates an increase in nitrogen in the soil. Elsewhere in De Kaaistoep, for example, the elder trees have increased enormously in number.

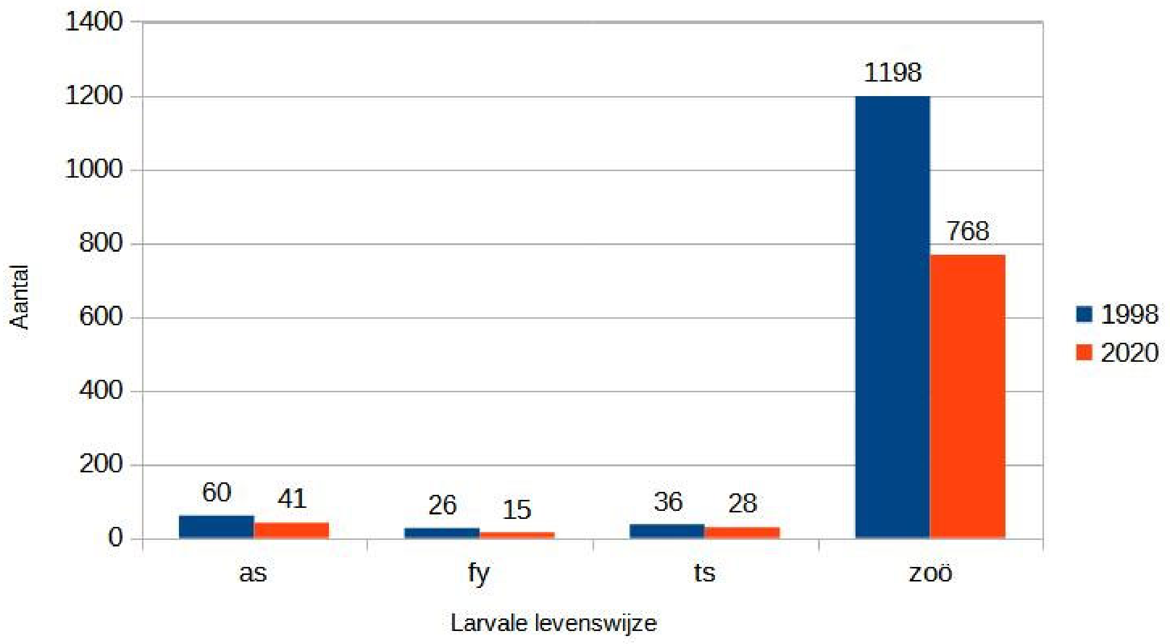

Figure 9: Species and numbers of hoverflies in the Malaise trap in De Kaaistoep, sorted by larval lifestyle, in 1998 and 2020. Legend: see table 4. Aantal = Numbers; Larvale levenswijza = Larval lifestyle.

As a further consequence of the coarsening and succession the area between the Malaise trap and the opposite fen has overgrown heavily (Fig. 5). Over the years, some developments have taken place throughout the area in terms of management.

Between 1998 and 2020, various changes have been made to the site, aiming to promote biodiversity. For example, a large part of the nearby stream, which drained its water by a straight channel through the meadows and forest, was deepened, while meanders were constructed and more swamp-like situations were created. Measures have also been taken to (re)introduce, for example, European tree frog (*Hyla arborea*) and garlic toad (*Pelobates fuscus*). It would take a bit far to cover all changes; for more information we refer to the Kaaistoep reports available online from the KNNV Tilburg department (https://tilburg.knnv.nl/publicaties/).

Some meteorological data
As an illustration , two periods of four years each are compared in terms of precipitation and temperature (Fig. 10). A three years period prior to the Malaise trap projects in De Brand, in 1990 and 2020 respectively are taken into account. Both research areas fall under the Gilze-Rijen weather station, so the data also apply to the 2020 Malaise trap project in De Kaaistoep.

April and May were relatively dry months in both four year periods, but immediately noticeable is the extreme drought during the month of May in the period 2017–2020. The other months show a more even picture, in which relative differences in precipitation are significantly smaller. This means that the period 1987–1990 had fairly dry months of April and May, while the drought of April in the period 2017–2020 was followed by an even drier month of May.

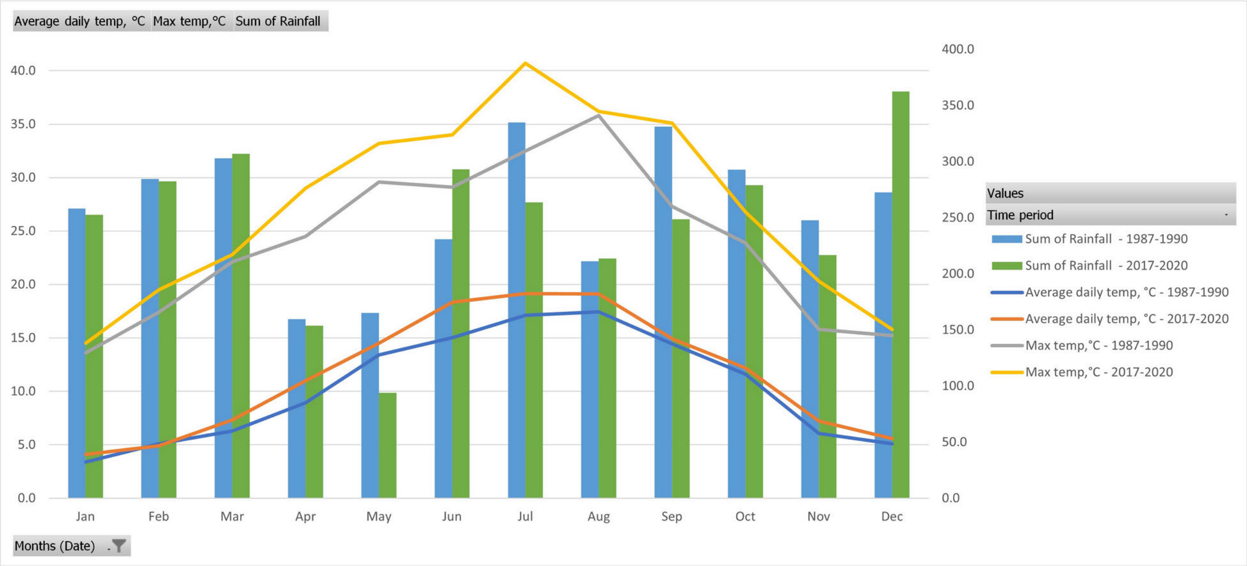

Figure 10. Two periods of four consecutive years each, compared in terms of cumulative precipitation, average daily temperature and maximum temperature, plotted per month. On the x-axis = month. y-axis = temperature in degrees Celsius. z-axis = sum of precipitation in mm. The weather station = Gilze-Rijen. (source: KNMI)

Univoltine spring species could be particularly affected by unfavourable conditions during several years in a row; drought, for example, has consequences for larval development, since suitable substrates, in which the larvae develop, become less available – think of pools, for example, or marsh – and food for the larvae might be less available. Species filtering for micro-organisms that run out of substrate will therefore not find any food. Larvae can often survive for some time under unfavourable conditions, but if the situation does not improve in time, they will still die. And if the development of sufficiently large numbers of aphids remain inadequate, for example as a result of drought, zoophagous larvae will starve.

Furthermore, the line graph clearly shows that the climate has become milder in thirty years; In the period 2017–2020, Gilze-Rijen weather station, near Tilburg, tipped 40 degrees Celsius for the first time since the beginning of climate monitoring in the Netherlands.

In Fig. 11 we also recognise the dry period of April and May 2020 in the amount of sunshine hours. This period actually started in March, after excessive rainfall in February, which continued in March. After that, the summer also remained relatively sunny, with an extreme in September 2020. Incidentally, 1990 apparently also had a fairly sunny spring, compared to 1991 and 1998.

For more detail, let us look at some overviews concerning the research years in question, 1990, 1991, 1998 and 2020 (Fig. 12a–d). Now we see that both 1990, 1991 and 2020 had a very dry spring. Dry to very dry periods occur in all years, except 1998. In three of the four years, maximum temperatures reached high averages early in the year, while the average maximum of 25 degrees gradually occurred earlier in the spring.

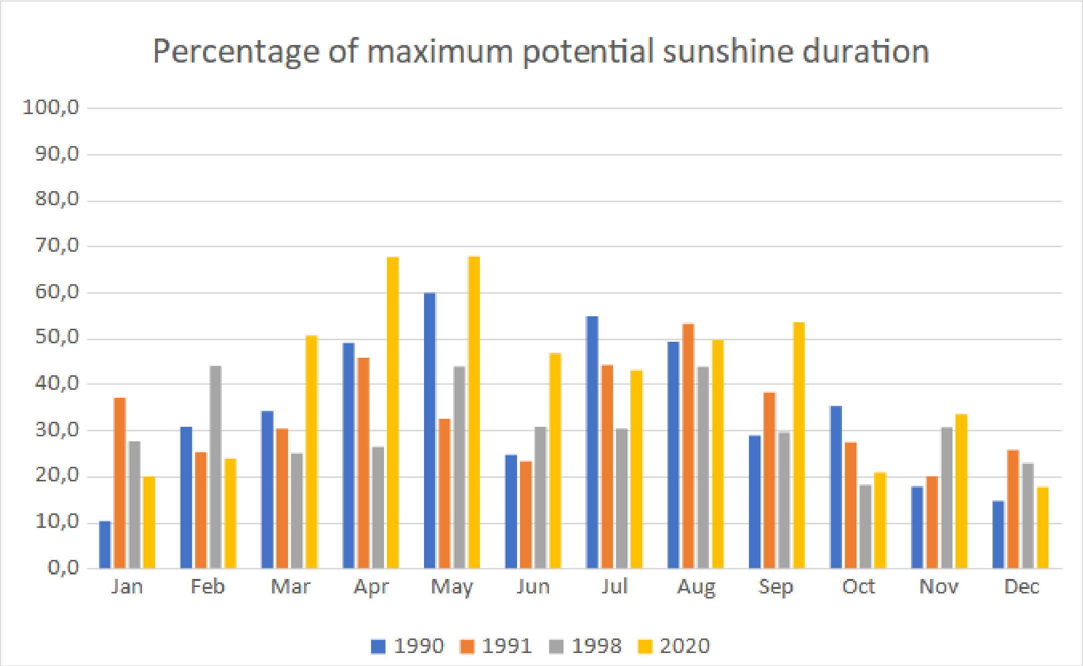

Figure 11: Percentage of maximum potential sunshine duration per month, for the years 1990, 1991, 1998 and 2020, at Gilze-Rijen weather station. x-axis = month. y-axis = percentage. (source: KNMI)

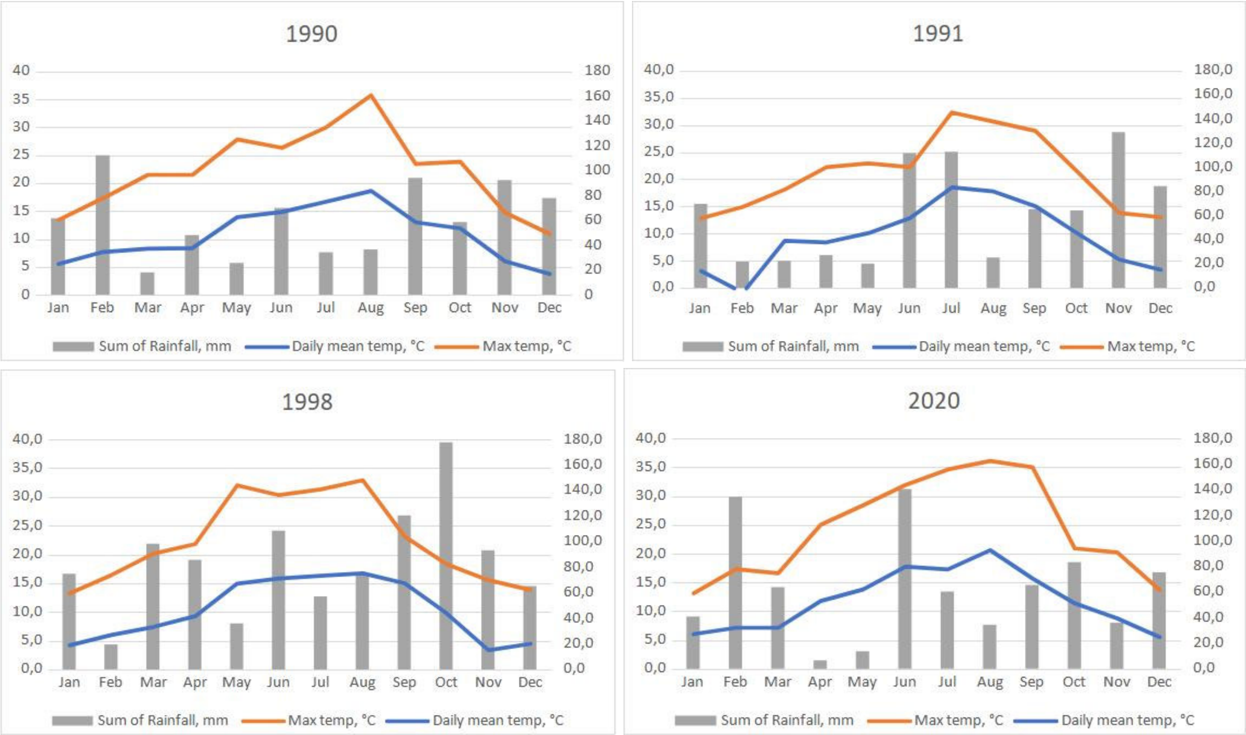

Figure 12a–d. Amount of precipitation, average daily temperature and average maximum temperature, shown per month (Gilze-Rijen weather station). x-axis = month, y-axis = degrees Celsius, z-axis = cumulative precipitation in mm. (source: KNMI). **12a** = 1990. **12b** = 1991. **12c** = 1998. **12d** = 2020.

DISCUSSION

De Brand

Changed management and the unmistakable effects of succession and overgrowth make it difficult to provide clear reasons for a number of changes in the hoverfly composition. If we look at the entire De Brand nature reserve, we see a clear effect of the aging forest on the insect composition, whether they are hoverflies, beetles, or otherwise. This effect can be seen in hand catches and on the biodiversity recording website https://waarneming.nl. Species characteristic for old or decaying trees (pedunculate oak, poplar) have increased and in recent years – similar to the national trend – species can be found in De Brand that did not occur here thirty years ago. Almost without exception these are species favouring old forests with (many) dead or dying trees. However, several xylobiont species have also declined and even disappeared; the reasons for this can only be guessed at. As an example we name *Xylota meigeniana* which appears to have become extinct in the area. Various *Xylota* species have a hard time in dry years, for them the summer situation seems to be decisive, despite the increased proportion of dead wood. This also seems to apply to *Temnostoma bombylans*.

There also appears to be a (regional) change in the grasslands, which may be due to a combination of changed management and climate change. Is the expansion of a species such as *Pipizella virens* – new in De Brand and De Kaaistoep – due to changed grassland management? The species is also increasing in the adjacent Belgian Kempen (personal communication by Frank Van de Meutter).

Particularly striking in the results of the Malaise trap project in De Brand is that soldier flies and hoverflies from herb-rich grasslands, and species whose larvae have an aquatic or semi-aquatic saprophagous lifestyle, had decreased in the catches of 2020. This also applies, for example, to a common species such as *Helophilus pendulus*. Some species appear virtually extinct, which is also reflected in hand catches in the entire De Brand area (pers. obs.). One of these is *Trichopsomyia flavitarsis*. It may well be that the decline had already started in or before 1990, but is obscured by the still partly high number of specimens at the time; older data are not available. Fact is that the restoration of ground- and surface water levels of recent years is not yet reflected in these figures. There are also some notable new appearances. Perhaps most striking is the appearance of *Neoascia interrupta*. On the other hand, *N. podagrica* and *N. meticulosa* have disappeared which is also evident from hand catches in a wider area in De Brand.

The soldier flies seem to have a hard time in De Brand. Numerical differences are very large, larger than one would expect, even if 2020 had simply been a very bad year. In both 1990 and 1991, there was a pasture grazed by cows near and in front of the trapping site, but it is impossible to estimate what the influence of this has been. It is possible that succession causes a large part of the typical herb-rich grassland species to disappear?

De Kaaistoep

De Kaaistoep underwent quite some management dynamics in the intervening years. It is difficult to say to what extent this influenced the results of 2020. Succession also plays a role in this area, especially in expansions of forest and bushes between the trap site and the opposite grasslands and fens. *Tropidia scita* doesn't seem to be affected by this; the species may benefit from the swampy places resulting from the newly formed meander. *Sphaerophoria batava* may suffer from the disappearance, since 1998, of the heath-like grassland near the Malaise trap. This may also apply to species from the genus *Chrysotoxum*.

As far as soldier flies are concerned, we see a similar picture in De Kaaistoep as in De Brand. The genera *Beris* and *Microchrysa* have completely disappeared from the catches, while *Pachygaster atra* has been caught in considerably higher numbers.

In Table 2 we can see the differences in the numbers and species of soldier flies and hoverflies for the De Brand area. Table 7 lists the numbers of hoverfly species caught in De Brand and De Kaaistoep, classified by larval lifestyle. In addition to the clear differences in numbers, the differences in the number of species found per larval lifestyle are also clearly visible. It is striking that the two areas both show the largest differences in the zoophagous species group. Moreover thirty years later, the number of species caught in De Brand is lower in all species groups. In De Kaaistoep however, the number of species within the non zoophagous species groups is approximately the same 22 years later.

The meteorological data are shown in this article to include one of the few well-documented variables. Since different species often have different individual requirements to thrive, it would go too far in the context of this paper to discuss individual cases. The meteorological data shown will enable the reader to relate these to the results concerning soldier flies and hoverflies in both areas. It is clear that climate is warming up and fluctuates in each of the seasons. But to what extent this contributes to changes in the insect populations is a complex question. Succession, management, natural fluctuations within a species, but also in the supply of their food – whether that of larvae or imagos, climate change, chemical changes in the environment; many factors influence the success of a species. In De Brand a short lasting Malaise trap project was performed in 2021, near the 1991 location, with a different type of Malaise trap that was differently positioned. In 2022 a Malaise trap project was performed for a longer period of time, with two traps placed near each other, which were of the same type as used in 1990 and 2020. However, these stood in another, comparatively moist, meadow in the same the area, between high-grown (oak) forest. But study of the data is still in progress. This and future research will have to show how insect populations will change in these reserves.

Table 7: Number of species in "De Brand" and "De Kaaistoep", classified by larval lifestyle. See table 4 for the legend.

	De Brand	**De Brand**	**De Kaaistoep**	**De Kaaistoep**
L	**1990**	**2020**	**1998**	**2020**
as	20	14	7	9
fy	6	4	5	8
ts	15	11	9	8
zoö	36	29	45	32
Total	**77**	**58**	**66**	**57**

Acknowledgment

We thank Jan Willem van Zuijlen for providing the necessary entomological data. Paul van Wielink and Milly Verpraet are responsible for the vegetation recordings. Theo Peeters has critically reviewed the text. Livia de Felici took care of the translation of the KNMI weather data into beautiful graphs. Arnold de Boer improved the translated text. All determinations and counts were carried out by members of the insect working group of the KNNV Tilburg branch, the Netherlands, for which many thanks!

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Fly Times 73 (2024)

Additions and comments to the monograph of Gisondi *et al*., 2020: The world Polleniidae (Diptera, Oestroidea): key to genera and checklist of species

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Abstract

Recently presented faunistics data omitted in the monograph (Gisondi et al., 2020) are given for 44 species of "polleniine flies" (Diptera: Calliphoridae) retrieved from 67 literary sources, including 51 that are not indicated in the monograph; type locality of *Xanthotryxus auratus* (Séguy, 1934) is incorrectly given as "China: Sichuan, Moupin", and that of *X. ludingensis* Fan, 1992 – as "China: Sichuan: Yanzigou, Mt. Gonggashan, 3600 m." There is no information in Gisondi et al. about the species *Pollenia tragica* Rayment, 1955 from Australia (Victoria); the authors indicate two species from different genera with the same source data, including the species epithet: *Dexopollenia sakulasi* Kurahashi, 1987 at p. 115 and *Pollenia sakulasi* (Kurahashi, 1987) at p. 139. The data on presence of *Pollenia paupera* Rondani, 1862 and *P. similis* (Jacentkovský, 1941) in Ukraine are erroneous; the political and administrative name "China (Xizang)" is recommended to be given as "Tibet".
Keywords: Polleniinae, world catalog, additions, corrections.

Introduction

Polleniinae (Diptera: Calyptratae: Calliphoridae) comprise 8 genera and 152 species worldwide (after Gisondi *et al*. 2020, with some subsequent corrections of species number). Polleniinae, especially, *Pollenia* spp., or cluster flies, are named for their habit of assembling (clustering) on tree trunks, walls and other vertical surfaces during overnight stay, and they enter buildings *en masse* in autumn to overwinter. The larvae of some *Pollenia* spp. are known as obligate endoparasitoids or predators of earthworms. Females oviposit on soil, especially earthworm casts, from where the newly hatched larvae make their way through the soil in search of hosts. Adults often visit flowers and, sometimes, excreta of homopterans or vertebrate carcasses (Draber-Mońko 2004; Falk 2016; Grunin 1970b; Jewiss-Gaines et al. 2012; Martínez-Sánchez *et al*. 1998; Mihályi 1980; Pratt *et al*. 1975; Rognes 1987, 1991a; Thomson and Davies 1973a, b; 1974; Verves 2005b; Yahnke and George 1972).

The complex revisions of high-ranking systematic groups (families, etc.) determine the direction of study of these taxa for many decades to come. Therefore, all data in such publications should be as complete as possible. Unfortunately, a comparison of the materials of the species catalog (Gisondi *et al*. 2020) with previously published data showed a number of drawbacks. There is an incomplete reflection of the data of the cited works and a lack of information from a number of important publications (Langhoffer 1920; Rayment 1955; Coe 1960; Gregor and Povolný 1960; Trofimov 1965; Kano and Shinonaga 1968; Park 1977; Mihályi 1979; Gosseries 1989; Schembri *et al*. 1991; Pakalniškis and Podénas 1992; Pape *et al*. 1995; Kurahashi *et al*.1997; Vaňhara and Rozkošný 1997; Greenberg 1998; Chandler *et al*. 2000; Gudjabidze, 2000; Grigorieva 2001; Truphanova and Khitzova 2001; Carles-Tolrá 2002; Rognes 2002; Raffone 2005; Verves 2005b; Xue 2005; Xue and Wang 2006; Chandler *et al*. 2008; Karpa 2008; Országh *et al*. 2009; Kurahashi 2010; Rognes 2010; Liu *et al*. 2012; Çoban and Beyarslan 2013; Draber-Mońko 2013; Chen and Zhu 2014; Parchami-Araghi *et al*. 2014; Rognes 2014; Feng 2015; Koçak and Kemal 2015; Kurahashi *et al*. 2016; Liu *et al*. 2018; El-Hawagry and El-Azab 2019; Verves and Khrokalo 2020). Below is given the necessary additions that are not in the catalog for the use of the faunal data of the peer-reviewed work.

Faunistic corrections

1. ***Dexopollenia bicolor*** Malloch, 1935. Oriental: Malaysia: Malaya (Kurahashi *et al*. 1997; Verves 2005a).
2. ***Dexopollenia bicoloripes*** Malloch, 1931. Oriental: Malaysia: Malaya (Kurahashi *et al*. 1997; Verves 2005a).
3. ***Dexopollenia chrysothrix*** Bezzi, 1927. Australasian: Papua New Guinea: New Guinea (Kurahashi 1987).
4. ***Dexopollenia flava*** (Aldrich, 1930). Oriental: China: Guangdong (Liu, Y. *et al*. 2018).
5. ***Dexopollenia geniculata*** Malloch, 1935. Palaearctic: China: Qinghai (Xue and Wang 2006); Tibet [as "Xizang"] (Xue and Wang 2006).
   Comments. "Tibet" is the original name of what is now a province-level autonomous region of the People's Republic of China.
6. ***Dexopollenia hirtiventris*** Malloch, 1935. Oriental: Malaysia: Malaya (Kurahashi *et al*. 1997; Verves 2005a).
7. ***Dexopollenia maculata*** (Villeneuve, 1933). Palaearctic: China: Qinghai (Xue and Wang 2006); Tibet [as "Xizang"] (Xue and Wang 2006).
8. ***Dexopollenia nigriscens*** Fan, 1992. Palaearctic: China: Qinghai (Xue and Wang 2006); Tibet (Verves 2005a) [as "Xizang"] (Fan 1992).
9. ***Dexopollenia uniseta*** Fan, 1992. Palaearctic: China: Qinghai (Xue and Wang 2006); Tibet [as "Xizang"] (Xue and Wang 2006)
10. ***Morinia argenticincta*** (Senior-White, 1923). Palaearctic: Japan: Hokkaido (Kurahashi 2010), Kyushu (Verves 2005a); North Korea (Draber-Mońko 2013).
11. ***Morinia doronici*** (Scopoli, 1763). Palaearctic: China: Sichuan (Feng 2015); Croatia (Langhoffer 1920); Slovenia (Scopoli 1763).
12. ***Pollenia alajensis*** Rohdendorf, 1926. Palaearctic: China: Xinjiang (Fan 1992); Kazakhstan (Grunin 1970a).
13. ***Pollenia amentaria*** (Scopoli, 1763). Palaearctic: Algeria (Rognes 2010); Azerbaijan (Trofimov 1965); Italy: mainland and Sicily (Pape *et al*. 1995); Tibet [as "Xizang"] (Pape *et al*. 1995).
14. ***Pollenia angustigena*** Wainwright, 1940. Palaearctic: Ireland (Chandler et al. 2008); Turkey (Çoban and Beyarslan 2013).
15. ***Pollenia atramentaria*** (Meigen, 1826). = *Nesodexia corsicana*: Raffone, 2005: 106; misidentification: not *Nesodexia corsicana* Villeneuve, 1911, after Rognes 2014: 114. Palaearctic: Georgia (Gudjabidze 2000; Mihályi 1979).
16. ***Pollenia bulgarica*** Jacentkovský, 1939. = *Pollenia bisulca*: Jacentkovský, 1936: 114; misidentification: not *Pollenia bisulca* Pandellé, 1896, after Rognes, 1991: 464. Palaearctic: North Macedonia (Rognes, 1991); the occurence of this species in Slovakia (Gregor 1987, etc.) is erroneous (Országh et al. 2009).
17. ***Pollenia contempta*** Robineau-Desvoidy, 1863. Palaearctic: Algeria (Schumann 1986); Morocco (Schumann 1986).
18. ***Pollenia dasypoda*** Portschinsky, 1881. Palaearctic: Afghanistan (Koçak and Kemal 2015); Algeria (Koçak and Kemal 2015); Armenia (Rognes 2019); Libya (Koçak and Kemal 2015); Morocco (Koçak and Kemal 2015); Tunisia (Koçak and Kemal 2015); Uzbekistan (Koçak and Kemal 2015).
19. ***Pollenia fulvipalpis*** Macquart, 1835. Palaearctic: Czech Republic (Vaňhara and Rozkošný 1997).

20. ***Pollenia griseotomentosa*** (Jacentkovský, 1944). Nearctic: Canada: Newfoundland (Jewiss-Gaines *et al*. 2012); USA: Pennsylvania (Jewiss-Gaines *et al*. 2012). Palaearctic: Ireland (Chandler *et al*. 2000); Lithuania (Pakalniškis and Podénas 1992); Norway (Rognes 1985).
21. ***Pollenia hazarae*** (Senior-White, 1923) Oriental : Pakistan. Erroneously mentioned: India in Gisondi et al. 2020.

22. ***Pollenia huangshanensis*** Fan and Chen, 1997 in Fan, 1997. Palaearctic: China: Beijing (Fan 1997), Shaanxi (Fan 1997). Oriental: China: Fujian (Fan 1997; Verves 2005a), Zhejiang (Fan 1997; Verves 2005a).
23. ***Pollenia ibalia*** Séguy, 1930.
    Pollenia corinnae Gosseries, 1989: 3. New replacement name for *Pollenia funebris* Villeneuve, 1932.
24. ***Pollenia japonica*** Kano and Shinonaga, 1966. Palaearctic: Japan: Tsushima Islands (Kurahashi *et al*. 2016); South Korea (Park 1977).
25. ***Pollenia labialis*** Robineau-Desvoidy, 1863. Palaearctic: Albania (Gregor and Povolný 1960); Georgia (Gudjabidze 2000); Portugal: Azores (Carles-Tolrá 2002); Spain: Baleares (Carles-Tolrá 2002).
26. ***Pollenia mediterranea*** Grunin, 1966.
    Nitellia hermoniella Lehrer, 2007: 24.
    Palaearctic: Azerbaijan (Grunin 1966); Georgia (Gudjabidze 2000), Lebanon (Rognes 1991b); Syria (Rognes 1991b).
    Comments: Gisondi *et al*. (2020: 133) proposed *Nitellia hermoniella* as a "syn. nov." of *P. mediterranea* "for the first time" (published 29 September 2020). Actually, *N. hermoniella* was first synonymized with *P. mediterranea* three months earlier on 30 June 2020 (Verves and Khrokalo 2020: 40).
27. ***Pollenia paupera*** Rondani, 1862. Palaearctic: West Bank (Rognes 2002). The data on presence of this species in Ukraine (Verves 2005b) are erroneous.

28. ***Pollenia pectinata*** Grunin, 1966. Palaearctic: China: Beijing (Fan 1992); Henan (Liu J. *et al*. 2012); Neimenggu (Fan 1992); Shanxi (Fan 1992).
29. ***Pollenia pediculata*** Macquart, 1834. Palaearctic: China: Qinghai (Xue and Wang 2006); Iran (Parchami-Araghi et al. 2014); Ireland (Chandler *et al*. 2008); Israel (Parchami-Araghi et al. 2014); Tibet [as "Xizang"] (Xue and Wang 2006); West Bank (Rognes 2002).
30. ***Pollenia pseudintermedia*** Rognes, 1987. Palaearctic: Syria (Rognes 2002).
31. ***Pollenia rudis*** (Fabricius, 1794). Palaearctic: China: Qinghai (Xue and Wang 2006); Egypt (El-Hawagry and El-Azab 2019); Latvia (Karpa 2008); Malta (Schembri *et al*. 1991); Tibet [as "Xizang"] (Xue and Wang, 2006); United Kingdom: Northern Ireland (Parchami-Araghi *et al*. 2014); West Bank (Rognes 2002). Australasian: USA: Hawaii (Kurahashi 1989; Verves 2005a).
32. ***Pollenia semicinerea*** Villeneuve, 1911. Palaearctic: Russia: Krasnodar Krai, Stavropol Krai (Grunin 1966)
33. ***Pollenia shaanxiensis*** Fan and Wu, 1997. Palaearctic: China: Gansu (Xue 2005).
34. ***Pollenia sichuanensis*** Feng, 2004. Type locality: China: Sichuan: Mao County.
    Comments. The name of the type locality is incomplete; it is necessary to include additional data from Feng (2004: 808): "Mt Maoxian, 32˚15'N, 104˚08' E".
35. ***Pollenia similis*** (Jacentkovský, 1941). Palaearctic: Bulgaria (Schumann 1986).
    Comments. The data on presence of this species in Ukraine (Verves 2005b) is erroneous.
36. ***Pollenia townsendi*** Senior-White, Aubertin and Smart, 1940. Type locality: India: Himachal Pradesh.
    Comments. The name of type locality is incomplete; it is necessary to include additional data from Senior-White et al. (1940: 119): "Simla".

37. ***Pollenia vagabunda*** (Meigen, 1826). Nearctic: Canada: New Brunswick (Jewiss-Gaines *et al*. 2012); USA: Indiana (Greenberg 1998), Rhode Island (Jewiss-Gaines *et al*. 2012), Washington (Whitworth 2006). Palaearctic: North Macedonia (Coe 1960).
38. ***Pollenia viatica*** Robineau-Desvoidy, 1830. Palaearctic: Albania (Mihályi, 1980); Ireland (Chandler *et al*., 2000); Russia: Karachai–Cherkessia (Grunin, 1970a), Pskov (Grigorieva, 2001), Voroniezh (Truphanova and Khitzova 2001).
39. ***Xanthotryxus auratus*** (Séguy, 1934). Palaearctic: China: Qinghai (Xue and Wang 2006), Sichuan (Séguy 1934; Verves 2005a); Tibet [as "Xizang"] (Xue and Wang 2006).
    Comments. Type locality of *Pollenia auratus* after Séguy (1934: 22) has been given as "Thibet, Mou-Pin", or in interpretation of Gisondi *et al*. (2020: 143), "China (Xizang)". Moupin is placed in Sichuan Province of China; thus, the correct type locality is "China: Sichuan, Moupin".
40. ***Xanthotryxus draco*** Aldrich, 1930. Palaearctic: China: Qinghai (Xue and Wang, 2006); Tibet [as "Xizang"] (Xue and Wang 2006).
41. ***Xanthotryxus ludingensis*** Fan, 1992. Palaearctic: China: Qinghai (Xue 2006); Tibet [as "Xizang"] (Xue and Wang 2006).
    Comments. Gisondi et al. (2020: 143) errroneously gave the type locality of this species as "China, Sichuan, Luding". The type locality according to Fan (1992) is: "China: Sichuan: Yanzigou, Mt. Gonggashan, 3600 m." Luding, or Luding county, is an administrative part of Sichuan Province, and Yanzigou is one of the towns from this district.
42. ***Xanthotryxus melanurus*** Fan, 1992. Palaearctic: China: Beijing (Chen and Zhu 2014), Qinghai (Xue and Wang 2006); Tibet [as "Xizang"] (Xue and Wang 2006).
43. ***Xanthotryxus mongol*** Aldrich, 1930. Palaearctic: China: Qinghai (Xue and Wang 2006); Japan: Honshu (Kano and Shinonaga 1968; Verves 2005a) [not Kyushu, as erroneously published by Gisondi et al. (2020: 144)]; Tibet [as "Xizang"] (Xue and Wang 2006).
44. ***Xanthotryxus uniapicalis*** Fan, 1992. Palaearctic: China: Qinghai (Xue and Wang 2006); Tibet [as "Xizang"] (Xue and Wang 2006).

A nominal species not given by Gisondi et al. (2020):

Pollenia tragica Rayment, 1955: 134–136, figs 1–13, ♂. Holotype: ♂: Australia: Victoria: Portland, Cape Nelson Road. Holotype given as deposited in the private collection of Dr. T. Rayment and possibly in National Museum of Victoria.
Australasian: Australia: Victoria.

Two different names for a single species:

Dexopollenia sakulasi Kurahashi, 1987
Dexopollenia sakulasi Kurahashi, 1987: 68. Australasian: Papua New Guinea.
Dexopollenia sakulasi Kurahashi, 1987: Gisondi et al., 2020: 115.
Pollenia sakulasi (Kurahashi, 1987): Gisondi et al., 2020: 139.
Comments. The authors listed two nominal species from different genera with the same source data, including the species epithet. I consider the valid name to be the one given by the author of the species: "*Dexopollenia sakulasi* Kurahashi, 1987".

Acknowledgements

The authors acknowledge funding received via grant no. KPKVK 6541230 and SR/WOS-A/LS-187/2019(G).

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Genitalia vial alternative

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Diptera genitalia can be mounted on a slide, often in Canada Balsam or Euparal, or stored in glycerin in a genitalia vial. Slide mounting has the disadvantages that the chosen orientation for the genitalia is permanent, and the slide is usually stored other than with the actual fly. Genital vial storage has the advantage that the genitalia remain attached to the pinned fly and can be removed from the vial and examined at all angles. Cumming (1992) recommended examining genitalia in a depression slide containing glycerin and then storing the genitalia in glycerin [presumably in a genitalia vial]. One disadvantage of genital vials is the narrowness of the vial (2.5 or 4 mm) and when combined with the depth (10 mm) makes retrieval of the genitalia difficult. I prefer to store genitalia in the caps of polyethylene tubes. Such tubes come in various sizes: the cap of the 1.5 mL microcentrifuge tube with a 8 mm inner diameter and a depth of 4mm is the largest for practical use; the cap of 1 mm storage tube with a 4.8 mm inner diameter and a depth of 3.4 mm is suitable for small genitalia. Polyethylene genitalia vials are available with inner diameters of 2.5 mm or 4mm. Figure 1 shows the inner diameters of a 4 mm genital vial (1), a 1 mL storage tube (2), and a 1.5 mL microcentrifuge tube (3).

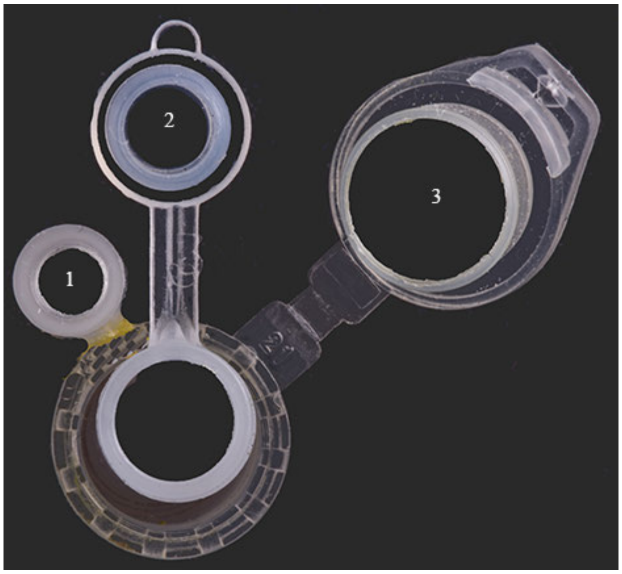

Figure 1. Inner diameters (ID) of caps of a: 1, genitalia vial (4 mm ID); 2, 1 mL storage tube (4.8 mm ID; 3, 1.5 mL microcentrifuge tube (8 mm ID).

In use, the caps are cut from the tubes so as to leave a convenient pinning tag. Figure 2 shows the genitalia of an asilid in the cap of a 1.5 mL tube filled with glycerine and the cap attached to the pin of the mounted fly.

Figure 2. Pinned male asilid with its genitalia in glycerine in the cap of a 1.5 mL centrifuge tube.

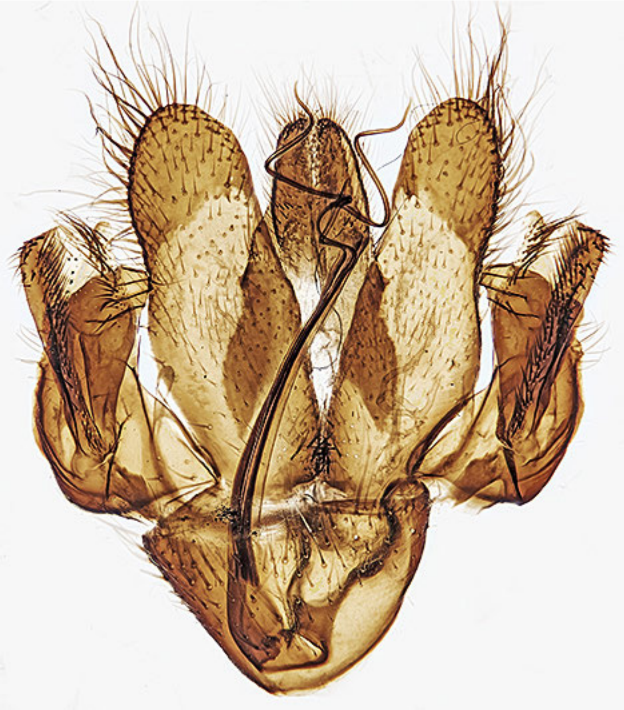

Figure 3. Ventral view of spread male asilid genitalia.

The genitalia can be photographed in the cap as in Fig. 2 (top) where the genitalia are seen in ventral view or removed from the cap and placed on a cavity slide for photographing. If needed it can be dehydrated, spread, cleared in cedarwood oil and photographed (Fig. 3). After photographing the genitalia can be cleaned of oil with alcohol and then returned to glycerine (Fig. 2, bottom).

Reference

Cummings, J. 1992. Lactic acid as an agent for macerating Diptera specimens. Fly Times 8: 7.

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Mites (Trombidioidea) taken from Diptera (Ceratopogonidae, Drosophilidae)

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In the course of routine surveillance for mosquitoes, a biting midge (Ceratopogonidae) was found with mites attached. The midge was collected in a dry ice-baited American Biophysics Company light trap.

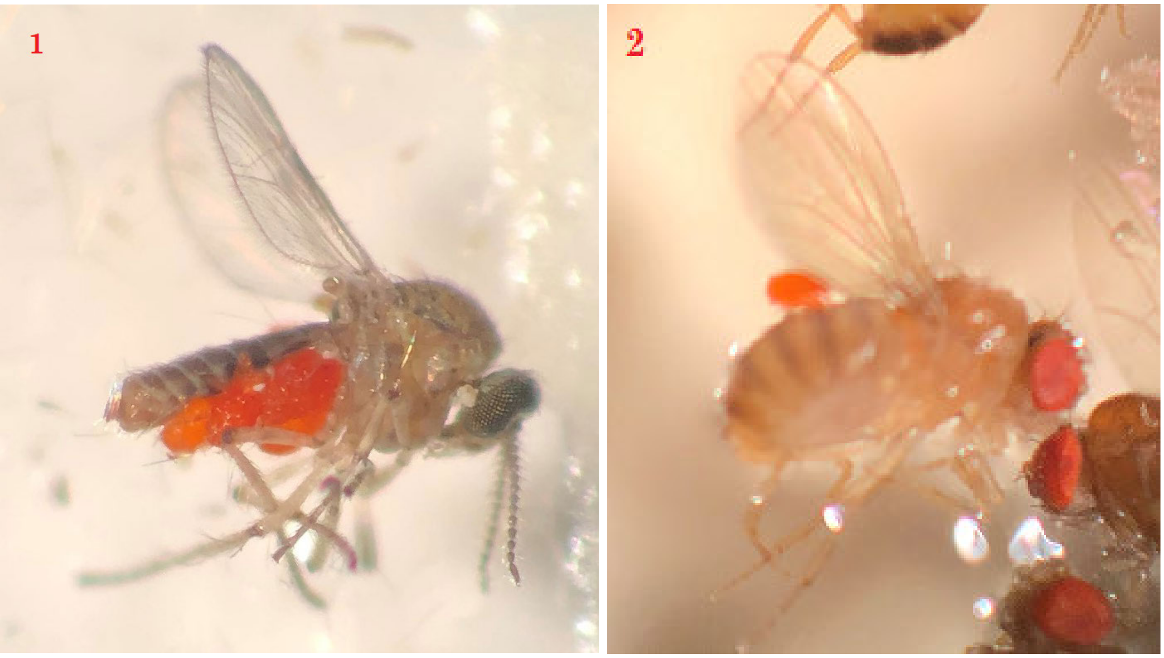
Collection data: FL, Monroe Co., City of Marathon, Flamingo Island, 19 Mar 2024, A. Loftus.

The midge is a female Forcipomyiinae, not identifiable further. Five mites were attached; they appear to be Trombidiidae or a related family (Fig. 1). Larval Trombidoidea are known to attach to ceratopogonids (Baker 1999) and drosophilids (Haitlinger and Šundić 2018).

During screening of baits for Drosophilidae, a fly with an attached mite was collected (Fig. 2).

Collection data: FL, Monroe Co. City of Marathon, Vaca Key, 19 Jul 2024, L. Hribar.

The fly is a female *Drosophila*. One mite was attached; it appears to be in the family Microtrombidiidae based on morphology of the mouthparts (Fain and Grootaert 1994).



Figures 1–2. Mites attached to flies. **1.** Female Forcipomyiinae. **2.** Female *Drosophila*.

Trombidioid mites have been collected from Ceratopogonidae and Drosophilidae (Baker 1999, Haitlinger and Šundić 2018). Not much known about the impact of mites on invertebrate hosts (Paré and Dowling 2012). A great many mites simply use their arthropod hosts for phoretic transfer (Szymkowiak et al. 2007).

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Two species of Tabanidae new to Florida: *Chrysops variegatus* (De Geer, 1776), *Tabanus nefarius* Hine, 1907

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Chrysops variegatus

Images of a specimen from Brevard Co., FL, March 15 2022, was posted on BugGuide. https://bugguide.net/node/view/2098825 (pictured right).


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Unfortunately this species was omitted from Fly Times Supplement 6 (February 2024) “The horse flies, deer flies, and yellow flies of Florida (Diptera: Tabanidae.” https://dipterists.org/assets/PDF/flytimes_supplement06.pdf

A second specimen from Desoto Co., FL, November 9 2024 was posted on BugGuide. https://bugguide.net/node/view/2417011

Fairchild (1986) gave the range as "from southern Mexico and Cuba south through the Antilles and Central America to northern Argentina and Paraguay." It seems possible that this South American species may now be established in Florida.

Its placement in the *Chrysops* key couplets in Fly Times Supplement 6; page 61:

18(16) Abdomen with a wide continuous middorsal yellow stripe, at least on the first three tergites.......... 19
– Abdomen without a continuous middorsal yellow stripe.......... 27
19(18) Wing first basal cell (1stbc) hyaline.......... 19a
– Wing first basal cell mostly infuscated.......... 20
19a(19) Wing apical spot never reaches beyond second submarginal cell. Wing discal cell lacking a clear central spot.......... *univittatus*
– Apical spot reaches hind margin of wing. Discal cell with a very large clear central area.......... ***variegatus***

Tabanus nefarious

This species was included (in the key and with images) in Fly Times Supplement 6 as possibly occurring in Florida. Its presence was confirmed from a specimen from Hendry Co., FL (June 1 2024); images on BugGuide:
https://bugguide.net/node/view/2418635

Acknowledgment

I thank Chris Rorabaugh for permission to include his image of the FL *C. variegatus.*

Reference

Fairchild, G.B. 1986. The Tabanidae of Panama. Florida Agricultural Experiment Station, Journal Series no. 6654. Contributions of the American Entomological Institute 22(3):139 pp.

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Systema Dipterorum
The BioSystematic Database of World Diptera

Systema Dipterorum Version 5.6 update
(posted online 9 December 2024)

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Flies just keep on being described!! We have now crossed the 170,000 living species-mark as we continue to enter data from taxonomic papers. We continue to encourage all authors to send pdfs (or links to them) to us so that the data can be assured of information being entered accurately. Apologies for any errors users may find. We strive to get as much accurate information into the database as possible, but there will inevitably be new synonymies, new combinations, classification changes, etc. that we have missed. There are a number of papers we can easily access from mainstream journals like Zootaxa and ZooKeys, and open access papers, but smaller journals, and those behind paywalls, are more tricky to access – and for others we may not even know about them. Again, we could not have gotten to where we are without the help of our sharp-eyed users, who have assisted by pointing out errors needing correction, and those who have helped by providing requested pdfs and information on dating, nomenclature, taxonomy, etc., etc.

Go here for more information and for searching the database: http://diptera.org/nomenclator

Shout-outs to those who have helped since the last notice in *Fly Times* in June 2024 (not in any particular order): Doug Yanega, Daniel Whitmore, Zachary Dankowicz, Verner Michelsen, Art Borkent, Ximo Mengual, Adrian Pont, Jan Ševčik, Dalton de Souza Amorim, Chris Angell, Steve Gaimari, Andrew Whittington, Sang Jae Suh, Elisabeth Stur, Stephen Smith, Arthur Frost, Carlos Lamas, Ralph Harbach, Gary Steck, Andrew Ross, Richard Pyle, Niladri Hazra, Greg Daniels, Brad Sinclair, John Ponting, Jere Kahanpää, Jim O’Hara, Pjotr Oosterbroek, Francesco Vitali, Rita Oliveira dos Santos, Debdeep Pramanik, Iain MacGowan, Gil Miranda, Stefan Ungricht, Yury Roskov, Geoff Ower, Dmitri Dimitrov, Michael von Tschirnhaus, Mihaly Földvári, Mousumi Chowdhury, Jens-Hermann Stuke, Sander Bot, Carlo Monari, Dan Bickel, Manuel Ayala, Stuart Longhorn, Dimitar Bechev, Ashley Kirk-Spriggs, Carolina Bellodi, Mitsuhiro Iwasa, Thalles Pereira.

Current numbers (as of 15 December 2024):
Extant valid species-group names: 170,100
Valid genus-group names: 12,727
References Databased: 41,218

Reminder:
Four years ago we began the new series “Systema Dipterorum Nomenclatural Notes” (SDNN) that provides a medium for short notes relating to nomenclatural “cleaning-up”. There are many instances when a novel nomenclatural act is needed for proper treatment of names (e.g., multiple original spellings requiring First Reviser action, preoccupied names, genus-group names lacking a type

species designation, genus-group names lacking included species, etc.). Also, when electronic-only papers do not fulfill the ICZN *Code* requirements (mostly because of lack of included ZooBank registration number), SDNN offers a quick remedy to make available those names (we have published a handful of such papers over the last 4 years). Note here that *Systema Dipterorum* only includes information that has been formally published, and SDNN offers an efficient means of documenting nomenclatural effects of phylogenetic advances, for example new combinations from synonymized nominal genera. Any worker wishing to submit articles should contact the editors for further information. All submitted manuscripts undergo peer-review. All publications in this series and new nominal taxa proposed therein are registered with ZooBank and are Open Access.

Go here for more information and current articles:
http://hbs.bishopmuseum.org/sd/

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HISTORICAL DIPTEROLOGY

Obituary of Antonio Marcelino do Carmo-Neto (1992–2024)

Maria Virginia Urso Guimarães

Federal University of São Carlos, Department of Biology, Diptera Systematics Laboratory,
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It is with great sadness that we learn of the death of Antonio Marcelino do Carmo-Neto, who passed away July 16, 2024, in Brazil after a prolonged bout of pneumonia. Neto, as he was known to his friends, was born on October 26, 1992, in Sorocaba, São Paulo State, Brazil, the son of Neudemar and Márcia do Carmo, and brother of William. He was the inseparable companion of Monique Romeiro, his dearest partner, and friend of many good people. He was educated in a public school in Sorocaba and graduated from the high school from SENAI – São Paulo. In 2011, he was admitted to study Biological Sciences at the University of São Carlos – Sorocaba Campus. During this time, he developed an active interest in the biodiversity of mycophagous cecidomyiids, which culminated in the development of his final monograph on the taxonomy of the Lestremiinae, Micromyinae and Stomatosematidi (Diptera, Cecidomyiidae) of the state of Mato Grosso do Sul, Brazil. After that, in 2014, he obtained his bachelor's degree.

Figure 1. Antonio at the Ecological and Experimental Station of Assis, SP, Brazil.

In 2015, he was admitted to the Museum of Zoology of the University of São Paulo, in the city of São Paulo at the postgraduate level to develop his knowledge in Neotropical Stomatosematidi under the supervision of Dr. Carlos Lamas and me. He obtained the MSc degree in Systematics, Animal Taxonomy and Biodiversity for his brilliant dissertation "Systematics of the species of the supertribe Stomatosematidi (Diptera: Cecidomyiidae), with emphasis on the positioning of species in the Neotropical Region", defended in 2017. In 2018, he continued his studies of mycophagous cecidomyiids in the same postgraduate program and supervisors at the doctoral level, including those of the subfamily Lestremiinae, never before studied in the Neotropical region. He traveled abroad for a six-month (September/2019 – February/2020) sandwich Ph.D. at the Museum für Naturkunde Berlin, where he was supervised by Dr. Eliana Buenaventura and visited the Swedish Museum of Natural History, in Sweden and the Senckenberg Deutsches Entomologisches Institut, in Germany. Back in Brazil, he faced the COVID pandemic period, stoically keeping his research activities active in the Diptera Systematics Laboratory.

Figure 2. Antonio with the Class of 2010 graduated in Biological Sciences.

Figure 3. Antonio with his advisors and fellow post-graduates at USP's Zoology Museum. From left to right: Carlos Lamas, Virginia Urso, Antonio, Carolina Garcia, Michelle Vicente.

At the end of 2022, he received his degree, for a thesis entitled "Cladistic Analysis of Lestremiinae (Diptera: Cecidomyiidae)". Currently, he was associated as a postdoctoral researcher in the Laboratory of Diptera Systematics of the Biology Department/ UFSCar, under my responsibility. He had been working on several manuscripts on the taxonomy, systematics, expansion of the geographical distribution and understanding of the evolution of the ultrastructural characters of the antennal and palpus sensilla of Lestremiinae, Mycromyinae and Stomatosematidi derived from his dissertation and thesis; he supervised undergraduate and graduate students in Biological Sciences; he was essential in the arthropod collecting expeditions in the projects related to the thematic project "Natural grasslands in the state of São Paulo: Diagnosis, management and conservation" and in the “Inventory of galls and gall makers of Boraceia Ecological Station/USP-SP”.

Figure 4. Antonio among colleagues from the Diptera Systematics Laboratory at UFSCar, Sorocaba, SP, Brazil, and his life partner Monique Romeiro (on his right).

He has always shown great enthusiasm and systematic curiosity for the biology of mycophagous cecidomyiids, for which he has studied methods of collection and rearing from material collected during the aforementioned expeditions. Last year he joined the UFSCar Choir as a tenor. A faithful friend, a companion at all times, an enthusiastic and dedicated researcher, we leave our tribute here with great sadness at this loss, knowing that what was to come in his career promised many new developments in Neotropical mycophagous cecidomyiids, which we hope to be able to publish in his memory.

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PHILAMYIANY

Diptera on stamps (8): Syrphidae excluding Syrphinae

Jens-Hermann Stuke

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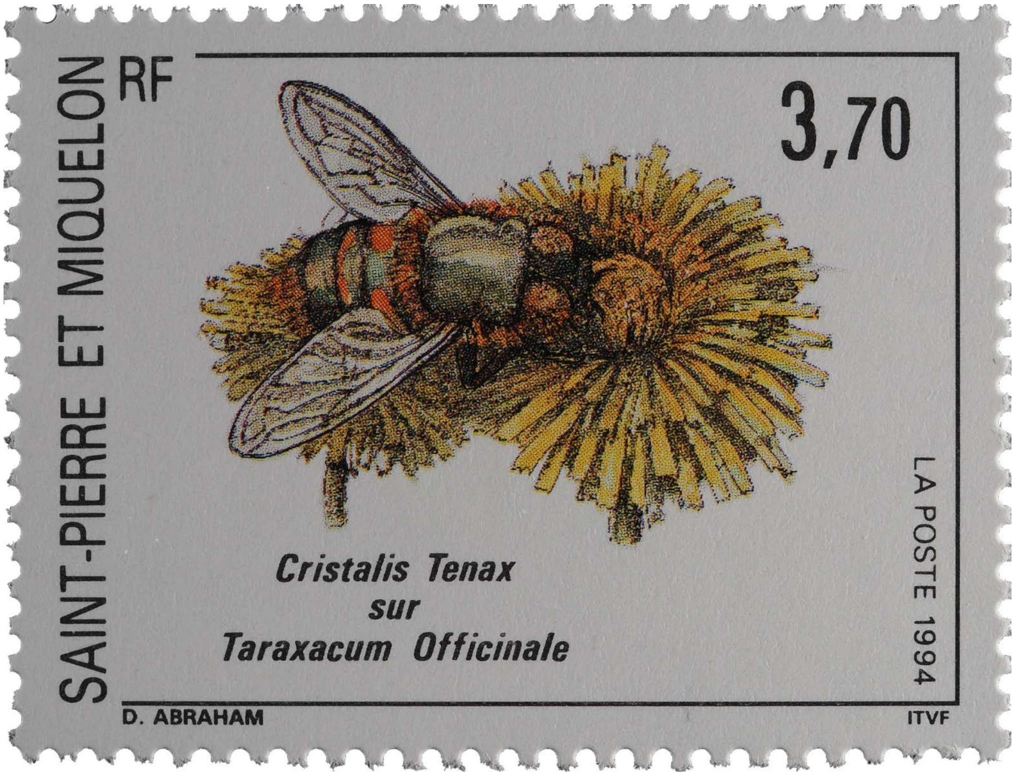
An exciting first stamp with a Syrphidae originates from Buthan 1969. It is not only the first stamp showing a Diptera without medical significance but until now it is the only 3D stamp with a Diptera. And it is remarkable that no obvious pretty or spectacular fly species is shown but only a less obvious Eristalini. Most Syrphidae shown here are striking insects and belong to the large and obvious flies like *Senaspis*, *Milesia*, *Volucella*. One additional reason for the occurrence of Syrphidae on stamps might be the fact that some species are very common and as migrating flies occur almost everywhere as for example *Eristalis tenax* on small islands like Saint-Pierre or the Marshall Islands. For each stamp I have provided the country and year of issue, title of stamp, title of stamp series (where available/relevant), face value, Michel number and stamp number (the latter both copied from https://colnect.com).

Acknowledgement
Martin Hauser (California) helped with the identification of the Syrphidae.

Eristalini spec. – Bhutan 1969: [illegible signs], 2 Ngultrum butanés. – Michel number: BT 272; stamp number: BT 101C.

***Eristalis tenax* (Linnaeus, 1758) – Germany [Berlin] 1984**: Schwebfliege [Für die Jugend], 120 + 60 Pfennig. – Michel number: DE-BE 715; stamp number: DE 9NB212.



***Eristalis tenax* (Linnaeus, 1758) – France [Saint-Pierre and Miquelon] 1994:** *Eristalis Tenax* sur *Taraxacum Officinale*, 3.70 French France. – Michel number: PM 672; stamp number: PM 599.

***Volucella zonaria* (Poda, 1761) – Britain [Jersey] 2008:** Large Migrant Hoverfly, *Volucella zonaria* [Insects, 2nd series], 52 Pennique de Jersey. – Michel number: JE 1374; stamp number: –.

Milesiini (cf *Milesia*) – Marshall Islands 2002: Flower Fly, 23 United States cent. – Michel number: MH 1650; stamp number: MH 808f.

Eristalis transversa **(Wiedemann, 1830) – Marshall Islands 2014:** Garden Insects, 49 United States cent. – Michel number: MH 3294; stamp number: MH 1078a.

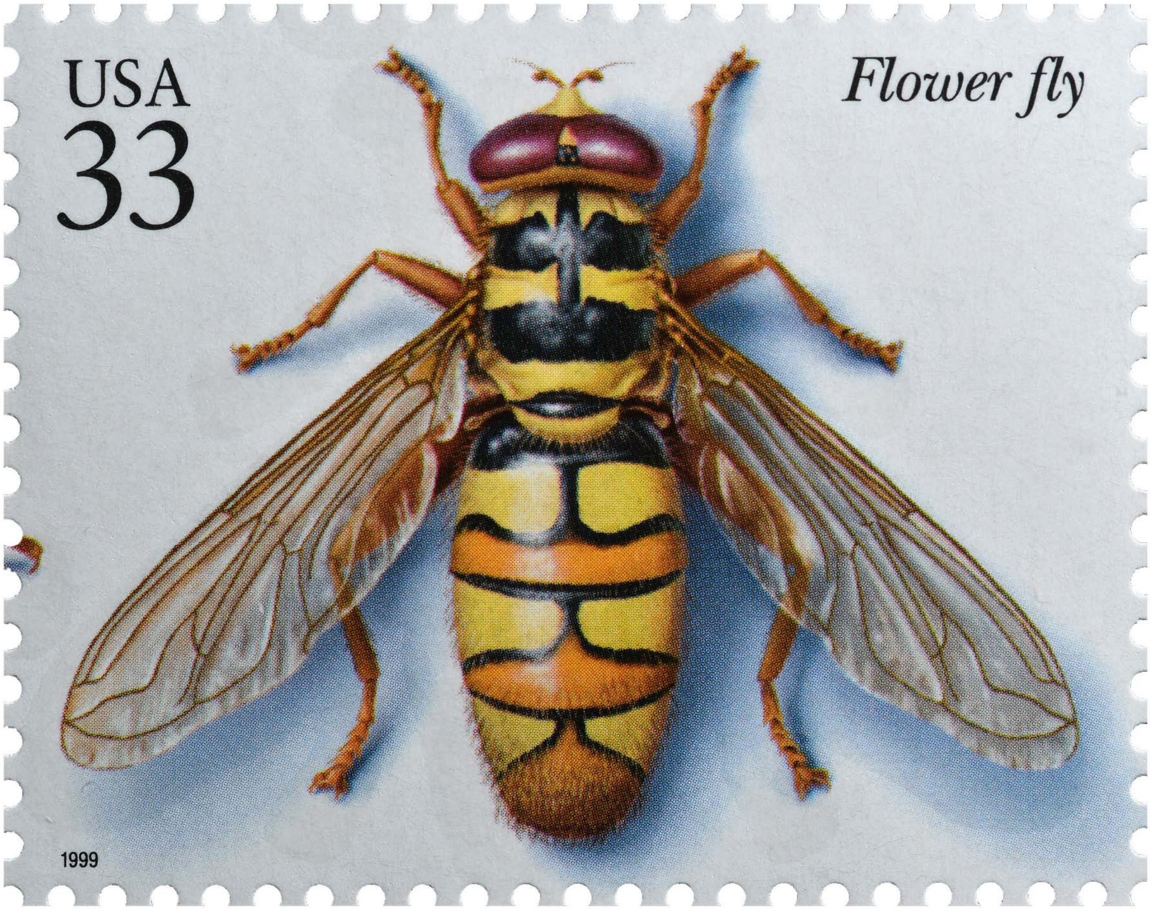
***Eristalis transversa* (Wiedemann, 1830) – Marshall Islands 2014:** Lacewing, 49 United States cent. – Michel number: MH 3296; stamp number: MH 1078c.

***Eristalis tenax* (Linnaeus, 1758) – Marshall Islands 2014:** Drone Fly, 49 United States cent. – Michel number: MH 3302; stamp number: MH 1078g.

***Eristalis tenax* (Linnaeus, 1758) – Marshall Islands 2014:** Garden Insects, 49 United States cent. – Michel number: MH 3303; stamp number: MH 1078j.

Eristalini spec. – Tajikistan 2016: – [Turtles], 3.50 Tajikistani somoni. – Michel number: TJ 747A; stamp number: TJ 466b.

***Senaspis aesacus* (Walker, 1849) – Rwanda 1973**: *Senaspis oesacus* [Insects], 18 Rwanda franc. – Michel number: RW 544A; stamp number: RW 501.



Milesia virginiensis **(Drury, 1773) – USA 1999:** Flower fly [Insects & Spiders], 33 United States Cent. – Michel number: US 3197; stamp number: US 3351f.

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Team Diptera presents: Insect Coven

Constance Taylor[1], Charlotte Alberts[2a], Ezra Bailey[3], Allan Cabrero[2b], Brittany Kohler[4]

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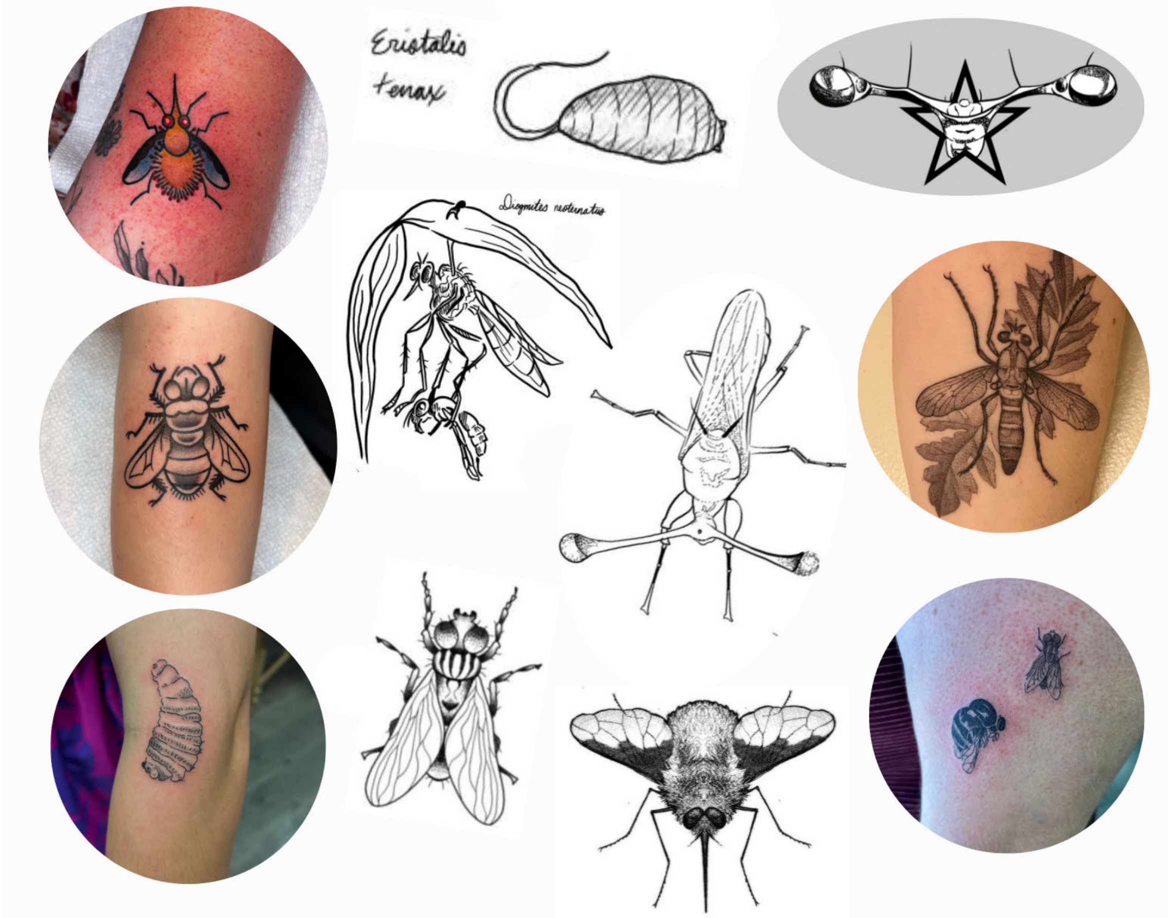
For the 2024 Entomological Society of America conference in Phoenix, Arizona, USA, Team Diptera partnered with Coven Tattoo to offer entomological flash (pre-made tattoo designs) for attendees. We sent Coven a list of suggestions, which was obviously weighted heavily in favor of flies. We considered sending them *only* flies, then figured it was probably best to play nice with other taxa. There were a number of conference-goers who ended up with various arthropods punched into their skin, so we created this fly-edit photo montage of some tattoos and flash. To take a look at all of the flash, visit https://tinyurl.com/insect-coven. If you want to peep the non-fly tattoos on ESA attendees, visit @coven.phx on Instagram.



Artists: @shay_k_art; @adeleines_garden; @hobgoblin.ink; @jamiedovetattoos; @_dcb_designs_; @godpity.ink

Enjoy! https://linktr.ee/team_diptera

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DipterART: Some more flies from Paris

Stephen D. Gaimari & Martin Hauser

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Of course we know flies are ubiquitous, but that does not mean that they show up very often in artwork. In *Fly Times* issue 69, the first author presented five paintings held in the Louvre. Here we add two more, with photos taken by the first author. A yellow arrow points to the fly on the full painting. In addition, there are photos of a beautiful vase held at the Musée d'Orsay.

"Mets, fruits et verres sur une table"
(Dish, fruits and glasses on a table)

Oil on panel, before 1632.

By Peter Binoit (ca. 1590–1632), a German still life painter during the early Baroque period.

Certainly a Calyptratae, likely Muscidae.

(https://collections.louvre.fr/en/ark:/53355/cl010062473)

"Fleurs, fruits, oiseaux et insectes dans un paysage avec ruines, avec une souris pénétrant dans un nid" (Flowers, fruit, birds and insects against a background of ruins, with a mouse entering a nest)

Oil on canvas, after 1660.

By Abraham Mignon (1640–1679), a German-born Dutch painter specializing in still ife paintings of flowers. Interestingly, Mignon trained the young pupil Maria Sibylla Merian, who became famous for her entomological work.

A Tipulidae, possible *Nephrotoma*? I'm sure our tipulid specialists will tell me otherwise if wrong!

(https://collections.louvre.fr/en/ark:/53355/cl010060918)

Unnamed “Vase forme Étrusque de Naples”
(Etruscan-shaped vase from Naples)

Hard porcelain, chiseled and gilded bronzes, 1854.

By Maximilien Mérigot (1822–1892), French painter on porcelain.

With a fair amount of artistic license by the painter, this is a Tabanidae, with some of the patterns looking typical of *Chrysops* as being the possible model.

Presented at the Exposition Universelle de Paris in 1855, then housed in the Palais de Saint-Cloud.

Held at Musée d’Orsay, not currently on display. (https://www.musee-orsay.fr/en/artworks/vase-forme-etrusque-de-naples-153789)

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Meeting News

Dipterists Society Field Meeting, The Evergreen State College, Olympia, Washington, July 15–19, 2024

Barbara Hayford[1] & Andrew Fasbender[2]

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Overview

The eighteenth Field Meeting of the Dipterists Society was held July 15–19, 2024, at The Evergreen State College in Olympia, Washington. The host venue included lodging, a meeting and presentation venue, and meals. The College is located within 400 hectares of coastal forest in the Puget Sound Lowlands. Participants had permission to sample along streams, marine shoreline, and forest during the meeting (Fig. 1). Field trips were organized to sample the Chehalis River surge plain, Mima Mounds prairies/oak savannah ecosystems at two different locations, rainforests, and montane and mountain stream habitat in the Olympic Mountains.

Figure 1. Meeting venue, forest, shoreline, and stream habitat at The Evergreen State College location of the 2024 Field Meeting of the Dipterists Society. Photo credit Barbara Hayford.

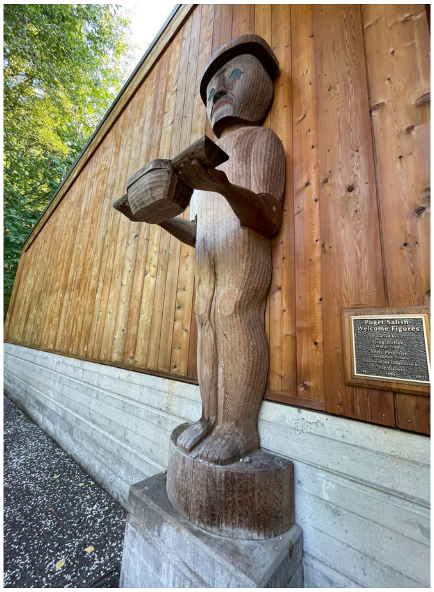
The meeting had 25 attendees (Fig. 2) including one undergraduate, four graduate students, six government researchers, nine researchers from academia, one museum collection manager, three other researchers, and an artist. International attendees traveled from as far away as Brazil and from nearby British Columbia, Canada whereas attendees from the US traveled from as far away as Gainesville, Florida and as near as Ocean Shores, Washington. The attendees shared their interests in taxonomy and evolution, ecology and natural history, insect control and conservation, mosquito rearing and other laboratory techniques, and insect photography. One researcher shared Diptera related research on his land in the Mackenzie River watershed, Oregon.

Figure 2. Photo of participants of the 2024 Field Meeting of the Dipterists Society held at The Evergreen State College, Olympia, Washington. Front row from left: Nina Miller, Megan Asche, Michael Palmer, Zachary Glass, Solange Akimana, Bridget White, Stephen Luk, Bill Murphy, Severyn Korneyev, Kristina Kernytska, Will Bouchard, Zell Smith. Back row from left: Susan Cumming, Sarah Oliveira, Tristan McKnight, Karl McKnight, Luc Leblanc, Brittany Wingert, Jim Hogue, John Stireman, Greg Dahlem, Jeff Cumming, Andrew Fasbender. Not pictured, Zachary Dankowicz and Barbara Hayford. Photo taken in front of the Welcome House. Photo credit Zachary Dankowicz.

Field collecting techniques included sweep and aerial netting, beating sheet, light trapping, collection of surface-floating pupal exuviae, photography, pan trapping, and others. (Fig. 3). The general schedule for the meeting included breakfast at The Evergreen State College followed by assembly and travel to field sites, field work (Fig. 4), return to The Evergreen State College for dinner and evening presentations at the s'gʷi gʷi ʔ altxʷ "House of Welcome" Longhouse Education and Cultural Center (Figs 2, 5, 6). In addition to the opening presentation, 15 presentations were given over three evenings (Figs 7, 8). Permission to sample field sites was provided by The Evergreen State College, Washington Department of Natural Resources (WDNR), Washington Department of Fish and Wildlife (WDFW), and the Olympic National Forest (ONF).

Figure 3. Field collecting techniques used during the 2024 Field Meeting. Clockwise from upper left: light trapping (photo credit Luc Leblanc), beating sheet, pan trapping, sweep and aerial netting, photography, surface floating pupal exuviae. Photo credit Barbara Hayford unless otherwise noted.

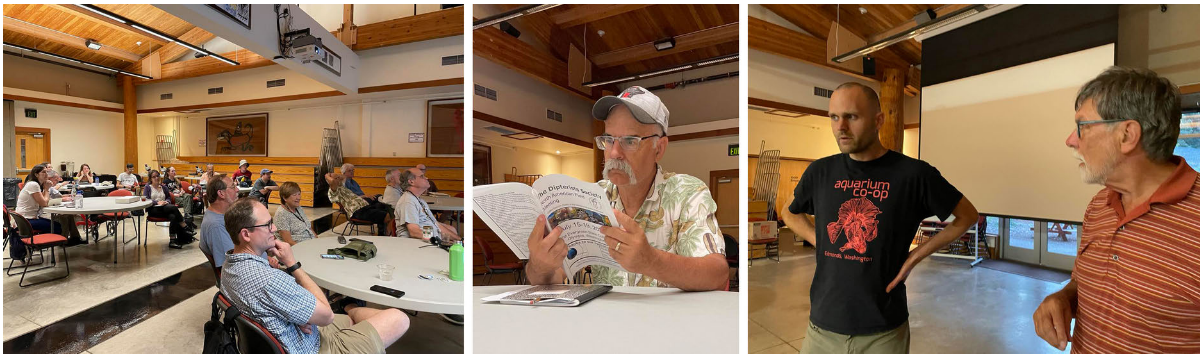
Figure 4. Field collecting during the 2024 Field Meeting. From upper left, clockwise: Zach Dankowicz, Stephen Luk, and Michael Palmer at Mima Mounds preparing to photograph and look for leaf miners, Zach Glass, Nina Miller and Megan Asche preparing specimens at the Chehalis River Surge Plain, Severyn Korneyev and Kristina Kernytska searching for suitable fruit fly habitat at the Chehalis River Surge Plain, Zach Dankowicz and Luc Leblanc at Mima Mounds, Bill Murphy examining specimens at the Chehalis River Surge Plain, Jeff and Sue Cumming seeking suitable sampling habitat at Mima Mounds. Photo credit Barbara Hayford.



In keeping with previous field meetings, the 2024 Field Meeting provided space and time for attendees to examine specimens collected during field work and to share specimens with each other. Luc Leblanc provided meeting buttons for the attendees and on the last evening of presentations brought his button maker to create unique buttons for all who wanted them (Fig. 9).

Figure 5 (left). Detail of the Welcome House, venue for the 2024 Field Meeting of the Dipterists Society on the campus of The Evergreen State College. Photo credit Barbara Hayford.

Figure 6 (below). Presentations and specimen examination and exchange were done in the Welcome House of The Evergreen State College. From left: participants watching a presentation, Jim Hogue reviewing the night's presentations, Andrew Fasbender and Karl McKnight discussing the day's events. Photo credit Barbara Hayford.



SAMPLING SITES

At least 11 sites were sampled during the 2024 Field Meeting of the Dipterists Society with habitats ranging from prairie and oak savannah to alpine and coastal forest to estuaries and streams. Sites were sampled prior to, during, and after the meeting by attendees. The following sites represent the field trips and venue sampling sites for the meeting.

The Evergreen State College Campus and Trails

Numerous trails cross 400 hectares comprised of coastal forest, streams, and saltwater shorelines provided access to diverse sampling opportunities (Fig. 10). In addition to daytime collecting, black-lighting sessions were held on several evenings during the meeting.

Chehalis Tidal Surge Plain (WDNR)

This large wetland area is spread out at the lower end of the Chehalis River, just upstream from where it empties into Grays Harbor. According to WDNR, this preserve contains the largest and best quality tidal surge plain wetland in Washington and one of the best along the west coast of the lower 48 states. Freshwater and estuarine marshes were surrounded by vegetative communities identified by WDNR as "Sitka spruce/red-osier dogwood/skunk cabbage vegetation, lady fern coastal herbaceous vegetation, soft stem bulrush herbaceous vegetation, red-osier dogwood–willow species shrubland, Lyngby's sedge herbaceous vegetation, and cattail surge plain herbaceous vegetation." The group initially sampled around the Preachers Slough East trailhead, before moving on to the Preacher's Slough West trailhead (Fig. 11).

Mima Mound NAP (WDNR)

Mima Mounds Natural Area Preserve (NAP) was the a unique site for sampling during the 2024 Dipterists Society Field Meeting (Fig. 12). The NAP was established in 1976 to protect rare examples of "Mima mound" landforms and Puget prairie grasslands. The site includes woodlands that differ from the other coastal forests we visited and include Douglas fir, deciduous trees, and a Garry oak woodland and savannah (widely spaced oak trees with grass understory). Prairie plants and grasslands were also present. Participants collected Diptera using sweep and aerial nets, beating sheets, and pan traps. However, due to the heat dome and drought that impacted western Washington during June and July 2024, Diptera were relatively scarce at this site.

West Rocky Prairie Wildlife Area Unit (WDFW)

This wildlife unit was like the Mima Mounds NAP but had more riparian habitat, though it lacked paths and was less accessible than the Mima Mounds NAP (Fig. 13). The WDFW conserves this unique south Puget Sound prairie ecosystem including a two-mile stretch of Beaver Creek and an Oregon white oak community that is uncommon on the west side of the Cascade Mountains. While the group who visited this site in the morning had little luck finding Tipuloidea, a second group who visited in the afternoon was successful in finding several Brachyceran taxa.

Olympic National Forest South Fork Skokomish Access (SKONF)

The South Fork Skokomish access for the Olympic National Forest provided access for an easy trail and hiking area through a subalpine to alpine forested ecosystem. The low wet coastal forests transition to subalpine forests at this site was characterized by Douglas fir, western white pine, and deciduous trees such as alder and maple. Streams and rivers crossed this site as well as a riparian wetland (Fig. 14). Collecting was initially focused on the stream-side riparian areas and the trail around the perimeter of Brown Creek Pond. In the afternoon participants split up and traveled up to collect at higher elevations surrounding the Skokomish valley.

Figure 7. Presentations clockwise from the upper left: Solange Akimana presenting on the *Tipula unicincta* group of desert craneflies, Zach Dankowicz presenting on his work as high school student in the USNM Diptera collections, Nina Miller sharing her work on urban hover flies, and Zach Glass talking about his work on the IUCN Red List and insect conservation. Photo credit Barbara Hayford.

SCHEDULE OF ACTIVITIES

The 2024 Field Meeting of the Dipterists Society was organized to emphasize collecting during the day and presentations and specimen examination during the evenings after dinner. Treats were provided for the evening presentations and some participants did light trapping on The Evergreen State College campus afterwards. Here is a brief summary of activities.

Monday, July 15 – Arrival of attendees, dinner, introductory presentation by Hayford and Fasbender, light trapping on The Evergreen State College campus.

Figure 8. Presentations from top to bottom, left to right: Bri White sharing methods for rearing mosquitos, Sarah Oliveria fielding questions on her Diptera conservation efforts in Brazil, Will Bouchard talking about his work on the taxonomy of the biting midge, *Diamesa*, John Stireman presenting work on the Afrotropical bristle flies, Michael Palmer describing research opportunities at his McKenzie Valley Bug Lab, Tristan McNight regaling participants with his adventures searching for the robber fly, *Lasiopogon pacificus* in British Columbia, Greg Dahlem talking about flesh flies and apple orchards, Severyn Korneyev sharing his research on the new world species of *Tephritis*, Luc Leblanc presenting on his work on fruit flies in the Pacific. Photo credit Barbara Hayford.

Figure 9. Luc Leblanc brought his button maker and created buttons celebrating the 2024 Field Meeting and then made buttons upon request for the meeting participants. Photo credit Barbara Hayford.

Figure 10. Coastal forest was available for sampling on The Evergreen State College campus with some help from the local black-tailed deer. Photo credit Barbara Hayford.

Tuesday, July 16 – Collecting in the Chehalis Tidal Surge Plain, Washington Department of Natural Resources (WDNR) with estuarine and coastal forest habitat, evening presentations, specimen examination and/or light trapping on The Evergreen State College campus.

Wednesday, July 17 – Collecting in the Mima Mound WDNR or West Rocky Prairie Wildlife Washington Department of Fish and Wildlife sites, which included oak savannah, stream and riparian habitat, and the unique Mima Mound prairies. Presentations were given after dinner followed by specimen examination and/or light trapping on The Evergreen State College campus.

Figure 11. Participants of the 2024 Field Meeting had two sites to choose from to sample along the Chehalis River Surge Plain. This tidally influenced area is some of the best coastal habitat along the Pacific Coast in the lower 48 states. Here Jim Hogue and Zell Smith collect near the backwater habitat of the Chehalis River. Photo credit Barbara Hayford.

Figure 12. The Mima Mounds, unique small hills with unknown origins, were the most unusual habitat available for sampling during the 2024 Field Meeting, but conditions were too dry to produce good results. Here Zach Dankowicz and Stephen Luk explore the site. Photo credit Barbara Hayford.

Figure 13. The West Rocky Prairie also had Mima Mounds but was more open to exploration. Photo credit Barbara Hayford.

Figure 14. The Olympic National Forest South Fork Skokomish site was the only subalpine site sampled during the 2024 Field Meeting. Gentle to steep alpine slopes and forest interlaced with wetlands and streams were sampled by participants. Photo credit Will Bouchard.

Thursday, July 18 – Collecting in the Olympic National Forest South Fork Skokomish Access which included subalpine forest, alpine stream, and riparian habitat. Presentations were given after dinner followed by specimen examination and/or light trapping on The Evergreen State College campus.

Friday, July 19 – Most attendees returned after breakfast or departed to explore the Pacific Northwest.

Outcomes

A preliminary list of site identifications has been shared by participants and will be delivered to permitting agencies and The Evergreen State College. To date, 175 taxa were identified from samples collected during the 2024 Field Meeting of the Dipterists Society (Fig. 15). Most identifications were made during the meeting to yield a preliminary list of Diptera biodiversity for the permitting agencies and The Evergreen State College. Clearly this list will grow over time as the participating researchers study the Diptera they collected during their time in the Pacific Northwest.

Figure 15. Some Diptera photographed during the 2024 Field meeting. From top left: *Chrysophilus tomentosus* (Photo Stephen Luk), *Phytoliriomyza melampyga* (Photo Michael Palmer), *Silvius gigantulus* (Photo Stephen Luk). From lower left: *Leptopeza disparilis* (Photo Zachary Dankowicz), *Scathophaga spurca* (Photo Zachary Dankowicz), *Tricholinocera dolicheretma* (Photo Zachary Dankowicz).

Costs

The cost for early registration was $510 (inclusive of lodging at The Evergreen State College and breakfast and dinner meals), while late registration was $600. Student registration remained the same at $350 through the end of registration on June 30. Costs were elevated relative to previous meetings because of the catered meals for breakfast and dinner and provided lodging. Attendees provided their own lunch for field work. The Dipterists Society provided grant funding totaling $2268 for travel and meeting participation for five of the participants (three students and two non-students).

Acknowledgements

We thank the Board of the Dipterists Society for funding and guidance for the 2024 Field Meeting. Early work and support for this meeting was provided by the Coastal Interpretive Center. Thanks to Bryce Winkelman and Jake Lind of The Evergreen State College for helping secure the venue and particularly to Jake for helping us with all our needs during the meeting. We are grateful for the

permission to sample provided by the Olympic National Forest (Nominal Effects Letter File Code: 2720), Washington Department of Fish and Wildlife (WDFW granted to the Coastal Interpretive Center, #CALDWELL 24-141), Washington Department of Natural Resources (Research Permit #2024-02), and The Evergreen State College. Thanks to Rhithron Associates, Inc. and Tethysphere sponsored refreshments for evening presentations.

Presentations given at the Dipterists Society 2024 Field Meeting

July 15

Barbara Hayford: “From the Palouse to the Pacific”

July 16

Solange Akimana: “Systematics and Historical Biogeography of the *Tipula unicincta* group (subgenus *Lunatipula*) of desert crane flies (Diptera: Tipulidae)”
Will Bouchard: “The pupae of *Diamesa* from the Nearctic region with 11 new descriptions, 1 redescription, and a key to known species”
Greg Dahlem: “Flesh flies (Sarcophagidae) from Pennsylvania apple orchards”
Zach Dankowicz:"Efforts of a high school volunteer in the USNM Diptera collection"
Karl McKnight: “Adult phenology and ethology of the robber fly *Lasiopogon currani* (Diptera: Asilidae) in a northeastern forest near Pierrepont, New York”

July 17

Andrew Fasbender: “What exactly is the *Eukiefferiella cyanea* group? (Chironomidae, Orthocladiinae)”
Zac Glass: “The IUCN Red List, experience and process of assessing underrepresented insect taxa”
Severyn Korneyev: “Revision of the New World species of the genus *Tephritis* (Diptera, Tephritidae, Tephritinae)”
Luc Leblanc: “The regional fruit fly projects in the Pacific”
Tristan McKnight: “Freelancing for flies: insights from preparing a COSEWIC status report on a long-lost robber fly, *Lasiopogon pacificus*”

July 18

Sarah Oliveira: “Diptera surveys in Conservation Units of the Brazilian Cerrado”
Michael Palmer: “Introducing the McKenzie Valley Bug Lab”
John Stireman: “The Afrotropical bristle flies (Diptera: Tachinidae)”
Bridget White: “Evaluating agar-based mosquito larval feeding formulations for *Aedes aegypti*”
Nina M. Emond Miller: “Flower visiting Syrphidae in urban gardens”

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Dipterists Society organized meeting wrap-up (Phoenix, Arizona, USA)

Charlotte Alberts[1], Allan Cabrero[1], Jessica P. Gillung[2]

[1] Smithsonian National Museum of Natural History, Washington, DC, USA
[2] Lyman Entomological Museum, McGill University, Sainte-Anne-de-Bellevue, Québec, Canada

The Organized Meeting of the Dipterists Society took place on Tuesday, November 12th from 7:00pm– 9:00pm MST during the Annual Meeting of the Entomological Society of America held in Phoenix, Arizona, USA. We received many abstract submissions and a total of 5 talks were given at the meeting. This was the first time the meeting has been held in a hybrid formant. A total of ~30 attended in person, with 17 virtual attendees via Zoom. Attendees were composed of undergraduate students, graduate students, postdoctoral researchers and other professionals. There was a wide range of interests and backgrounds represented, including many non-Diptera specialists who hoped to learn more about the order or connect with Diptera researchers.

This was the first meeting in which snacks and drinks were provided by the Dipterists Society. This was a huge success, boosting participation and engagement, and being well received and appreciated by all. This encouraged attendees to mingle and converse; in fact many stayed past the official end time of the meeting! We look forward to seeing you all in Portland next year!

Here are the 5 talks given at the meeting:

“Against all odds: the evolution of ecological niche-sharing in Hawaiian *Trupanea* flies”
by Spencer L. Pote, Graduate student at the University of Hawai’i at Mānoa, and Daniel Z. Rubinoff, Professor at the University of Hawai’i at Mānoa

“History and challenges of fruit fly eradication programs in Oceania”
by Luc Leblanc Curator of the William F. Barr Entomological Museum University of Idaho, Department of Entomology, Plant Pathology and Nematology (EPPN)

"Arctic blow fly biodiversity in Abisko, Sweden"
by Michael A. Monzón and Andrew Meeds, ASU Forensic Entomology & Wildlife Lab (FEWL) Graduate Student

“Resisting resistance: Identifying biochemical biomarkers for pyrethroid resistance in *Aedes aegypti* mosquitoes”
by Carla-Cristina Edwards, UC Davis Graduate Student

Team Diptera Announcement
by Allan Cabrero, Smithsonian National Museum of Natural History

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XII All-Russian Dipterological Symposium

Emilia Nartshuk

Zoological Institute, Russian Academy of Sciences, St Petersburg, Russia

The XII All-Russian Dipterological Symposium (https://www.zin.ru/conferences/dipt12/) was held at the Zoological Institute of the Russian Academy of Sciences in St. Petersburg from 21–24 October. The symposium was attended by 130 specialists (with co-authors of the papers) from 29 cities of Russia, as well as from Armenia, Belarus, Kazakhstan and Uzbekistan. There were 63 oral presentations and 18 poster presentations. Plenary and section reports were devoted to various aspects of the study of Diptera, including morphology, systematics, phylogeny, evolution, palaeoentomology, molecular genetic studies, karyosystematics and cytogenetics, faunistics, zoogeography, ecology and the importance of Diptera in parasitology, as well as their role in agriculture and nature conservation. In addition, reports dedicated to the memory of some world-famous Soviet and Russian dipterologists were presented for the All-Russian dipterological symposiums.

Photo by Vladimir Neimorovets.

The program and abstracts volume were published, and can be downloaded at:

https://www.zin.ru/conferences/dipt12/www/dipt12_program.pdf
https://www.zin.ru/conferences/dipt12/www/dipt_12_abstracts.pdf

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Arbovirus Surveillance and Mosquito Control Workshop

Rudy Xue

Anastasia Mosquito Control District, 120 EOC Drive, St. Augustine, FL 32092; rxue@amcdfl.org

The 20th Arbovirus Surveillance and Mosquito Control Workshop will be held in St. Augustine, Florida, March 25–27, 2025. The workshop has been sponsored by Anastasia Mosquito Control District, USDA/ Center for Medical, Agriculture, and Veterinary Entomology, and the Florida Mosquito Control Association.

For more information about the workshop and previous workshops and workshop proceedings, please visit the website at https://amcdsjc.org/.

If you would like to give a presentation at the workshop please contact Dr. Rudy Xue, rxue@amcdfl.org.

The deadline for submission of presentation titles will be January 20, 2025.

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International Forum for Surveillance and Control of Mosquitoes and Vector-borne Diseases

Rudy Xue

Anastasia Mosquito Control District, 120 EOC Drive, St. Augustine, FL 32092; rxue@amcdfl.org

The 9th International Forum for Surveillance and Control of Mosquitoes and Vector-borne Diseases will be held in XiAn, China, May 25–30, 2025. The Forum has been sponsored by the Entomological Society of China. Also the forum will be in conjunction with the National Congress of Medical and Veterinary Entomology and the 6th Asian Society of Vector Ecology and Mosquito Control membership meeting.

For more information about the conferences please visit the website at https://asiansvemc.org.

Or contact:
Dr. Chun-Xiao Li, vectorlcx@126.com
Dr. Tong-Yan Zhao, tongyanzhao@126.com
Dr. Rudy Xue, xueamcd@gmail.com

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DIPTERA ARE AMAZING!

The two photographs submitted for this issue are both by Zachary Dankowicz. The first one graces the cover of this issue – *Coenosia tigrina* feeding on a male of the *Condylostylus caudatus* species group, spotted at a park near the CNC in Ottawa. And the following photo of *Syrphus torvus* perched on the photographer's finger by the Little River in the Great Smoky Mountains National Park, Tennessee, USA.

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SOCIETY BUSINESS

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

No documents are provided in this issue, as the minutes of the annual meeting of the Board of Directors, held on 10 December 2024, will be approved prior to publication in the next issue.

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

1. We have increased our Board of Directors to 10 people (see below)
2. We are opening a Dipterists Society Gifting Account, which will allow for donations of stocks and equities, as well as cash. Terry and Faye Whitworth are generously contributing the first $50,000 to fund this investment account, which is designed to grow and be useful to the dipterists community into the future. Please keep an eye on the Society website for ways to donate!

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

Directors

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

Officers

Stephen Gaimari, President
Martin Hauser, Vice President
John Midgley, Treasurer
Giar-Ann Kung, Secretary & Education Chairperson
Matthew Bertone, Meeting Chairperson & General Meeting Chair
Barbara Hayford, Field Meeting Co-Chair
Andrew Fasbender, Field Meeting Co-Chair

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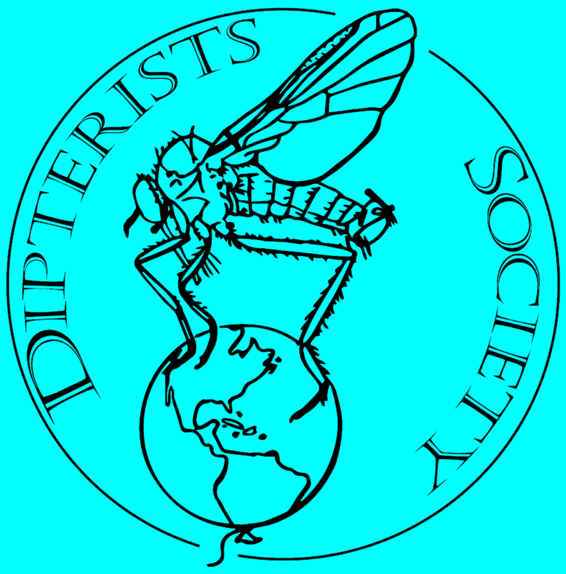

ISSN 2769-6111 (Print)
ISSN 2769-612X (Online)