

In Memoriam: D. Monty Wood

THE TACHINID TIMES

ISSUE 34



***D. Monty Wood
1933-2020***

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INSTRUCTIONS TO AUTHORS

This newsletter accepts submissions on all aspects of tachinid biology and systematics. It is intentionally maintained as a non-peer-reviewed publication so as not to relinquish its status as a venue for those who wish to share information about tachinids in an informal medium. All submissions are subjected to careful editing and some are (informally) reviewed if the content is thought to need another opinion. Some submissions are rejected because they are poorly prepared, not well illustrated, or excruciatingly boring.

Authors should try to write their submissions in a style that will be of interest to the general reader, in addition to being technically accurate. This is a newsletter, not *Science* or *Nature*. Try to illustrate submissions with high quality images sent to the editor as separate files at the same time as the text. Text files sent with embedded images will not be considered for publication. All content should be original; if copyrighted material (online or in print) is used then permission from the copyright holder is needed. Submitted pictures of tachinids in the field will be considered for the cover, table of contents, or a special section in the newsletter.

Student submissions are particularly welcome. Writing about a thesis study or a side project involving tachinids is a good way to inform others about a study that is underway before it has generated formal publications.

Please send submissions for the 2022 issue of *The Tachinid Times* to the editor by the end of January 2022.

FRONT COVER Monty Wood collecting at the summit of Eighty Mountain near Silver City, New Mexico, USA, during the 2007 NADS Field Meeting.

Photo: J.E. O'Hara, 15 August 2007

TABLE OF CONTENTS A male *Tachina* sp. (Tachinae, Tachinini) feeding from a flower in Lockett Meadow in the San Francisco Peaks, northern Arizona, USA.

Photo: J.E. O'Hara, 5 July 2017

BELOW Concern for the planet is expressed in "graffiti", such as shown here, throughout the Navajo Nation in the Four Corners area of USA.

Photo: J.E. O'Hara, 28 August 2015



Tachinidae from central Italy: an investigation of morphological and molecular diversity



by Alice Lenzi

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A deepening of our knowledge of biodiversity

has become more and more necessary in recent decades, especially in light of estimates that we have described just 20% of the living species on Earth (Mora *et al.* 2011, Wilson 2017). Fortunately, DNA barcoding (Hebert *et al.* 2003) has given a boost to the rate at which species can be recognized and delimited in hyperdiverse insects groups by detecting occurrences of both cryptic taxa (Hebert *et al.* 2004, Bickford *et al.* 2007) and highly variable taxa. Such studies have included the Tachinidae in recent years (Pohjoismäki *et al.* 2016, Lee *et al.* 2021).

During my Bachelor's studies at Sapienza University of Rome (Italy), I quickly became fascinated by the world of flies and especially the Tachinidae. Under the guidance of my supervisor, Prof. Pierfilippo Cerretti, I completed a B. Sc. thesis on tachinid systematics. I had by then become interested in ecobiology and began a Master's thesis on the topic of biodiversity assessment based on a total evidence approach, including morphological analysis and DNA barcoding. Prof. Cerretti was again my supervisor for this project. I recently completed my Master's and will briefly review its goals and discuss some of my findings.

My Master's thesis focused on the morphological and genetic characterization of inter- and intraspecific diversity of Tachinidae from the central Appennines in Italy. Two sites were chosen for the collection of specimens, both of them in protected areas bordering a national park, the Parco Nazionale D'Abruzzo, Lazio e Molise (PNALM), a significant hotspot for biodiversity in Italy. This park is just over 100kms east of Rome.

Figure 1. Malaise trap set in a small clearing among beech trees (1400m) in central Appennines, Italy.

Of course, such a project requires partnerships. Sampling was carried out with the help of Raggruppamento Carabinieri Biodiversità, the group responsible for protecting the natural resources in Italy and that manages some protected areas in Italian territory. Sequencing and molecular analyses were performed in collaboration with the Evolution Lab coordinated by Prof. Rudolf Meier of the National University of Singapore. Meier's research team has been a leader in DNA barcoding and molecular analyses on insects, and in particular Diptera, for many years (e.g., Meier *et al.* 2006, Srivathsan *et al.* 2018, Wang *et al.* 2018, Srivathsan *et al.* 2019, Yeo *et al.* 2020). Not only did they carry out the barcode sequencing for my project, but they also helped me with data processing, especially Sujatha Narayanan Kutty.

Fifteen Townes-style Malaise traps were set up in clearings among beech trees (Fig.1) at elevations from 1397 to 1637 metres above sea level. Flying insects were collected from July to October 2019 and the samples were preserved in bottles filled with 70% ethanol that I replaced every two weeks (Fig. 2).



Figure 2. Author at work during one trap check and bottle replacement.

In total, approximately 1000 tachinid flies were collected. From autumn 2019 to spring 2020 I dedicated myself to sorting and identifying the specimens to species level using the keys of Cerretti (2010) and with the assistance of the keys' author. After this was done, all the specimens were sent to Singapore University where the molecular analyses were carried out. DNA extraction and amplification followed new time-saving and inexpensive pipelines, including the use of QuickExtract™ DNA Extraction Solution that does not damage specimens, thus allowing for further morphological examination afterwards (Wang *et al.* 2018). The mitochondrial COI gene (313 bp, minibarcode) was sequenced using the cutting-edge MinION technology (Jain *et al.* 2016).

Sequences were compared with the ones stored in GenBank (<https://www.ncbi.nlm.nih.gov/genbank/>) and BOLD (<http://www.boldsystems.org>). I carried out barcode-based identifications considering matches with $\geq 97\%$ identity as valid. In addition, we sorted sequences into putative species (mOTUs) by Objective Clustering (Meier *et al.* 2006) and then constructed a neighbor-joining tree to obtain a graphic representation of the clusters.

My thesis focused on the data concerning 303 of the roughly 1000 tachinid specimens collected during the study. The others are still being processed, having been delayed due to complications from the Covid-19 pandemic and the lockdown period that occurred in the spring-summer of 2020 in Italy. These data, although belonging to a subset of the collected specimens, are already providing some interesting observations resulting from the comparison of both morphological and molecular analyses. This small number of samples has also allowed me to become familiar with the techniques and with the different results that can be obtained from such a total evidence approach. I here briefly report some of the more interesting results that I achieved and wrote about in my thesis.

In 60% of the specimens, the morphological identification corresponds to the molecular one (i.e., there is a $\geq 97\%$ match with one GenBank species). In the remaining 40% I found the following types of discrepancies between morphology and barcoding-based identifications:

- 1) some sequences correspond to species different from the morphological ones,
- 2) some sequences do not significantly match any barcode available in GenBank (i.e., no match $\geq 97\%$), and
- 3) others match multiple species (i.e., match $\geq 97\%$ with more than one GenBank species, with the same percentage).

These problematic matches may have resulted from errors in the morphological identifications, or in the labelling of specimens during analyses, or even

in the names associated with barcodes in GenBank or BOLD. However, some of the incongruences may, upon closer examination, reveal cryptic species or highly variable ones. For example, I identified three specimens as *Lomachantha parra* Rondani and one as *L. rufitarsis* Villeneuve (Fig. 3) based on morphology. The latter species is currently known only from the Middle East and Armenia. Even though the four flies differ in the colouration of their fore legs and abdomen (typically black in *L. parra* and orange in *L. rufitarsis*), they share the same barcode and were all identified (correctly we believe) as *L. parra* by comparison with sequences in GenBank. I found a similar situation with some specimens of *Phebellia* Robineau-Desvoidy. They differ in the presence/absence of well-developed setae on the posterodorsal surface of the hind coxae. According to the literature, the only species with these setae present is *P. nigricauda* Mesnil in Japan. Despite this morphological difference, the specimens all share the same barcode and are identified as *P. glauca* Meigen in GenBank (a species lacking setae on the hind coxae according to its description). Thus, my results likely indicate that in each of these two cases only one morphologically variable species is involved. I concluded at the end of all my analyses that the 303 specimens studied for my thesis belong to 73 species.



Figure 3. Specimen of *Lomachantha* Rondani with fore legs and part of abdomen orangish.

I also found that some congeneric species with similar and confusing characters are well divided by genetics (e.g., *Siphona* spp., *Dinera carinifrons* (Fallén) and *Dinera fuscata occidentalis* Ziegler), while others present a small genetic distance (i.e., <3%) even if they are clearly distinguishable by morphology (e.g., *Cylindromyia* spp.). In addition, the total evidence approach allowed me to also identify a possible new species, characterized by unique morphology and barcode (*Catharosia* sp.).

My early results agree with those obtained in recent works about Tachinidae in which morphological diversity is compared to differences in DNA barcodes (Pohjoismäki *et al.* 2016, Lee *et al.* 2021). However, there is still more to do: first of all, as soon as possible, my colleagues and I will resume work on the remaining 700 specimens and their barcodes, and then we will proceed with further analyses on morphology and sequences using also new investigative methods (e.g., other species delimitation methods and haplotype network analyses). By the end of the project we will be able to verify and compare the results of my thesis with those of the remaining specimens to better characterize this tachinid assemblage of the central Apennines. We can say at this point that the tachinid fauna of this study consists mostly of species that are recognizable by either morphology or barcodes but also has some cryptic species and highly variable species that can only be satisfactorily resolved by both morphology and barcodes.

This project is contributing new information about tachinid diversity in Italy and more barcodes to international databases. Moreover, it constitutes a starting point for further research into the hidden diversity and community composition of insects in our territory. In fact, once this work is completed, it would be interesting to extend the sampling on a national scale including different environments in order to get an overview of Italian insect diversity.

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Remarkable TACHINID DIVERSITY from Malaise trap samples in southern BRAZIL

by John O. Stireman III

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Introduction

Here, I report on Tachinidae recovered from two 6m Malaise traps from the Southern Atlantic Forest of Brazil, in the states of Paraná and Santa Catarina. The traps were erected in March 2015 by Allen Norrbom (U.S. National Museum of Natural History (USNM)) and Marcoandre Savaris (Universidade de São Paulo) to sample tephritid flies. This sampling was part of a larger collaborative project between Norrbom, Luciane Marinoni (Universidade Federal do Paraná, DZUP), and her students M. Savaris and Silvana Lampert, which was focused on Tephritidae, particularly the genus *Anastrepha*. Norrbom and technicians sorted much of the material to family, including Tachinidae. I borrowed this material from the USNM to examine the diversity and composition of the tachinid fauna. The traps collected a surprising number of estimated species (212) over a sampling period of less than one week, indicating a highly diverse tachinid fauna. I briefly examine the diversity and community composition of these communities.

Site details

Both sites are located in southern Brazil, in the general region of the southern Serra do Mar (Serra Geral) characterized by a Cfb-Oceanic Koppen climate type, which consists of a temperate oceanic climate with warm summers. This region consists of the southern portion of Brazil's Atlantic Forest (*Mata Atlântica*), a biome that is famous for its biodiversity and endemism, as well as for its highly endangered status (Eisenlohr *et al.* 2015, Ribiero *et al.* 2009). The Serra do Mar region has fared better than other Atlantic Forest areas in retaining about a third of its original forest cover, although this decreases as one moves inland (e.g., Araucaria region with 12.6% remaining, Interior region with 7% remaining; Ribiero *et al.* 2009). The sampling sites are located in the subtropical Araucaria moist forest ecoregion (also known as Paraná pine forest or mixed ombrophylous forest). One trap was located near the southern border of the state of Paraná, just north of Santa Catarina (FCC) in the Serra do Mar biogeographical sub-region, and the other was located near the southeastern border between Santa Catarina and Rio Grande do Sol (PNAS), in the Araucaria sub-region. The traps were situated approximately 340 km apart (Fig. 1).

FCC

BRAZIL: Paraná: Palmas, Linha Alegria, Fazenda Cerro Chato, 1224m, 26.50252°S, 51.67033°W, Malaise on hill in campo, 3–4 Mar 2015, M. Savaris & A.L. Norrbom [area with grass & shrub vegetation and gallery forest along a river]. This trap was placed in a region of pastureland with scattered forest patches and *Araucaria* gallery forest along the Rio Chopim (Fig. 1). Specifically, it was erected near the river in an area of grass and shrub vegetation next to the gallery forest. Fazenda Cerro Chato is a working ranch in a larger area of grassland with scattered patches of forest that is neighbored by larger areas of moist *Araucaria* forest. The trap was erected for two days, 3–4 March, 2015.

PNAS

BRAZIL: SC: Parque Nacional Aparados da Serra, Rio do Boi, river crossing, 29.20231°S 50.05032°W, 209m, Malaise, 6–9 Mar 2015, A. L. Norrbom & M. Savaris.

This trap was placed in El Parque Nacional Aparados da Serra. This small, ca. 10,000 Ha, scenic park was one of Brazil's first national parks and is characterized by dense *Araucaria* forest and deep canyons. The Malaise trap was placed at the bottom of Itaimbezinho Canyon, beside the Rio do Boi, which was surrounded by forested canyon slopes (Fig. 1). It was run for four days, from 6–9 March, 2015.

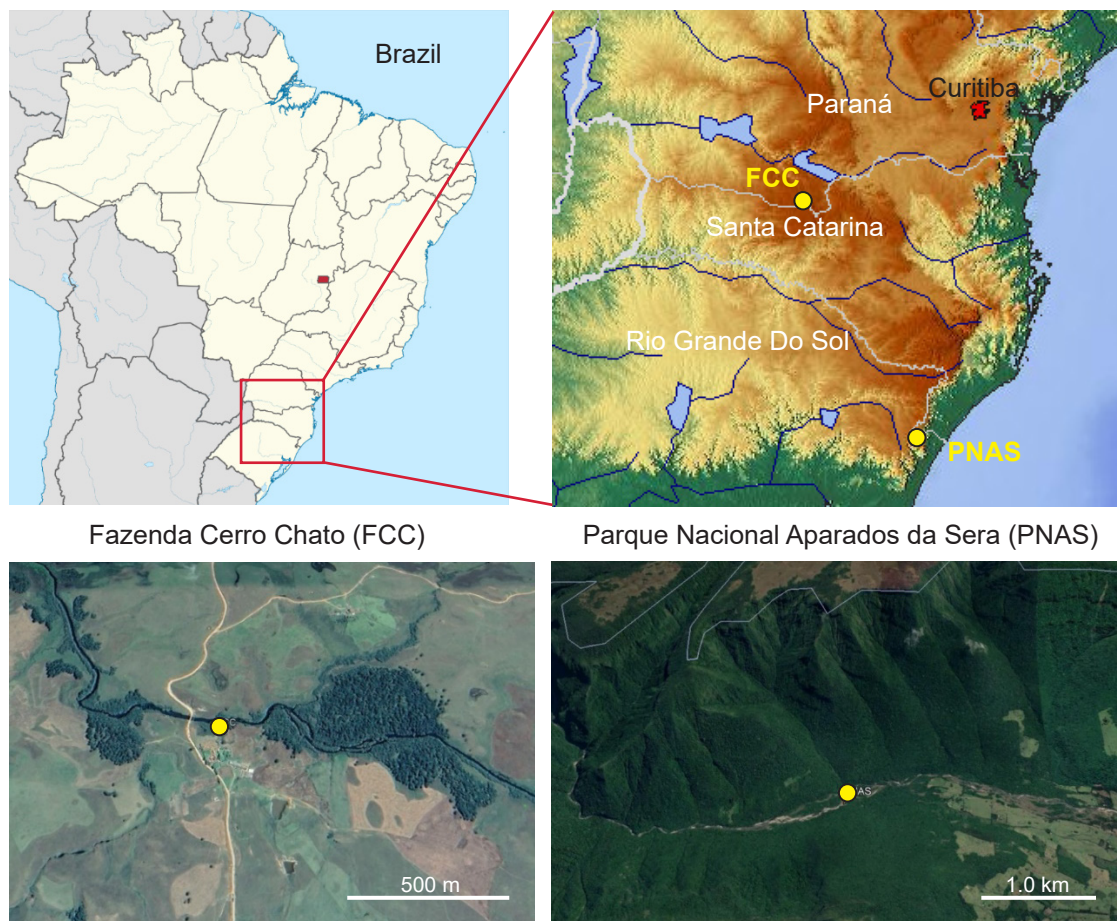


Figure 1. Top: Location of the trapping sites in southern Brazil. At left is an outline map of Brazil including states and at right is a zoomed in relief map with the sampling locations indicated by yellow circles. Bottom: Google Earth satellite views of the area surrounding each of the sites (not to same scale), with Fazenda Cerro Chato (left) situated in an open rangeland area with patchy forest and Parque Nacional Aparados da Serra, being surrounded by protected forest land.

Methods

Tachinid specimens were stored in vials with 70% ethanol for several years before I brought them to my laboratory at Wright State University, Dayton, OH, USA. I chemically dried the specimens in ethyl acetate before they were mounted on points or glued to pins (some large specimens were pinned before drying). Specimens were sexed and identified to genus with the help of keys in Wood & Zumbado (2010), Wood (1985) and Townsend (1927), and by comparisons with specimens in the Stireman (JOSC) collection at Wright State University. In assigning species to genera, I used the broad generic concepts of Wood & Zumbado (2010), and it is possible that some specimens may actually belong to other related named genera. Thus, in many cases the generic assignments are tentative, and this is often indicated by “G. nr.” or “?”. In some cases, the genus of a specimen could not be determined (even tentatively) and these are indicated as “nr. genus X” or “unknown genus”. Comprehensive generic keys for Brazilian tachinids are lacking except for the works of Townsend (1927, 1936–1941), in which the keys tend to be difficult to use and outdated in terms of synonymy. Townsend’s (1927) key to muscoid genera of the humid tropics (which does not explicitly cover the region sampled) was useful, but this work must be used with care as it appears to be based on limited representatives of each genus and my specimens often did not key to the correct genus. To make matters worse, it lacks illustrations and the abbreviations used for characters (to cut down on the length of couplets) must first be learned.

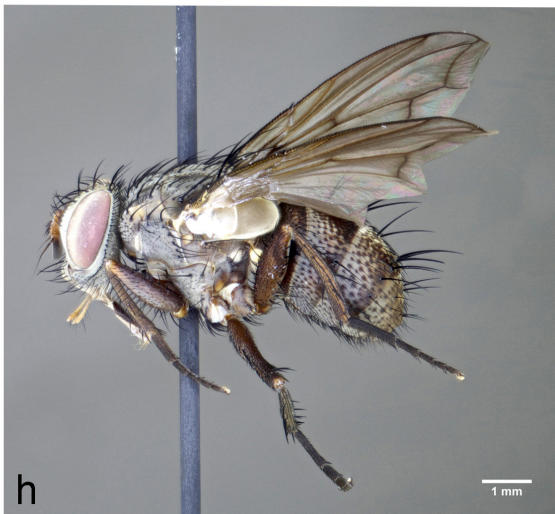
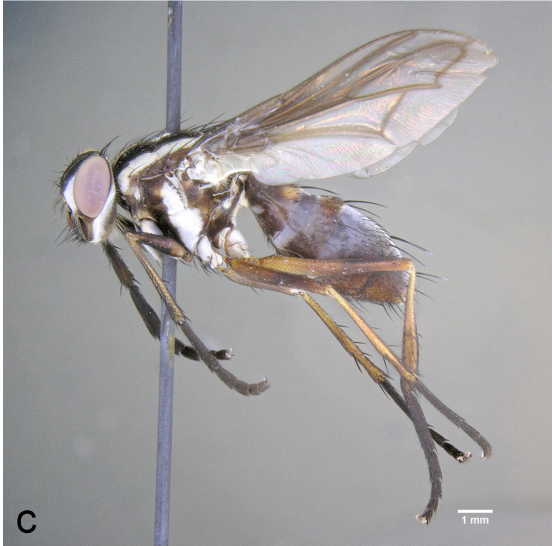
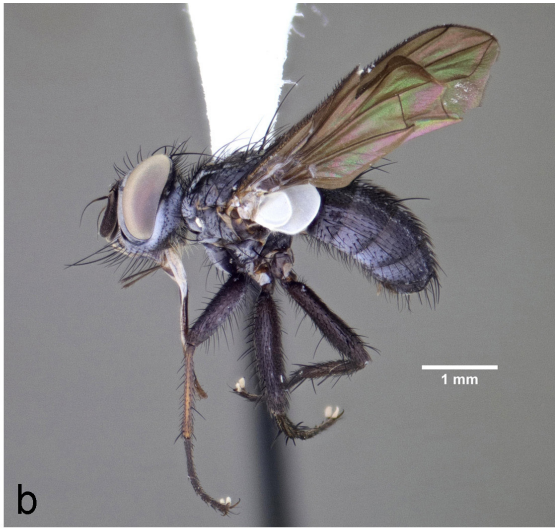
Once a tentative generic identification was established, specimens were sorted into morphospecies on the basis of external morphology. Such morphospecies designations are subject to several sources of error including the artificial separation of sexually dimorphic species or aberrant individuals and the lumping of morphologically cryptic species or those only separable by examination of terminalia. I have made notes in several instances where it is possible that two apparent morphospecies could represent one genetic species as well as where I may have lumped multiple genetic species into a single morphologically variable one. Genetic analyses of tachinid taxa have revealed both patterns; i.e., morphologically variable species that have been artificially separated (Lee *et al.* 2020) as well as cryptic species (Smith *et al.* 2007, Fleming *et al.* 2019). I probably erred in both directions, but if there is an overall bias in my morphospecies designations, it is probably towards underestimating the total number of species.

Data manipulation and quantification was conducted using the R programming language v. 3.6.0 (R core team 2019). Species rarefaction, interpolation, and extrapolation were conducted using the *iNEXT* package (Chao *et al.* 2014, Hsieh *et al.* 2016).

Results & Discussion

Over the six total trap-days, the combined traps collected 473 individual tachinids, comprising 266 males and 207 females. These specimens were divided among 212 morphospecies (Appendix 1), with 121 found at FCC (278 individuals; 2 days) and 115 found at PNAS (195 individuals, 4 days). A sample of some of the taxa that were recovered is shown in Fig. 2 (see discussion).

Figure 2 (see following page). A sample of the genera and morphospecies collected. Dexiinae: **a.** *Euoestrophasia* nr. *townsendi* sp. 1 (♀) (Dufouriini). **b.** *Epigrimyia* sp. 1 (♂) (Epigrimyini). **c.** *Euanthoides* sp. 1 (♀) (Sophiini). Exoristinae: **d.** *Eucelatoria* (or G. nr.) sp. 1 (♂) (Blondeliini). Phasiinae: **e.** *Xanthomelanopsis* sp. 2 (♂) (Gymnosomatini). Tachininae: **f.** *Actia* sp. 5 (♀) (Siphonini). **g.** *Copecrypta* sp. 1 (♀) (Tachinini). **h.** New genus 12 sp. 1 (♀) (Polideini).



As is suggested by the species and specimen totals, most species were represented by just one or a few individuals in each trap (Fig. 3). A total of 133 species were recorded only once, and an additional 32 only twice. This high number of singletons suggests that we have just scratched the surface of tachinid diversity in these areas. Species rarefaction curves support this conclusion, displaying high slopes and no sign of reaching an asymptote (Fig. 4). Extrapolated richness estimates for each sampling site at twice the actual sample sizes predict species richness of 172 and 190 species for PNAS and FCC respectively. Total species richness estimates based on the Chao estimator (Chao 1987) are 273 (95% CI: 196–422) for PNAS and 324 (229–504) for FCC. These diversity estimates rival that of any other site that has been examined thus far, even tropical forest sites (see Burington *et al.* 2020). For example, a single 2m Malaise trap that was operated for over a year in Ohio (U.S.) found a total of 117 species (Chao est. 190; Inclán and Stireman 2011), and another 2m Malaise trap operated in the Ecuadorian Andes (ca. 2000m) for six weeks recovered 138 species (Chao est. 240) (Burington *et al.* 2020). Even more impressive is that the tachinid species richness of these Brazilian Atlantic Forest sites is comparable to that recovered from other trapping studies despite only a few days of sampling versus months in other cases (Burington *et al.* 2020). Interestingly, although fewer species were recorded from the PNAS site, rarefaction curves for the current Brazilian trap samples resulted in a higher slope and potentially higher asymptote for this site relative to FCC.

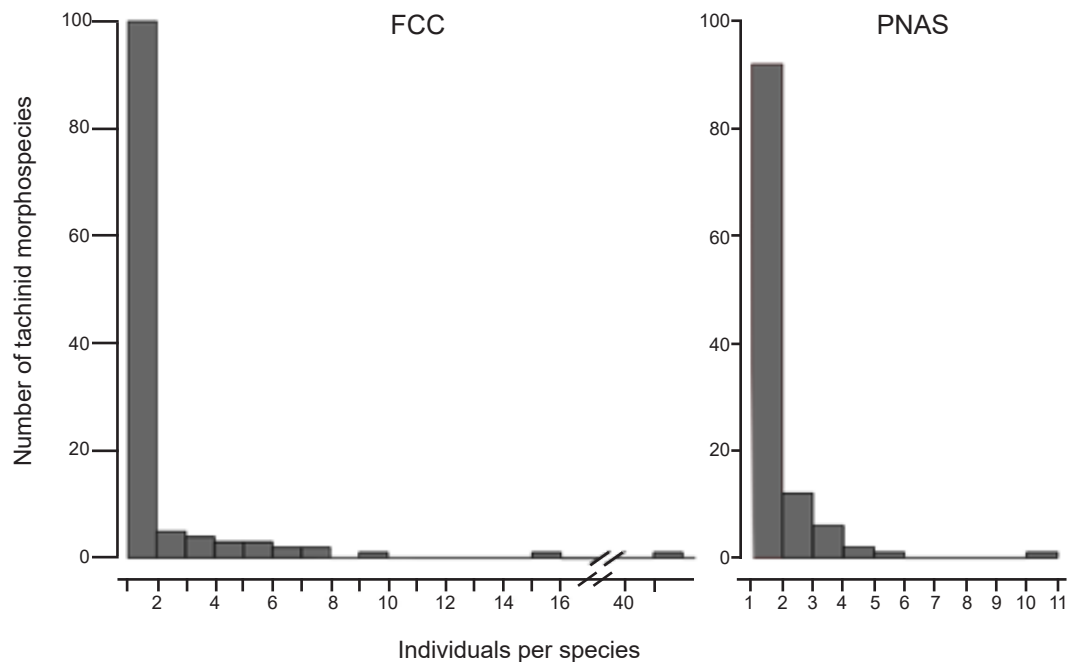


Figure 3. Histograms of the number of individuals per species in trap samples from each of the two sites (FCC left and PNAS right).

One factor contributing to the high diversity from these samples is the type of trap used. Anecdotal observations by myself and colleagues (e.g., J.E. O’Hara, G.A. Dahlem) suggest that the large 6m Malaise traps can be exceedingly effective at collecting tachinids, even when the results of other collecting methods, including standard 2m Malaise traps, are poor.

Only 22 species (about 10% of the total) were shared between the two sampling sites, resulting in high dissimilarity estimates (Bray-Curtis distance = 0.860, Jaccard distance = 0.925, where a value of one indicates maximum dissimilarity). However, this should not necessarily be interpreted as high species turnover or beta diversity between the sampling locations because so many of the species were represented by only one or a few individuals. More extensive sampling at each site could reveal many more shared species as the communities are more thoroughly characterized.

Composition

Trap samples of both communities were dominated by the tribes Blondeliini and Siphonini (Fig. 5; Appendix 1). These two tribes made up well over half of all recorded species and individuals. The rank of other tribes with regard to species richness and abundance was also similar between the two sites, with some exceptions. I briefly summarize some of the taxonomic patterns below.

Dexiinae (Fig. 2a–c)

Voriini comprised the third most species rich tribe at both sites, largely due to the genus *Campylocheta*, which was represented by an estimated 10 species. Otherwise, there were relatively few Dexiinae, except for a species of *Prosenia* found at FCC, which was the most abundant species recovered with 41 individuals (this resulted in Dexiini being the fourth ranked tribe in terms of abundance at FCC). Perhaps the most notable dexiines were the odd-looking *Euanthoides* spp. (Sophiini), with their boldly marked bodies, petiolate abdomens and extremely short, sunken, antennae (Fig. 2c). Four species of Dufouriini were collected, including three species of *Euoestrophasia* (all from FCC; Fig. 2a). The presence of the genus *Epigrimyia* (Epigrimyiini; Fig. 2b) at FCC was somewhat unexpected, as the genus was not previously known from South America (O’Hara *et al.* 2020), although its relative *Beskia aelops* (Walker), which was also collected, is known from throughout the Americas.

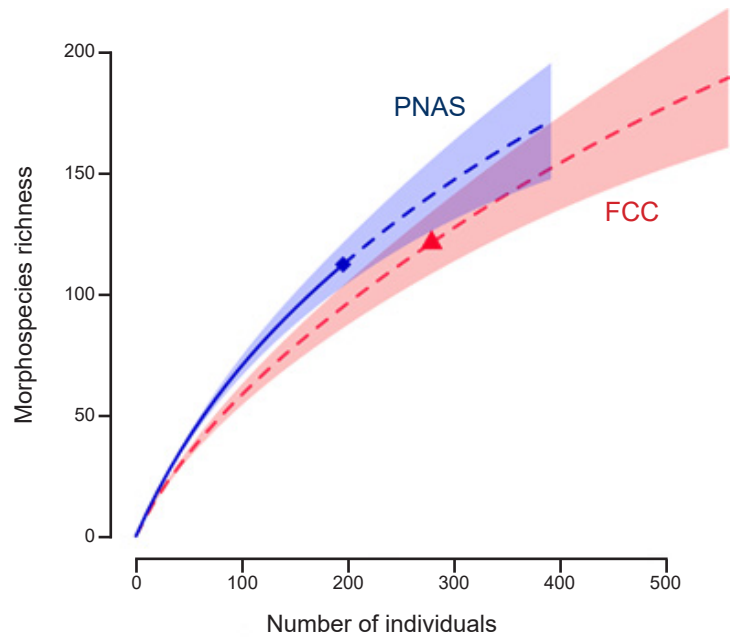


Figure 4. Rarefaction curves of species richness calculated for each site with 95% bootstrap confidence intervals (colored regions) using *iNEXT* (see text). Symbols indicate the observed values of species richness and total number of individuals. Dotted lines indicate extrapolated richness values to twice the observed number of individuals.

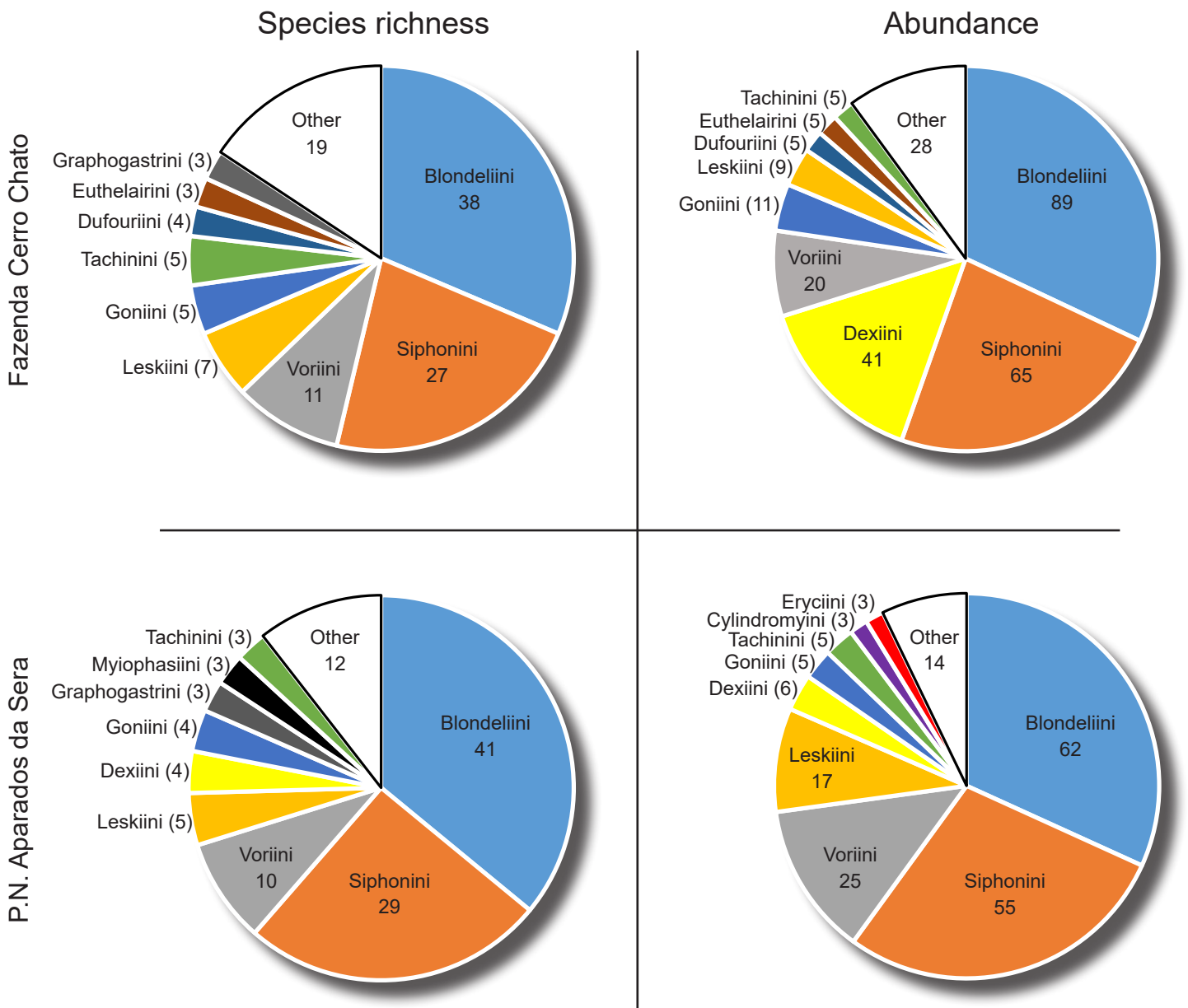


Figure 5. Pie charts showing the relative dominance of tachinid tribes in terms of species richness (left) and number of individuals (right) for traps at each sampling site (top: Fazenda Cerro Chato, bottom: P.N. Aparados da Serra). Numbers of species or individuals are given after tribe names.

Exoristinae (Fig. 2d)

Within the Blondeliini, the dominant genera included *Chaetostigmoptera* (7–8 spp.), *Eucelatoria* (8), *Lixophaga* (13), and *Myiopharus* (13). As stated previously, some of these generic placements are tentative and there were several blondeliines in which the genus was unclear (e.g., Fig. 2d). Goniini (8 spp.) and Eryciini (3 spp.) appeared to be under-represented given the large numbers of described species within these tribes, and Exoristini were entirely absent.

Phasiinae (Fig. 2e)

Very few phasiines were collected between the two traps, with only eight species in six genera being recorded (12 individuals, ~ 2.5% of the total; see Appendix 1). Of note was the finding of two species of *Xanthomelanopsis* (Fig. 2e) and two species of the unusual phasiine *Neobrachelia*.

Tachininae (Fig. 2f–h)

Siphonini dominated the contribution of the subfamily Tachininae to trap samples. *Siphona* was the most species rich genus in the samples, with 28 apparent species, although *Actia* (10 spp.; Fig. 2f) and *Ceromya* (8) were also well represented. Other tachinine groups that were relatively well represented include the Leskiini (12 spp.) and Graphogastrini (6 spp., all *Phytomyptera*). The relative lack of Tachinini (7 spp.) was notable for such a large tribe, although several species of *Copecrypta* (Fig. 2g) were collected. Of the two Polideini collected, one represents an unnamed genus (Fig. 2h, N. Genus 12; Perilla-López pers. comm.).

Interpretation and conclusions

It is clear from this preliminary study of Malaise trap samples from southern Brazil that this region harbors a rich and diverse tachinid fauna. It is surprising that more than 200 species were recovered from these traps over a span of only six days, and extrapolations from rarefaction hint at a much richer total fauna. As mentioned previously, part of the explanation for this great diversity of tachinids collected may be the high efficacy of 6m Malaise traps versus smaller traps or other survey methods. In addition, at both sites the traps were placed near waterways alongside gallery forests that could represent major flyways for tachinids. In this way, the traps may have sampled a much larger area than expected based on their size. I found it interesting that the FCC site harbored more species, given that it was sampled for a shorter period of time and the area is locally more impacted by grazing and other intensive land uses. However, this area likely possesses a greater variety of habitats including gallery forest, shrubby areas, grasslands, and pastures, which may enhance local tachinid diversity. The PNAS site, on the other hand, was surrounded by relatively unbroken forest, although just outside the park boundaries forest clearing and other human impacts are evident. Still, rarefaction analyses indicated that the PNAS site may actually possess greater diversity than FCC at comparable sample sizes of individuals, suggesting that the more intact forested site may hold more species. As indicated previously, the species turnover between the two sites was very high, but this may partly be an artifact of the large numbers of singletons, doubletons, etc.

The patterns of taxonomic composition of the sampled tachinids are difficult to interpret. The two sites had similar relative abundance and richness of tachinid tribes, suggesting that observed patterns are not just due to the local conditions of the traps. On the other hand, we know very little about how Malaise trap catches are biased relative to local community composition. The traps caught many individuals and species of smaller bodied taxa (e.g., Blondeliini, Siphonini, *Phytomyptera*, *Campylocheta*), which hand collecting is likely biased against. The paucity of Goniini+Eryciini, is however striking. A cursory examination of other tachinid trap data in the Neotropics (from upland sites) supports the dominance of Blondeliini and Siphonini and a relatively low diversity of Goniini and Eryciini, however Tachinini tend to comprise a much higher fraction of species in these other datasets (J.O. Stireman, unpub. data). Finally, it should be remembered that the traps were out for only a very short period at a particular time of year (early March). Sampling over a larger temporal scale may reveal a much different taxonomic composition as well as many additional species.

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Appendix 1. Table of species

Tachinid genera and morphospecies recorded from two traps in southern Brazil. M = number of males, F = number of females, Tot = total individuals, FCC = Fazenda Cerro Chato, PNAS = Parque Nacional Aparados da Serra (x's indicate species presence).

Species	M	F	Tot	FCC	PNAS	Notes
SUBFAMILY DEXIINAE						
Tribe Dexiini						
<i>Billaea</i> sp. 1	0	1	1		x	
<i>Billaea</i> sp. 2	0	1	1		x	
<i>Leptodexia</i> (?) sp. 1	0	1	1		x	
<i>Myiomima</i> (or <i>G.</i> nr.) sp. 1	2	1	3		x	
<i>Prosenia</i> sp. 1	35	6	41	x		
Tribe Dufouriini						
<i>Ebenia</i> sp. 1	0	1	1		x	
<i>Euestrophia</i> <i>aperta</i> (B. & B.)	1	0	1	x		
<i>Euestrophia</i> nr. <i>townsendi</i> Guimarães sp. 1	0	3	3	x		
<i>Euestrophia</i> nr. <i>townsendi</i> Guimarães sp. 2	0	1	1	x		
Tribe Epigrimyiini						
<i>Beskia aelops</i> (Walker)?	1	0	1	x		
<i>Epigrimyia</i> sp. 1	2	0	2	x		
Tribe Sophiini						
<i>Euanthoides</i> sp. 1	0	1	1	x		not <i>E. petiolata</i> Tnsd.
<i>Euanthoides</i> sp. 2	1	0	1	x		M of sp. 1?
Tribe Telothyriini						
<i>Telothyria</i> sp. 1	3	1	4	x		>1 sp.?
<i>Telothyria</i> sp. 2	1	0	1		x	
<i>Telothyria</i> sp. 3	0	1	1		x	F of sp. 2?
Tribe Uramyini						
<i>Itaplectops</i> sp. 1	1	0	1		x	
Tribe Voriini						
<i>Arrhinactia</i> sp. 1	0	1	1	x		
<i>Campylocheta</i> sp. 1	0	1	1	x		
<i>Campylocheta</i> sp. 2	0	1	1		x	
<i>Campylocheta</i> sp. 3	1	2	3		x	
<i>Campylocheta</i> sp. 3a	1	0	1		x	
<i>Campylocheta</i> sp. 4	8	2	10	x	x	
<i>Campylocheta</i> sp. 4a	3	0	3	x	x	= sp. 4?
<i>Campylocheta</i> sp. 4b	0	1	1		x	
<i>Campylocheta</i> sp. 5	3	2	5		x	
<i>Campylocheta</i> sp. 6	1	2	3		x	
<i>Campylocheta</i> sp. 7	3	1	4		x	= sp. 4 or sp. 5?
<i>Cyrtophloeoba</i> / <i>Minthoplagia</i> sp. 1	0	2	2	x		
<i>Micronychiops</i> sp. 1	3	0	3	x		

Species	M	F	Tot	FCC	PNAS	Notes
<i>Neosolieria</i> sp. 1	1	0	1	x		
<i>Neosolieria</i> sp. 2	0	1	1		x	
<i>Phasiophyto</i> sp. 1	0	1	1	x		
<i>Spathidexia</i> sp. 1	0	2	2	x		
<i>Trafoia</i> or <i>G.</i> nr. sp. 1	1	0	1	x		
<i>G.</i> nr. <i>Trochilodes</i> sp. 1	0	1	1	x		
SUBFAMILY EXORISTINAE						
Tribe Blondeliini						
<i>Admontia</i> or <i>G.</i> nr. sp. 1	1	0	1	x		
<i>Admontia</i> sp. 1	1	0	1	x		
<i>Anisia</i> sp. 1	4	1	5	x		F diff., >1 sp. ?
<i>Borgmeiermyia</i> or <i>G.</i> nr. sp. 1	1	7	8	x	x	
<i>Borgmeiermyia</i> or <i>G.</i> nr. sp. 2	0	1	1		x	
<i>Calodexia</i> sp. 1	0	3	3		x	
<i>Calodexia</i> sp. 2	0	1	1		x	
<i>Calodexia</i> sp. 3	1	1	2		x	
<i>Calolydella</i> sp. 1	0	1	1	x		
<i>Celatoria</i> sp. 1	0	1	1	x		
<i>Celatoria</i> sp. 2	0	2	2	x		
<i>Chaetodoria</i> sp. 1	2	0	2	x		
<i>Chaetona</i> sp. 1	0	1	1		x	
<i>Chaetonodexodes</i> sp. 1	0	1	1		x	
<i>Chaetostigmoptera</i> <i>G.</i> nr. sp. 1	0	3	3		x	
<i>Chaetostigmoptera</i> sp. 1	0	2	2		x	
<i>Chaetostigmoptera</i> sp. 1a	0	1	1		x	
<i>Chaetostigmoptera</i> sp. 2	1	0	1	x		
<i>Chaetostigmoptera</i> sp. 3	8	0	8	x		
<i>Chaetostigmoptera</i> sp. 4	1	0	1		x	
<i>Chaetostigmoptera</i> sp. 5	0	4	4	x		
<i>Chaetostigmoptera?</i> sp. 6	1	1	2	x		
<i>Erythromelana</i> cf. <i>leptoforceps</i> (Inclán)	2	0	2		x	
<i>Eucelatoria</i> cf. <i>strigata</i> (van der Wulp)	0	2	2		x	
<i>Eucelatoria</i> sp. 1	11	6	17	x	x	F diff., >1 sp. ?
<i>Eucelatoria</i> sp. 3	0	1	1		x	
<i>Eucelatoria</i> sp. 4	0	1	1	x		
<i>Eucelatoria</i> sp. 4a	1	0	1	x		
<i>Eucelatoria</i> sp. 4b	0	1	1		x	
<i>Eucelatoria</i> sp. 5 <i>aurata</i> (Tnsd.) grp.	0	1	1		x	
<i>Eucelatoria</i> sp. 6 <i>tinensis</i> (Tnsd.) grp.	0	1	1		x	
<i>Eucelatoria</i> sp. 7	0	1	1		x	
<i>Italispidea</i> sp. 1	1	0	1	x		
<i>Italispidea</i> sp. 2	4	0	4		x	1M diff., >1 sp. ?
<i>Leptostylum</i> sp. 1	0	1	1		x	
<i>Lixophaga</i> (or <i>Calolydella</i>) sp. 1	1	0	1	x		

Species	M	F	Tot	FCC	PNAS	Notes
<i>Lixophaga</i> sp. 1	1	0	1	x		
<i>Lixophaga</i> sp. 2	10	0	10	x		
<i>Lixophaga</i> sp. 2b	0	1	1		x	F of sp. 2?
<i>Lixophaga</i> sp. 3	1	2	3		x	
<i>Lixophaga</i> sp. 5	0	2	2	x		
<i>Lixophaga</i> sp. 6	0	1	1		x	
<i>Lixophaga</i> sp. 7	0	1	1	x		
<i>Lixophaga</i> sp. 8	2	0	2	x		
<i>Lixophaga</i> sp. 9	1	0	1	x		
<i>Lixophaga</i> sp. 10	0	1	1	x		
<i>Lixophaga</i> sp. 11	0	1	1	x		
<i>Lixophaga</i> sp. 12	0	1	1	x		
<i>Lydinolydella</i> or G. nr. sp. 1	1	0	1	x		<i>Opsomeigenia?</i>
<i>Myiodoriops</i> (or <i>Lixophaga</i>) sp. 1	4	0	4	x		nr. <i>Erythromelana</i>
<i>Myiopharus</i> sp. 1	0	4	4		x	1F diff., >1 sp.?
<i>Myiopharus</i> sp. 2	1	2	3		x	
<i>Myiopharus</i> sp. 2a	1	0	1	x		
<i>Myiopharus</i> sp. 3	0	1	1		x	
<i>Myiopharus</i> sp. 4	0	3	3	x		
<i>Myiopharus</i> sp. 5	0	1	1		x	
<i>Myiopharus</i> sp. 6	3	1	4		x	
<i>Myiopharus</i> sp. 7	1	0	1	x		
<i>Myiopharus</i> sp. 8	0	1	1		x	
<i>Myiopharus</i> sp. 9	0	1	1		x	
<i>Myiopharus</i> sp. 10	0	1	1	x		
<i>Myiopharus</i> sp. 11	0	1	1		x	
<i>Myiopharus</i> sp. 12	1	0	1		x	
<i>Phyllophilopsis</i> (G. nr.) sp. 1	1	0	1		x	
<i>Steleoneural/Trigonospila</i> G. nr. sp. 1	0	1	1		x	
<i>Thelairodoriopsis</i> sp. 1	0	1	1		x	
<i>Zaira</i> or G. nr. sp. 1	0	1	1	x		
<i>Zaira</i> or G. nr. sp. 2	0	2	2	x	x	
Blondeliini unk. G. 1 nr. <i>Eucelatoria</i> sp. 1	2	0	2	x		
Blondeliini unk. G. 2 nr. <i>Celatoria?</i> sp. 1	1	0	1	x		
Blondeliini unk. G. 3 nr. <i>Leptostylum</i> sp. 1	0	1	1		x	
Unknown genus (Blondellini?) sp. 1	0	1	1	x		
Tribe Eryciini						
<i>Ametadoria</i> sp. 1	1	1	2		x	
<i>Drino</i> sp. 1	1	0	1	x		
<i>Zizyphomyia</i> (G. nr.?) sp. 1	0	1	1		x	
Tribe Euthelairini						
<i>Neomintho/Pelecotheca</i> sp. 1	2	0	2	x		
<i>Pelecotheca</i> sp. 2	1	1	2	x	x	
<i>Pelecotheca</i> sp. 3	2	0	2	x		

Species	M	F	Tot	FCC	PNAS	Notes
Tribe Goniini						
<i>Choeteprosopa</i> sp. 1	0	1	1		x	
<i>Chrysoexorista</i> sp. 1	0	1	1	x		
<i>Hyphantrophaga</i> (G. nr.?) sp. 2	1	0	1	x		
<i>Hyphantrophaga</i> sp. 1	0	2	2		x	
<i>Leschenaultia</i> sp. 1	0	1	1		x	
<i>Patelloa</i> sp. 1	0	6	6	x		
<i>Patelloa</i> sp. 2	0	1	1	x		
<i>Patelloa</i> sp. 3	1	2	3	x	x	
SUBFAMILY EXORISTINAE						
Tribe Cylindromyiini						
<i>Neobrachelia</i> sp.	2	0	2		x	
<i>Neobrachelia</i> sp. 2	0	1	1		x	
Tribe Gymnosomatini						
<i>Gymnoclytia</i> sp. 1	1	1	2	x		
<i>Xanthomelanopsis</i> sp. 1	1	0	1		x	
<i>Xanthomelanopsis</i> sp. 2	1	1	2	x		
Tribe Leucostomatini						
<i>Clairvillia</i> sp. 1	1	0	1	x		
Tribe Phasiini						
<i>Phasia</i> sp. 1	0	1	1	x		
Tribe Strongygastrini						
<i>Strongygaster</i> sp. 1	0	2	2	x		
SUBFAMILY TACHININAE						
Tribe Ernestiini						
<i>Linnaemya</i> sp. 1	0	1	1		x	
Tribe Graphogastrini						
<i>Phytomyptera</i> sp. 1	1	0	1	x		
<i>Phytomyptera</i> sp. 1b	1	0	1	x		
<i>Phytomyptera</i> sp. 2	1	0	1		x	
<i>Phytomyptera</i> sp. 3	1	0	1	x		
<i>Phytomyptera</i> sp. 4	0	1	1		x	
<i>Phytomyptera</i> sp. 5	0	1	1		x	
Tribe Leskiini						
<i>Clausicella</i> sp. 1	1	0	1	x		
<i>Clausicella</i> sp. 2	0	2	2		x	
<i>Clausicella</i> sp. 2a	1	0	1	x		
<i>Genea</i> (<i>Geneopsis</i>) sp. 2	1	0	1	x		
<i>Genea</i> sp. 1	3	0	3	x		
<i>Ginglymia</i> sp. 1	8	3	11		x	>1 sp.? M = F?
<i>Ginglymia</i> sp. 2	1	0	1		x	
<i>Leskia</i> sp. 1	1	0	1	x		
<i>Leskia</i> sp. 2	1	0	1	x		

Species	M	F	Tot	FCC	PNAS	Notes
<i>Trochiloleskia</i> G. nr. sp. 1	0	1	1	x		
<i>Trochiloleskia</i> G. nr. sp. 2	0	1	1		x	
<i>Urummyobia</i> sp. 1	0	2	2		x	
Tribe Megaprosopini						
<i>Acronacantha</i> (G. nr.) sp. 1	0	1	1	x		
Tribe Minthoini						
<i>Actinochaeta</i> sp. 1	0	1	1		x	
<i>Paradidyma</i> sp. 1	0	1	1	x		
<i>Paradidyma</i> sp. 2	0	1	1	x		
Tribe Myiophasiini						
<i>Cholomyia</i> sp. 1	0	1	1	x		
<i>Cholomyia</i> sp. 2	0	1	1		x	
<i>Cholomyia</i> sp. 3	0	1	1		x	
<i>Cholomyia</i> sp. 4	0	1	1		x	
<i>Gnadochaeta</i> sp. 1	1	0	1	x		
Tribe Polideini						
<i>Chrysotachina</i> cf. <i>willistoni</i> Curran	1	0	1	x		
N. Genus 12 sp. 1	0	1	1	x		
Tribe Siphonini						
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 14	1	2	3	x	x	
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 15	1	0	1	x		
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 16	1	0	1	x		
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 17	0	1	1		x	
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 18	0	5	5		x	
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 19	10	1	11	x	x	
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 20	1	1	2	x		
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 20a	1	0	1	x		= sp. 20?
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 21	0	1	1		x	
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 22	0	1	1	x		
<i>Siphona</i> (<i>Pseudosiphona</i>) sp. 23	1	0	1	x		
<i>Siphona</i> (<i>Siphona</i>) sp. 1	1	1	2	x		
<i>Siphona</i> (<i>Siphona</i>) sp. 2	1	0	1		x	
<i>Siphona</i> (<i>Siphona</i>) sp. 2a	6	0	6	x		
<i>Siphona</i> (<i>Siphona</i>) sp. 3	0	3	3		x	
<i>Siphona</i> (<i>Siphona</i>) sp. 3a	3	0	3	x		M of sp. 3?
<i>Siphona</i> (<i>Siphona</i>) sp. 4	1	0	1	x		
<i>Siphona</i> (<i>Siphona</i>) sp. 5	0	3	3	x		
<i>Siphona</i> (<i>Siphona</i>) sp. 6	3	0	3	x	x	
<i>Siphona</i> (<i>Siphona</i>) sp. 6a	1	1	2	x	x	= sp. 6?
<i>Siphona</i> (<i>Siphona</i>) sp. 7	5	2	7	x	x	2 spp.?
<i>Siphona</i> (<i>Siphona</i>) sp. 8	2	4	6		x	
<i>Siphona</i> (<i>Siphona</i>) sp. 8a	6	2	8	x	x	= 8a?, or > 1 sp.?
<i>Siphona</i> (<i>Siphona</i>) sp. 9	1	2	3	x	x	
<i>Siphona</i> (<i>Siphona</i>) sp. 10	2	1	3	x	x	F diff. sp.?

Species	M	F	Tot	FCC	PNAS	Notes
<i>Siphona (Siphona)</i> sp. 11	1	0	1	x		
<i>Siphona (Siphona)</i> sp. 12	1	0	1		x	
<i>Siphona (Siphona)</i> sp. 13	0	1	1	x		
Tribe Tachinini						
<i>Archytas?</i> sp. 1	1	0	1	x		
<i>Copecrypta</i> sp. 1	0	2	2	x	x	
<i>Copecrypta</i> sp. 2	0	1	1		x	= sp. 1?
<i>Copecrypta</i> sp. 3	3	0	3		x	
<i>Gymnommopsis?</i> sp. 1	1	0	1	x		
<i>Neosarromyia</i> sp. 1	1	0	1	x		
<i>Parepalpus</i> sp. 1	0	1	1	x		
Totals	266	207	473			

Misidentifications have consequences: examples from Tachinidae (Diptera) and recommendations

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We recently came across a couple of published studies that inspired us to write this cautionary note about misidentifications and their consequences. We review the circumstances of each and offer a few recommendations to authors and editors about how to help recognize misidentifications before or after publication. Both of our examples (coincidentally) concern field studies of the fall armyworm, *Spodoptera frugiperda* (J.E. Smith) (Lepidoptera: Noctuidae).

Fall armyworm in Mexico

Pierfilippo discovered our first misidentifications while searching for host data on the New World genus *Distichona* van der Wulp, 1890. He found a report of *Distichona auriceps* Coquillett (Exoristinae, Goniini) parasitizing the fall armyworm in Guanajuato, Mexico. The study was published in Spanish by Salas-Araiza & González-Márquez (2017) and (in less detail) in English by Salas-Araiza (2017). The study also reported *Hypovoria discalis* (Brooks) (Dexiinae, Voriini) parasitizing *S. frugiperda*. Prior to this study, hosts were unknown for the eight species of *Distichona* or five species of *Hypovoria* Villeneuve, although both *D. auriceps* and *H. discalis* were known from Mexico (O'Hara *et al.* 2020).

Images of reared specimens of "*Distichona auriceps*" and "*Hypovoria discalis*" were included in Salas-Araiza & González-Márquez (2017: 291, as Fig. 1 and Fig. 2, respectively) but not in Salas-Araiza (2017). Both identifications are wrong to subfamily: their "*D. auriceps*" is a member of the tribe Tachinini (Tachininae) and their "*H. discalis*" is a member of the tribe Winthemiini (Exoristinae).

Fall armyworm in Indonesia

Our second example came to light for quite a different reason. Shannon Henderson (Jim's technician) was searching for new tachinid taxa and new distributions in recently-published papers to update their tachinid database and checklist (O'Hara *et al.* 2020). Two distributions in Ginting *et al.* (2020) looked suspicious: records of the New World species *Archytas marmoratus* (Townsend) and *Winthemia trinitatis* Thompson as parasitoids of fall armyworm in Sumatra, Indonesia. Both tachinids are known parasitoids of fall armyworm (Guimarães 1977) but neither has been recorded from the Old World. Jim noticed that neither tachinid had been reared from fall armyworm; instead, flies found on corn plants infested with fall armyworm were photographed and identified as

“*Winthemia trinitatis*” and “*Archytas marmoratus*” (Ginting *et al.* 2020: 112, as Fig. 5a and Fig. 5b, respectively). Both identifications are wrong to family: their “*Winthemia trinitatis*” is a species of Calliphoridae and their “*Archytas marmoratus*” is a species of Sarcophagidae.

We contacted the editors of the journal *Serangga* and informed them of the misidentifications published in their journal, in the paper by Ginting *et al.* (2020). A correction will be published in an errata in the April 2021 issue of *Serangga*.

Recommendations

The misidentifications above caused two different types of errors to enter the world literature on Tachinidae. In the first case, two wrong host records were reported, in the second two wrong distributions were involved. If not for the published images in both papers, these errors of identification would have been difficult to detect.

Tachinid misidentifications are bound to happen for a variety of reasons including carelessness, difficulty with identifications, and unresolved taxonomic issues. It is primarily the responsibility of authors to ensure that identifications are correct in their papers but editors bear some of responsibility as well. Misidentifications could be reduced in number, or identifications later checked, if the following recommendations were adopted by journals publishing host records:

- 1) ask authors to deposit voucher specimens of reared tachinids in a permanent collection,
- 2) ask authors to publish pictures of reared tachinids, and
- 3) ask a systematist familiar with tachinids to review the manuscript to assess the likelihood of correct identifications.

The process of identifying tachinid specimens to genus or species can be difficult or impossible for non-specialists, and sometimes for specialists as well. DNA barcoding (of the COI gene) of reared tachinids is an option for authors if they have the financial and/or laboratory means to take advantage of this technology. The world inventory of DNA barcodes is growing and the accuracy of species identifications for Tachinidae is very high, greater than 95%.

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Impact of pesticides on an insect with double ecosystem services: the case of *Exorista larvarum* (Linnaeus)

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Pollinator's biodiversity and their importance

Animal pollination is a key ecosystem service that allows the maintenance of wild and cultivated plant species. Pollinators such as bees, birds, bats and mammals have an extremely important role in determining food availability for other animals. In terms of human food supply, it has been estimated that approximately 35% of global crops benefit from animal pollination services (Klein *et al.* 2007). Insect pollination for food production has an evaluated value of around \$361 billion USD/year, worldwide (Lautenbach *et al.* 2012). Within the impressively diverse Insecta, there is thought to be approximately 150,000 flower-visiting species throughout the world, including bees, flies, butterflies, moths, wasps, ants and beetles (FAO 2008).

Among all the insect pollinators, bees are undoubtedly the most valuable. The managed western honey bee, *Apis mellifera* L., is the most commonly used insect due to its versatility and ubiquitous distribution (Winfrey *et al.* 2007, Arena & Sgolastra 2014). Yet, the great majority of pollinators are wild species. In fact, wild bees can improve seed set and quality of agricultural goods, and even enhance the honey bee effectiveness, thereby increasing the commercial value of several crops (Chagnon *et al.* 1993, Greenleaf & Kremen 2006, Rader *et al.* 2016). In addition, a meta-analysis of 39 field studies, across five continents, showed that non-bee insects were responsible for 39% of the total number of crop flower visits, strongly demonstrating the importance of other insect orders for pollination (Rader *et al.* 2016). Wild bees and non-bee insects can therefore provide these services, and even in some cases more efficiently than honey bees (Garibaldi *et al.* 2013, Orford *et al.* 2015). Moreover, Diptera families, such as Syrphidae and Tachinidae, not only provide pollination services as adults, but are also major biocontrol agents as larvae due to their entomophagous activity towards insect pests. This well demonstrates the importance of biodiversity in increasing and stabilizing ecosystem services.

Pollinators in decline

Worldwide, high losses in honey bee colonies and in the diversity and abundance of wild pollinators have been noticed both in agricultural landscapes and natural habitats (Burkle *et al.* 2013). Recent studies have associated these declines with a variety of interacting factors, such as climate change, pathogen and parasite loads, pesticide use and habitat loss (Goulson *et al.* 2015). These stressors are collectively reducing the resources used by pollinators for foraging, nest opportunities and shelter. High profile examples of the negative impacts of agricultural intensification and land-use changes include many native European butterflies (FAO 2008) and the North American monarch butterfly (*Danaus plexippus* (L.)) (Olaya-Arenas & Kaplan 2019), which are facing serious long-term declines of their populations.

The role of wild bees and non-bee insects in providing pollination has often been overlooked and there are few studies currently available on the potential effects of agricultural practices on these insects (Uhl & Brühl 2019). Agricultural intensification characterized by a high use of pesticides may expose wild pollinator species to a plethora of pesticide mixtures. In addition, until now, the current risk assessment of pesticides regarding pollinators has relied only on honey bees (OEPP/EPPO 2010). The risk assessment schemes for pesticides have assumed that the worst-case scenarios projected for honey bees are sufficiently conservative to protect other pollinator species, or even that the predictions for these other species can be extrapolated directly from honey bees. However, different bee species have different sensitivities to pesticides and routes of exposure, which depend on their specific life history traits (Arena & Sgolastra 2014, Sgolastra *et al.* 2019). The extrapolation from honey bees could be even more hasty if we consider non-bee insects (Uhl & Brühl 2019). These differences highlight the necessity to properly evaluate the effects of pesticide use on these important ecosystem service providers.



Figure 1. Adult male and female of *E. larvarum*. (Photos by C. Martins.)

Case study: *Exorista larvarum* (Linnaeus) (Diptera: Tachinidae) (Fig. 1)

In the framework of my doctoral thesis, under the guidance of Prof. Fabio Sgolastra and in collaboration with Prof. Maria Luisa Dindo (both at University of Bologna, Italy), I aim to assess the impacts of pesticides on non-*Apis* pollinators and develop new test methods that can be used in the risk assessment scheme. To this end, we selected the tachinid *Exorista larvarum* as a study species. *Exorista larvarum* is a gregarious larval parasitoid recorded as a natural enemy of several lepidopteran pests of forest and agricultural interest (Cerretti & Tschorsnig 2010, Tschorsnig 2017, Dindo *et al.* 2019). It is widespread in Europe, Asia and North Africa, and is established in North America where it was introduced as a biological control agent of the gypsy moth, *Lymantria dispar* (L.) (Sabrosky & Reardon 1976). Although it is well-known as

a parasitoid, *E. larvarum* can also provide pollination services, since it visits flowers as an adult to feed. It is well known that most adult parasitoids (including *E. larvarum* and other tachinids) feed on pollen and nectar and these provide them with energy and may also positively influence egg maturation (Thompson & Hagen 1999, Wäckers 2003). European tachinids visit many flowering plants, especially in the family Apiaceae (Mellini & Coulibaly 1991). Interestingly, laboratory studies have shown that honey bee-collected pollen is a highly suitable food for captive *E. larvarum* adults (Dindo *et al.* 2019). Therefore, similarly to other Diptera, *E. larvarum* can perform a double ecosystem service, although its role as pollinator has been unexplored.

As with bees, tachinid flies may be exposed to pesticides or other toxic chemicals either topically, during pesticide application, or via consumption of residue-containing food in treated flowers. To address this potential risk, we performed an acute contact toxicity study following, as much as possible, the standard protocol used for honey bees.

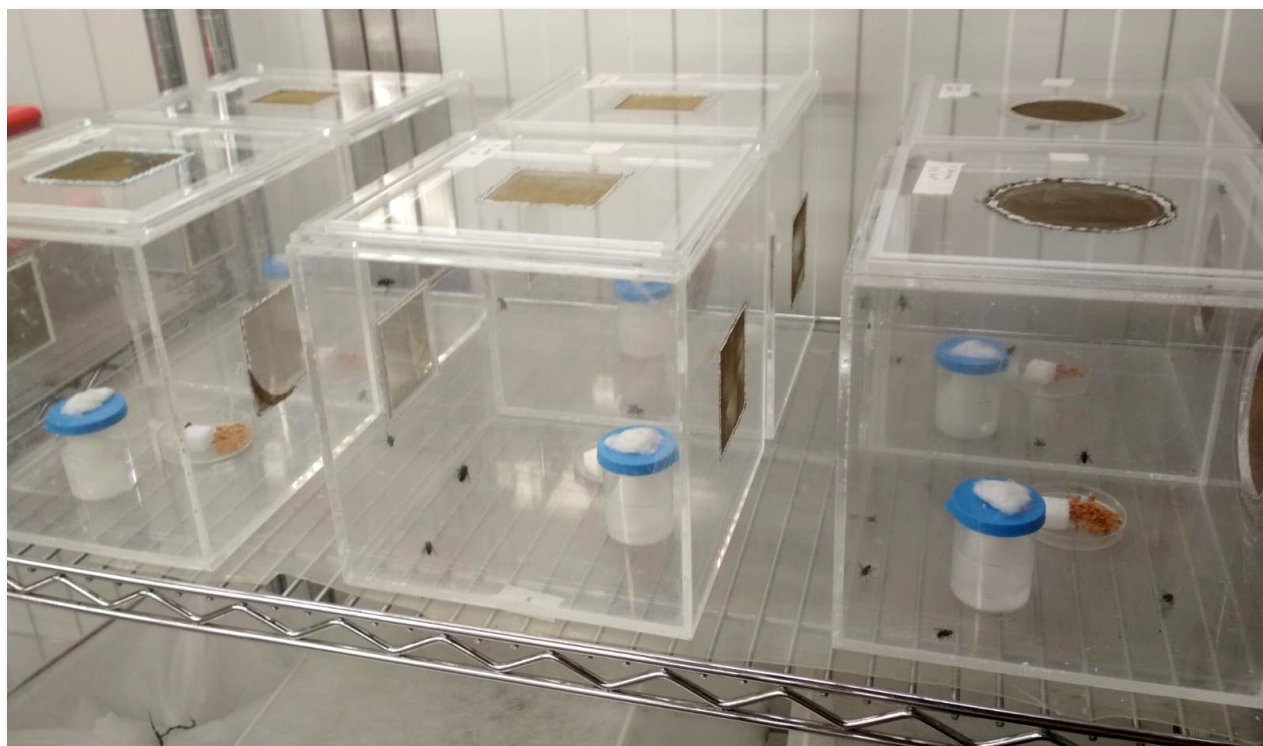


Figure 2. Test cages of treated adult mated females of *E. larvarum*. (Photo by C. Martins.)

Pesticide trials

Under laboratory conditions, we topically exposed adult mated females to a neonicotinoid pesticide. The tests were carried out with a population of *E. larvarum* reared and maintained at the DISTAL (University of Bologna) laboratory, using *Galleria mellonella* (L.) (Lepidoptera: Pyralidae) as factitious host (Figs. 2, 3). The experiment was conducted to obtain the median lethal dose (LD50) and to detect potential sub-lethal effects at the different tested doses. As sub-lethal endpoint, we assessed the oviposition rate on the host larva, which is a relevant ecological parameter. The results showed a lower sensitivity of *E. larvarum* to the tested pesticide compared to honey bees. However, sub-lethal effects on the fecundity of the females were observed at doses 13 times lower than the LD50 value. More complete details about the findings of this study will be published upon completion of my thesis.

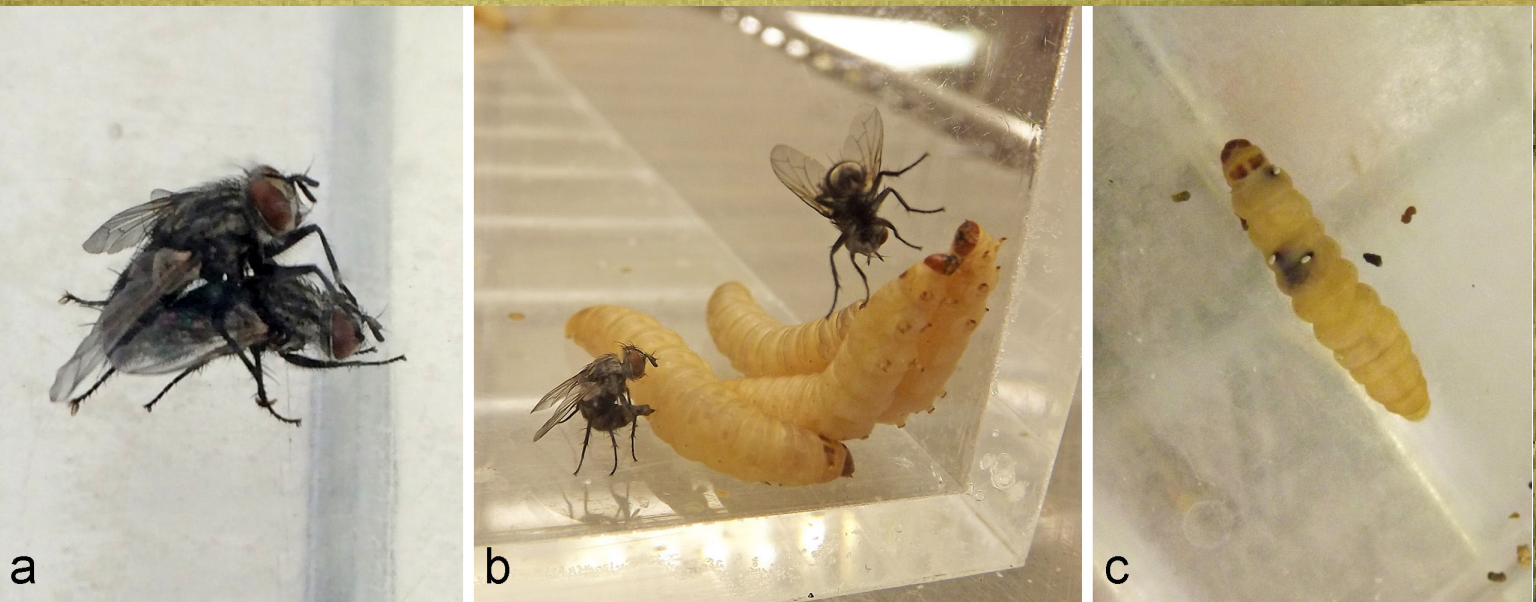


Figure 3. a. Mating activity of *E. larvarum*, before treatment with pesticide. b. Oviposition (exposure to *G. mellonella* after treatment with pesticide of the *E. larvarum* females). c. Eggs laid on *G. mellonella* caterpillars. (Photos by S. Francati.)

Final thoughts

To our knowledge, this was the first attempt to perform toxicological tests on a tachinid species under laboratory conditions. We believe this could be the first step to better understanding the potential risks that tachinid and other dipteran species face in intensive agro-ecosystems, as well as an opportunity to highlight their conservation value.

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Is that a
Chrysotachina?

And that one
also?

What is *Chrysotachina* B. & B. (Diptera: Tachinidae)?

by Juan Manuel Perilla López and John O. Stireman III

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It is well known that Tachinidae possess an extraordinary diversity not just in number of species, but also in terms of forms and life-history strategies. The family is notoriously difficult taxonomically for several reasons, some intrinsic and some due to the workers approach to them (O'Hara 2013). With over 1477 genera and 8592 species (O'Hara & Henderson 2020), it is difficult to gain a comprehensive knowledge of the family. The Tachinidae are characterized by recurrent rampant morphological homoplasies. This, along with the reliance of authors on a limited set of mainly chaetotaxic characters has led to historical taxonomic instability. Furthermore, there has been relatively little attention paid to immature stages and life histories in most taxonomic works. Despite the significant progress towards a classification that reflects the natural groupings within the family, there are still many limitations in the comprehension of the true relationships and diversity of tachinid taxa at all taxonomic levels (Wood & Zumbado 2010, O'Hara 2013).

The Polideini are a good example of the recurrent taxonomic difficulties faced in the study of this family, especially in the Neotropical Region. Revised only two decades ago, this tribe gathers together taxa previously assigned to nine different tribes (O'Hara 2002). Within the Polideini, the New World genus *Chrysotachina* Brauer & Bergenstamm is one of the most diverse genera. It has drawn the interest of several dipterists since its description in 1889 (e.g., Townsend 1912, 1919, 1931, Aldrich 1926, Curran 1939, Nunez *et al.* 2002, Nunez & Couri 2002), and despite significant progress over the last century in its classification, new taxonomic changes are needed to approach a definition of the genus that reflects its evolutionary history.

Here we provide an introduction to the diversity of the *Chrysotachina sensu lato* lineage and briefly discuss several aspects of the morphology and life history of the group. Our goal is also to use *Chrysotachina* as an example to highlight the importance of incorporating other characteristics complementary to adult morphology into tachinid systematics, such as immature stages, internal structures and natural history traits to find the “signal through the noise” (Cerretti *et al.* 2014) and better understand the evolutionary relationships of Tachinidae.

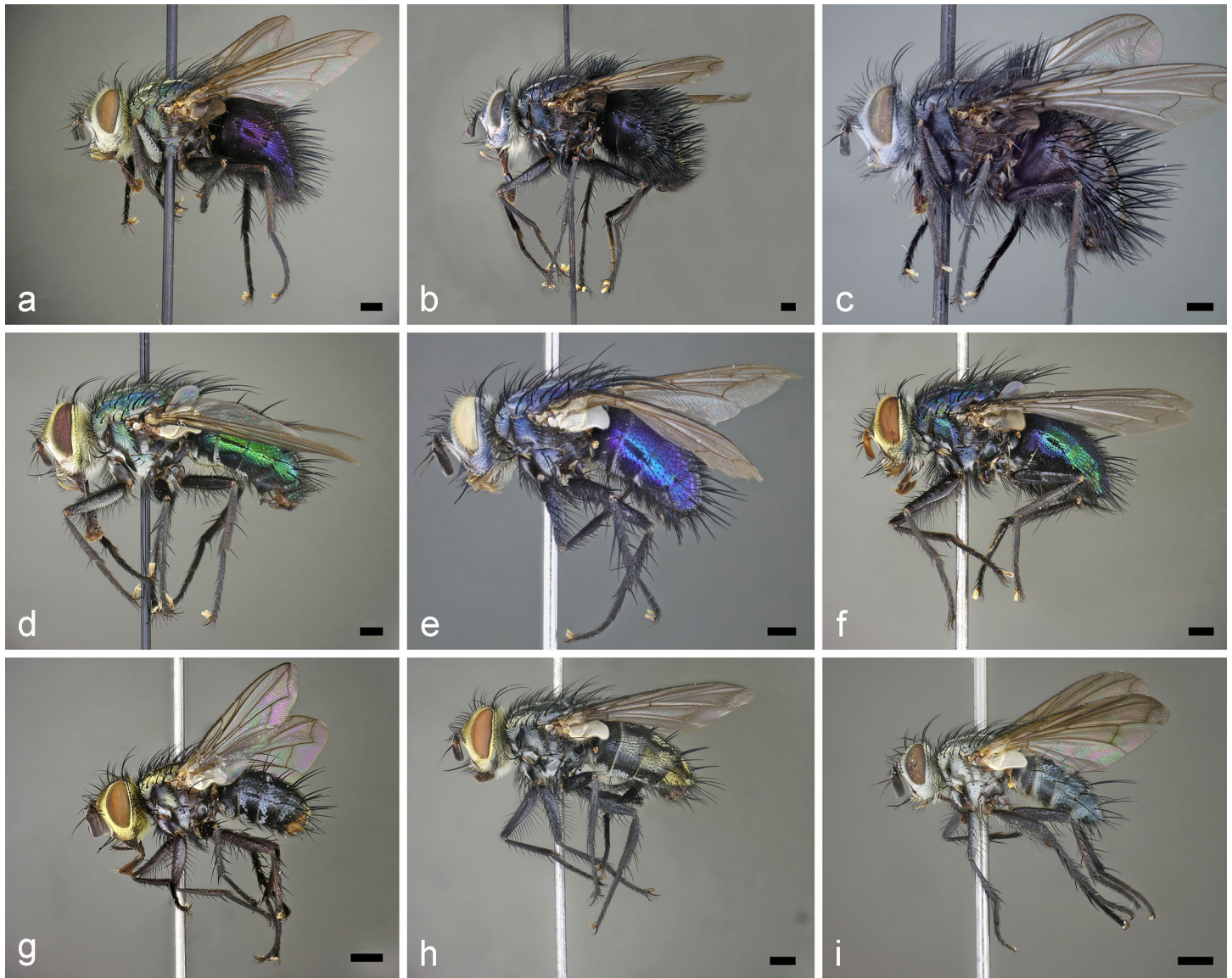


Figure 1. A small sample of representative species of *Chrysotachina sensu lato*: **a.** *Chlorohystricia reinwardtii* Wiedemann. **b.** *Chrysotachina s.l.* ("*Chlorohystricia*") n. sp. 1. **c.** *Chrysotachina s.l.* ("*Chlorohystricia*") n. sp. 6b. **d.** *Chrysotachina* n. sp. ald2. **e.** *Chrysotachina* n. sp. ery3. **f.** *Chrysotachina* n. sp. tow2. **g.** *Chrysotachina* n. sp. C3. **h.** *Chrysotachina* n. sp. W10. **i.** *Chrysotachina* n. sp. 49. (Scale bars = 1.0 mm.)

History of *Chrysotachina* classification

Brauer & Bergenstamm (1889) made a brief morphological description of *Chrysotachina* based on a comparison with *Gymnocheta* R.-D., a genus containing species with metallic green body color, which is distantly related and belongs to the tribe Ernestiini. Aldrich (1926, 1928) suggested a close relationship among genera of Tachinidae with metallic green color that are distributed around the world, grouping *Chrysotachina* with the ernestiine genera *Gymnocheta*, *Chrysosomopsis* Tnsd. and *Janthinomyia* B. & B.

Early work on *Chrysotachina* recognized only species with green or bluish metallic color as belonging to the genus (Townsend 1912, 1919, 1931, Aldrich 1926, Curran 1939), whereas non-metallic colored *Chrysotachina*-like species were included in other genera (e.g., Curran 1927, Aldrich 1932, Reinhard 1935). Later in the 20th Century, Guimarães (1971) included 13 species in *Chrysotachina* in his catalogue of the *Tachinidae of America South of the United States*. Guimarães included the strikingly similar non-metallic species *C. ornata* (Tnsd.) and *C. subcyanea* (v.d. Wulp) in the genus. Nunez & Couri (2002) later redescribed seven metallic-colored South American species of *Chrysotachina* and Nunez *et al.* (2002) added six new metallic-colored Neotropical species to the genus.

It was not until O'Hara's (2002) work on the delimitation of the Polideini and revision of Polideini of America north of Mexico that the boundaries of the genus were expanded to more of the non-metallic forms. Considering the similarities in the male terminalia and external morphology, O'Hara assigned to *Chrysotachina* all of the non-metallic species previously comprising the genera *Exoristopsis* Tnsd., *Helioplugia* Tnsd., *Mericina* Curran and *Neorigone* Tnsd., and described three new species for North America. In the same work, a morphological phylogenetic analysis of the Nearctic Polideini suggested that *Chrysotachina* may have affinities with *Chlorohystricia* Tnsd. (Fig. 1a–c) and *Hystricia* Macq., and the three genera were treated as the *Hystricia* genus group. No synapomorphies were found for *Chrysotachina*, raising doubts about the monophyly of this genus. However, the genus concept was maintained given the uniform appearance of the species. The cladistic analysis suggested that *Chlorohystricia* and *Hystricia* were sister taxa, a relationship supported by the presence of a globose abdomen and numerous discal setae on tergite 3. Historically, the genus *Chlorohystricia* has not received as much attention as *Chrysotachina*, perhaps because it contains only three described species. However, this limited number of species does not exclude it from the complicated taxonomic issues common in tachinid taxa (see O'Hara 2002: 83–84).

Is *Chrysotachina* monophyletic?

Defining *Chrysotachina*, following its current concept, through morphological methods based on the examination of adult specimens seems to be a task difficult to achieve. As currently defined, the genus comprises a group of remarkably similar but at the same time distinct and disjunct non-metallic (Fig. 1g–i) to metallic forms (Fig. 1d–f). Its species are characterized by the following combination of external characters: hairy eyes, bare parafacial, gena well-developed (about 0.2–0.25X head height), protruded lower facial margin, supra-alar row with 2 strong and subequal setae (in addition to other setae), metathoracic spiracle with dense and interwoven setae, wings usually hyaline or slightly infuscated, and in males proclinate orbitals are absent. In addition, they share the following combination of male terminalia characters: sternite 5 narrow and shallow, epandrium higher than long and pregonite long and curved. However, these are traits shared with other Polideini taxa (i.e., *Ecuadorana* Tnsd., *Spilochaetosoma* Smith, some *Hystricia*, and other undescribed Neotropical genera). Despite our detailed morphological examination, synapomorphies seem to be elusive in *Chrysotachina*. Some traits that are frequently used for the recognition of other tachinid genera are variable in *Chrysotachina*. For example, in *Chrysotachina* the inner vertical setae are crossed or parallel, in males the outer vertical setae are not differentiated to well-developed, palpi are usually yellow but in a few species are completely dark (almost black), prosternum is haired or bare, abdominal tergites 3 and 4 usually each bear one pair of median discal setae (but in some species these setae are missing), and in the wing, vein R_1 is usually devoid of setae but can be partially or completely haired.

Despite the absence of synapomorphies in the adults of *Chrysotachina*, JMPL's preliminary data obtained through next-generation sequencing of ultraconserved elements (UCEs), life-history traits, and previously unexplored morphological characters support the monophyly of an expanded *Chrysotachina*.

Redefining the genus *Chrysotachina*

The phylogenetic reconstruction of the Polideini using UCEs included only six representatives of *Chrysotachina sensu stricto*, and two representatives each of *Chlorohystricia* and *Hystricia*. However, this small representation provided a foundation for the understanding of the relationships, generic limits and diversity of these polideines. Preliminary results of the reconstructed phylogeny (Fig. 2) suggest that: a) *Chlorohystricia* species arose within the genus *Chrysotachina*, meaning that they are indeed larger and spiny *Chrysotachina*, b) the large spiny body form of *Chlorohystricia* species has arisen at least twice independently, and c) non-metallic forms are sister to the more derived metallic species. Although such results for *Chlorohystricia* species might seem shocking at first, a close relationship between these two genera was previously suggested by O'Hara (2002). However, our phylogenomic analysis did not reconstruct *Hystricia* as sister of these two genera as suggested by O'Hara. Nevertheless, results of these two phylogenetic analyses cannot be directly compared given the geographic focus of O'Hara's (2002) work on the Nearctic Polideini. These relationships uncovered by our phylogenetic analysis represent remarkable discoveries considering the historical assumption that spiky tachinids are closely related, and it is just one more example of the recurrent evolutionary convergence present across the family.



Figure 2. Left: A phylogeny of *Chrysotachina s.l.* based on ultraconserved elements. Species names in green indicate their metallic color. Right: Lateral and ventrolateral views of the abdomens of large and bristly *Chrysotachina s.l.* species included in the phylogenetic analysis: **a,b.** *Chrysotachina s.l.* ("*Chlorohystricia*") n. sp. 2, a species with sexual patches on abdominal tergites 4 and 5. **c,d.** *Chrysotachina s.l.* ("*Chlorohystricia*") n. sp. 6, a species without sexual patches.

Inclusion of *Chlorohystricia* in the genus *Chrysotachina* will reduce even more the possibility of finding synapomorphies in the adult stage to support the monophyly of a broadened *Chrysotachina*. However, despite this broader array of forms, external morphological characters and male terminalia may provide hints about the phylogenetic position of independent lineages of *Chlorohystricia* and their relationships with allied *Chrysotachina s.s.* species. We will use the term *Chrysotachina sensu lato* below for the grouping of *Chlorohystricia* and *Chrysotachina*. We are not formally synonymizing these two names here.

As currently understood, *Chrysotachina s.s.* and *Chlorohystricia* comprise 29 described species (5 Nearctic, 24 Neotropical) and 3 described species (1 Nearctic/Neotropical, 2 Neotropical), respectively. However, this is only a fraction of the true diversity of *Chrysotachina s.l.* During the examination of miscellaneous Polideini material borrowed from several collections for JMPL's Ph.D. dissertation project, over 70 undescribed Neotropical species of *Chrysotachina s.l.* were found. Examining this overwhelming, but not surprising, diversity of forms of *Chrysotachina s.l.* has allowed us to obtain a more complete picture of the diversity of this clade, and to explore traits that could be used to identify patterns of diversification. However, we have no doubts that the number of species in this genus is significantly greater than the number we have seen.

Sexual patches in *Chrysotachina s.l.*

Several authors have recorded the presence of sexual patches in descriptions of metallic-colored *Chrysotachina s.s.* species. However, limited attention has been given to these structures due to their uniqueness within the tribe. These specialized patches are exclusively found on male individuals, and they are characterized by abdominal areas with patches of setae and microtrichia that differ in morphology from the surrounding setae and are thought to have a secretory function (Cerretti *et al.* 2015). In *Chrysotachina s.l.* sexual patches are located on the ventrolateral surface of tergite 4 (T4) and tergite 5 (T5) or only on T5. Absence or presence of the sexual patches and their anatomical form, along with the male terminalia, seem to be the key to understanding the relationships among species of *Chrysotachina s.l.* with disparate body habitus.

Although sexual patches have been recorded only for some metallic species of *Chrysotachina s.s.*, we have found them also in *Chlorohystricia*. They are also present in at least two non-metallic species of *Chrysotachina*. The development of sexual patches across metallic, non-metallic and large bristly species of *Chrysotachina s.l.*, suggests that these organs may have developed independently multiple times across this lineage. Close examination of sexual patches across *Chrysotachina s.l.* reveals that these structures vary widely in the degree of area covered, lengths and densities of microtrichia and setae, degrees of punctuations at the base of the setae, and the presence of longitudinal grooves on the cuticle. Variation in the composition of these traits seems to form a continuous gradient, making it difficult to split these structures into well-defined types. However, patterns in the structure of these sexual patches seem to be conserved among closely related taxa. We have characterized four main configurations of abdominal tergites 4 and 5 in *Chrysotachina s.l.*: a) sexual patch absent (Figs. 2c,d, and 3a), b) sexual patch with strong punctures at the base of setae and grooves on the cuticle on T5 (Fig. 3b), c) sexual patch with strong punctures at the base of setae and grooves on the cuticle on T4 and T5 (Fig. 3c,d), and d) sexual patch without strong punctures at the base of setae and often with conspicuous microtrichia on T4 and T5 or only on T5 (Fig. 2a,b, and 3e–h). This last category includes the presence of a sexual patch on T4 and T5 or only on T5 since in several species there is only a slight difference in the density of setae present in the sexual patch or there is no clear definition of the sexual patch on T4. This is a temporary categorization that seems not to be exclusive to a particular group of species, but in combination with male terminalia traits it may help to cluster allied species. In considering these proposed categories for the sexual patches, it is not surprising that these organs had not been reported for *Chlorohystricia* species before, since the three described *Chlorohystricia* species present the last configuration of sexual patch, and the more setose and hairier body habitus of these species masks their presence. Sexual patches of some large-bodied *Chrysotachina s.l.* that are not evident at first glance can be recognized by the presence of slightly longer setae and a different angle orientation compared with the setae in the surrounding area. In addition, they are indicated by the presence of microtrichia covering the cuticle, albeit dense setation might interfere with their observation.

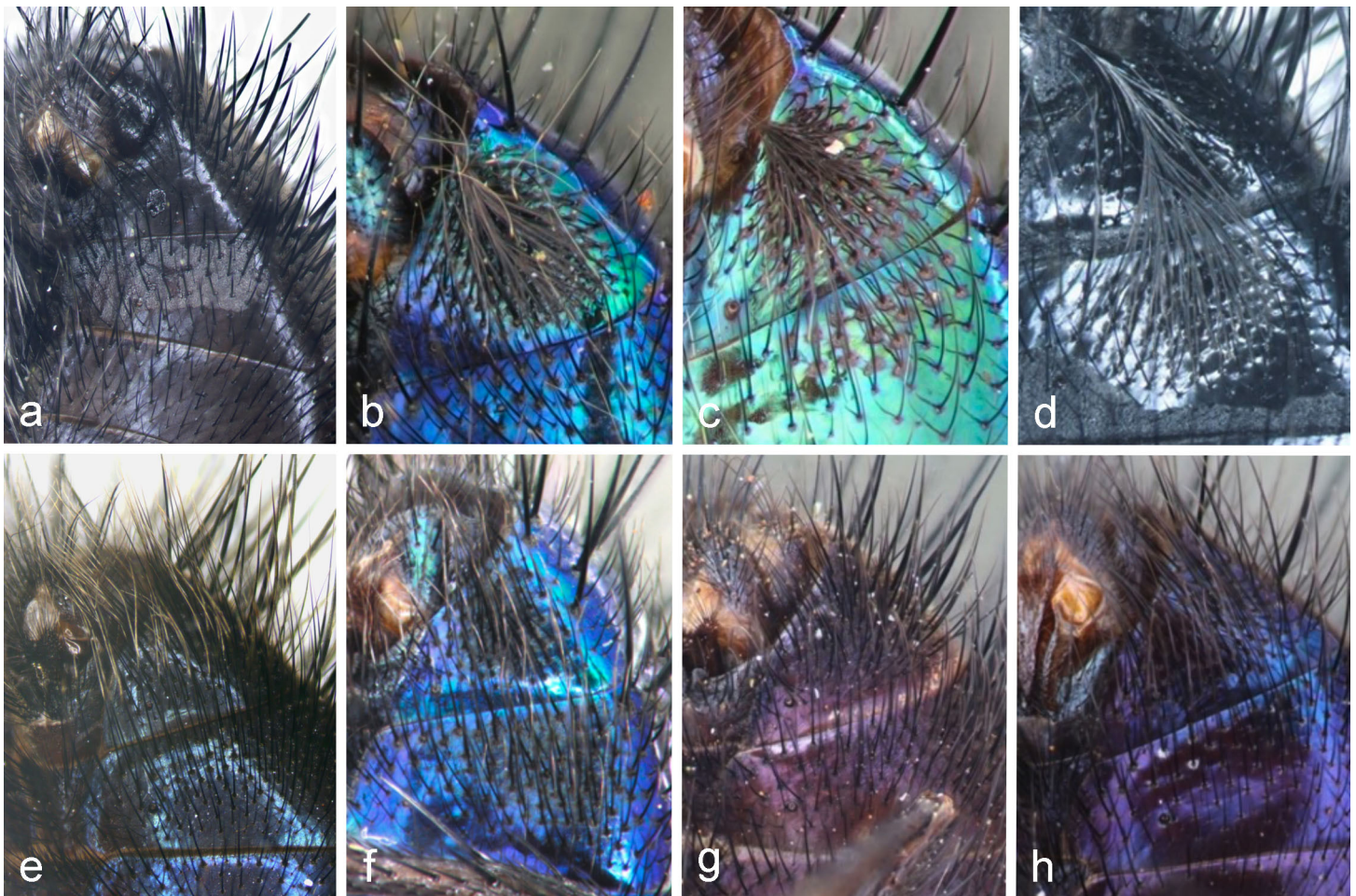


Figure 3. Ventrolateral view of male abdomens. **a.** Sexual patch absent, *Chrysotachina s.l.* n. sp. 8. **b.** Sexual patch with strong punctures at the base of setae and grooves on the cuticle on T5, *Chrysotachina willistoni* Curran. **c,d.** Sexual patch with strong punctures at the base of setae and grooves on the cuticle on T4 and T5. **c.** *Chrysotachina braueri* Tnsd. **d.** *Chrysotachina s.l.* n. sp. 1G. **e–h.** Sexual patch without strong punctures at the base of setae and often with conspicuous microtrichia on T4 and T5 or only on T5. **e.** *Chrysotachina s.l.* n. sp. 1b. **f.** *Chrysotachina* n. sp. J39, microtrichia conspicuous. **g.** *Chrysotachina s.l.* n. sp. rWA. **h.** *Chlorohystricia reinwardtii* Wiedemann.

In addition to the three described species of *Chlorohystricia*, we have found about 15 large and spiny undescribed species of *Chrysotachina s.l.* displaying each of the four configurations on T4 and T5. With the combination of these sexual patch characteristics and male terminalia traits we have clustered these large-bodied species into what appears to be six groups of closely related species. This clustering of large-bodied *Chrysotachina s.l.* allows us to associate these groups with closely related but distinct-looking and smaller species.

Finding the signal: puparium traits and host associations

Despite the absence of synapomorphies to define *Chrysotachina s.l.*, we have found that some traits of the posterior spiracle discs of the puparium and host-associations provide insight into relationships within this clade. Unfortunately, for most Neotropical tachinids there is virtually no information about their immature stages nor their host associations. However, data obtained mostly from the biodiversity inventory programs at the Area de Conservación Guanacaste (Costa Rica) and Yanayacu Biological Station and Center for Creative Studies (Ecuador) are providing useful information to help improve comprehension of these polideines.

Comparison of the posterior spiracular discs of the puparia of around 20 species of *Chrysotachina s.l.* and several representatives of other polideine genera with robust and hairy bodies (i.e., *Hystricia*, *Pseudobombyliomyia* Tnsd. and *Eucheirophaga* James) shows that *Chrysotachina s.l.* possess sinuous spiracular slits (Fig. 4a,b,d,e) in contrast to the straight or slightly curved slits present in the other genera (Fig. 4c,f). Additionally, we have found two states in these posterior spiracular discs of the puparia of *Chrysotachina s.l.* that concur with the relationships found in the UCE-based phylogenetic analysis. The more basal lineages of *Chrysotachina s.l.* (i.e., some of the non-metallic forms) possess a puparium that is posteriorly more or less evenly rounded on upper and lower surfaces with the posterior spiracular discs scarcely raised above the surface of the puparium. On the other hand, other species in this clade have the posterior spiracular discs larger and medially slightly sunken. An analysis of the structure of the posterior spiracular discs of the puparia of polideines could potentially support the presence of the sinuous spiracular slits in the posterior spiracular discs as a synapomorphy for the entire clade of *Chrysotachina s.l.*, and variation in it may help to resolve relationships of lineages within *Chrysotachina s.l.*

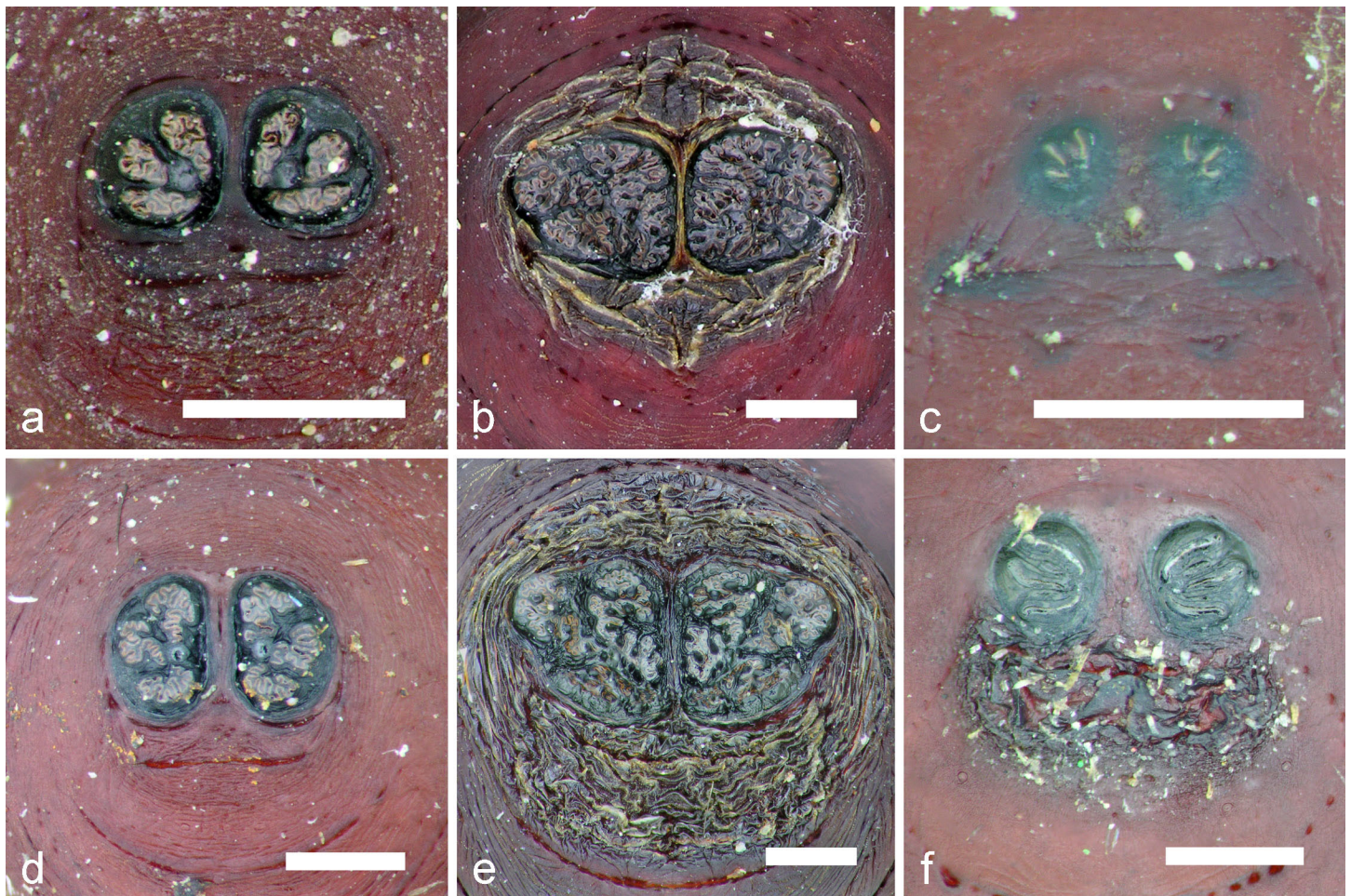


Figure 4. Posterior spiracular discs of the puparium: **a.** *Chrysotachina urichi* (Aldrich). **b.** *Chrysotachina* n. sp. subc2. **c.** *Pseudobombyliomyia* n. sp. YY. **d.** *Chrysotachina* n. sp. 5. **e.** *Chrysotachina s.l.* n. sp. CH3. **f.** *Hystricia* sp. 6. (Scale bars = 1.0 mm.)

Host associations of *Chrysotachina s.l.* can also provide evidence about their evolutionary relationships. During the initial stages of JMPL's work on the Polideini, we considered the undescribed species *Chrysotachina* n. sp. 49 (Fig. 1i) and three allied taxa as a potential separate genus sister to *Chrysotachina s.l.* *Chrysotachina* n. sp. 49 and closely related species have slender body habitus and reduced and lightened setae ventrally. Additionally, *Chrysotachina* n. sp. 49 parasitizes *Pedaliodes montagna* Adams & Bernard (Lep.: Nymphalidae), a free-living caterpillar whereas most *Chrysotachina s.l.* attack shelter-building caterpillars of the families HesperIIDae and Mimallonidae.

However, detailed examination of the host records available for *Chrysotachina s.l.* revealed a broader array of host associations. We found that at least one complex of undescribed species of *Chrysotachina s.s.* is associated with caterpillars belonging to five genera of metalmark butterflies (Riodinidae), a family known for the associations of its free-living caterpillars with ants, and at least three species of *Chrysotachina* have records of parasitizing free-living nymphalid caterpillars. Hence, although species of *Chrysotachina s.l.* are not strictly restricted to shelter-builder caterpillars, their hosts are restricted to these four above-mentioned families of Lepidoptera. Complementary to this, *Hystricia*, *Pseudobombyliomia* and *Eucheirophaga* are also caterpillar parasitoids, but they are not known to parasitize caterpillars in any of the families parasitized by *Chrysotachina s.l.* Furthermore, there is no overlap in the lepidopteran families parasitized by these three genera.

Notwithstanding the external heterogeneity of *Chrysotachina s.l.*, the examination of previously poorly-explored morphological traits, puparium characters and host associations, supports the monophyly of this disparate group of tachinids as well as the hypothesized independent evolution of lineages with large and bristly bodies. This raises the question of why several lineages of polideines have experienced this “hedge-hog fly syndrome”. The repeated evolution of robust, spiny bodies is an interesting pattern of morphological convergence present across several lineages of Tachinidae that have diversified extensively in the Andes. However, this pattern of diversification in tachinids remain largely unexplored.

The final steps in this study of *Chrysotachina s.l.* will include a redefinition of the genus and the description and/or redescription of its many species. Further observations and analyses are needed to better understand the evolutionary relationships within *Chrysotachina s.l.* A more detailed examination of sexual patches using SEM imaging is necessary to better understand their structure and variation across the genus, and new characters need to be found to add support for the multiple origins of the large and bristly *Chlorohystriicia*-like species.

Note to the reader: If you rear caterpillars from any of the lepidopteran families mentioned above, keep an eye open for parasitized specimens because you might be a happy witness to the emergence of a *Chrysotachina* specimen.

Acknowledgements

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Figure 1. Six-metre Malaise trap in author's yard on the edge of Ottawa, Canada, 28 September 2020. Insert: pinned and labelled tachinids (minus siphonines) caught in trap over nine days, 7–15 July 2020 (specimens turned on angle are interesting taxa or good for images).

How to make your tachinids *STAND OUT* IN A CROWD

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Dedicated to the memory of Monty Wood who taught me the importance of well prepared tachinid specimens.

Introduction

It is a cruel quirk of fate that the most fastidious of insect preparers tend not to be dipterists. They are more likely to be lepidopterists who arrange the wings of moths and butterflies with such precision that one side is a mirror image of the other, or coleopterists preparing specimens of the larger and more showy species. Yet these same collectors tend to treat flies as bycatch and only grudgingly stick pins through some of them for their dipterist colleagues. If a #3 pin is thrust through the centre of a 3mm long *Catharosia* sp. with the legs pinched up tight against the body, is anyone going to care? Sadly, variations of this practise are not uncommon even among some dipterists. How many times have I been brimming with excitement as I opened a box of specimens received in the mail, or pulled out a drawer in a foreign collection, only to gaze in horror at a multitude of ugly and misshapen specimens? It doesn't have to be like this.

Don't get me wrong, I am not advocating specialized spreading boards for flies or elaborate time-consuming methods. Just a little more care: relax your specimens, pin them, push up the wings, pull down the legs, straighten the head, and the basics are done. If your specimens are covered with moth scales then you can make them sparkling clean in a minute or two. If you are so inclined then male terminalia can be exposed for future study, legs that have dropped off can be saved for DNA barcoding, and delicate specimens can be frozen to allow them to dry slowly and not shrivel. These techniques will produce specimens ideal for taxonomic study. Characters that are useful for identification will be maximally exposed and specimens can be compared with less effort.

Early beginnings

In mid-1970s I took an introductory course on entomology taught by coleopterist Henry Howden at Carleton University in Ottawa. Our textbook was the new fourth edition of the beloved *An Introduction to the Study of Insects* (Borror *et al.* 1976). The chapter at the end of the book has 45 pages on collecting, preserving and studying insects. The methods reviewed there are still relevant today, but the same information is now readily available to all on the Internet.

My insect course at Carleton led to a summer job in the Diptera Unit of the Canadian National Collection of Insects (CNC) in 1977 and 1978. I also worked part-time for D. Monty Wood between the two summers, at Monty's personal expense. It was during this time that I learned why properly prepared tachinid specimens are so important. I received some instruction on this topic from Monty but I could also see for myself his meticulously prepared specimens throughout the Diptera collection and especially in the Tachinidae cabinets (e.g., Fig. 11). I believe I can say without hyperbole that Monty's specimens are unrivaled in collections the world over. In the CNC, I can generally spot his specimens immediately, before looking at the labels. It is this gold standard that I try to emulate with my own specimens.

“I believe I can say without hyperbole that Monty's specimens are unrivaled in collections the world over.”

Start fresh

The key to perfect specimens is to start with fresh and relaxed material. Last summer I worked a little harder on this step of the process. Stuck at home due to the COVID-19 pandemic, I ran a 6-metre Malaise trap in my backyard on the edge of Ottawa from April to November to survey local tachinids. This was a continuation of similar efforts in 2016 and 2017 but this time I could empty the catch two or three times a day (e.g., Fig. 2), seven days a week, for the entire period. The trap was run “dry” with a 3cm x 6cm section of Ortho® Home Defense® Max™ No-Pest® Insecticide Strip as the killing agent. The trap head was removed each evening around dusk and replaced early the next morning to cut down on the number of moths mixing with the other insects and coating them with scales.

My former method for holding dead tachinids until pinning time, whether in the field or at home, had been to place them in a container with soft leaves until evening or the next day, often in a cooler or refrigerator. Tachinids tend to stiffen when they die and a little time and moisture will relax them. Although this method is commonly used and produces good results, most of the time the specimens are a little shy of perfectly relaxed and their legs and head often spring back a bit from where they are put.



Figures 2–5. 2. Malaise trap sample from 4 July 2020 with Canadian quarter for reference. 3. Relaxing container with wet sphagnum moss for moisture and tissue-covered tray for specimens (26 July 2020). 4. Fresh specimen of *Ptilodexia* sp. coated with hairs and scales. 5. Same specimen after bath and arrangement of wings and legs.

I tried a few variations of the above last summer to increase the moisture content and more fully relax specimens. The method that worked best is shown in Fig. 3 and was used for all the specimens that illustrate this article (except Fig. 11). The bottom of a plastic container (my container having a rim diameter of 12cm and depth of 6cm) was lined with sphagnum moss (depth of 2cm) and generously watered. A small concave dish with a tissue liner was placed on top of the moss and held each day’s catch of tachinids. The container was covered with a tight-fitting lid, not refrigerated, and specimens were pinned sometime the next day. The tissue liner was replaced periodically but the same sphagnum moss was used continuously until the end of the season, with just the occasional replenishment of water. Sphagnum moss has anti-fungal properties and, as I learned, is quite resilient because I had no issues with it in a plastic container for six months.

Pinning

Just a quick review here of pinning fundamentals for those getting started. There is a lot of personal choice involved in whether to use stainless steel or black enameled pins (I prefer the latter) and what pin sizes to use (I like #00 to #2 and double-mounted minutens for tiny specimens in the 4mm range or smaller). The standard method for pinning is to hold a tachinid vertically between the thumb and forefinger of one hand and with the other hand push a pin through the thorax to the right of the midline and in front of the wing with an exit hole in front of the mid coxa. Wearing a headband magnifier (e.g., OptiVISOR 2.75X) is a great help for accurate pinning. Position the specimen low enough that it will not be touched when holding the pin and high enough that it will not interfere with a label below. Label height is a personal preference; I like a height of 15–16mm from the pin tip and make my own labelling blocks with this depth (Fig. 6; hole drilled with a 1/32" drill bit).



Figure 6. Labelling block.

Specimens mounted from alcohol or too brittle for pinning can be glued to the side of a pin. This is the method of choice for some dipterists who work with small to medium-sized specimens. There is much debate about what type of glue to use and this topic is nicely reviewed in the first link under Other Resources below. A favourite for decades in the Diptera Unit of CNC is shellac glue but you have to make it yourself and experiment with consistency (Walther 1997). It is also slow to dry and thus best suited for mounting smaller specimens.

Some further techniques and comments pertaining to the mounting of flies are given in:

Mounting tachinids from ethanol (O'Hara 1994)

Minuten mounts for micro-Diptera (Sabrosky & Mathis 1997)

Mounting methods for small Diptera (Wheeler 1997)

It's all in the details

If you are not particular about the look of your specimens, read no further...

For those still reading I will outline what you can do in less than a minute if you are starting with a pinned and well relaxed tachinid. This work is best done under a stereoscopic microscope except for the first step involving the wings.

Wings. If these are not already in the upright position, place tips of forceps under them and gently lift upwards, squeezing gently inwards on the thorax to help “lock” them in place when they reach the “up” position. This may have to be repeated a few times until they stay in place. If the wings continue to drop down then they can be braced upright later on a pinning board (Fig. 7).

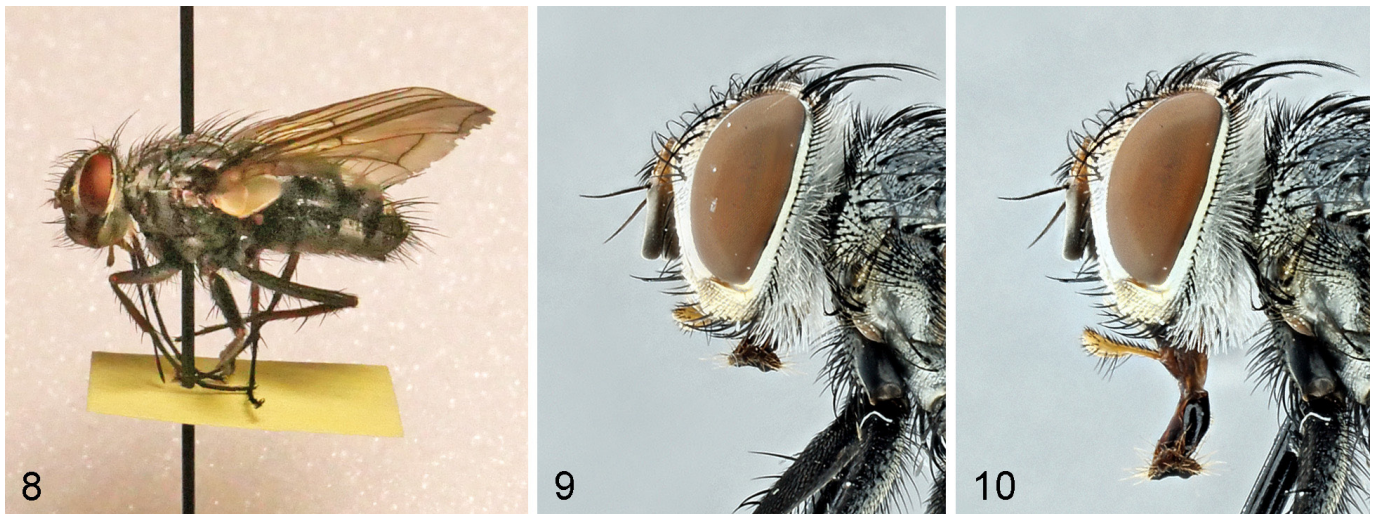


Figure 7. Specimens on Styrofoam pinning board showing three with wings braced in upright position and two with legs saved on sticky portions of yellow Post-it® notes.

Legs. These frequently get pinched up against the thorax when the specimen is pinned, obscuring diagnostic features on the side of the thorax. Pull each leg downwards and disentangle legs if necessary. They will partially spring back if not thoroughly relaxed, but this is often enough to see the sides of the thorax. Legs that are particularly long, like those of many Dexiini, can be pushed upwards by a piece of paper and arranged in that position (Fig. 8). The paper is removed when the locality label is attached.

Head. This rotates at 90° to the axis of the body, meaning that in death the vertex is often to the right or left of the centreline. Place one tip of a forceps against the proboscis and rotate the head into alignment with the thorax. As with legs, the head will not stay in its new position unless its flexible connection to the thorax is fully relaxed.

Proboscis. Sometimes the base of the proboscis is retracted into the oral cavity and its full length cannot be seen and the palps are partly obscured (Fig. 9). The palps will generally pop into view with a gentle pull on the proboscis (Fig. 10). I just started doing this last summer.



Figures 8–10. 8. *Mochlosoma* sp. with long legs held in bent position with piece of paper. 9. *Belvosia unifasciata* (R.-D.) with proboscis retracted and palps partially obscured. 10. Same specimen with proboscis and palps exposed.

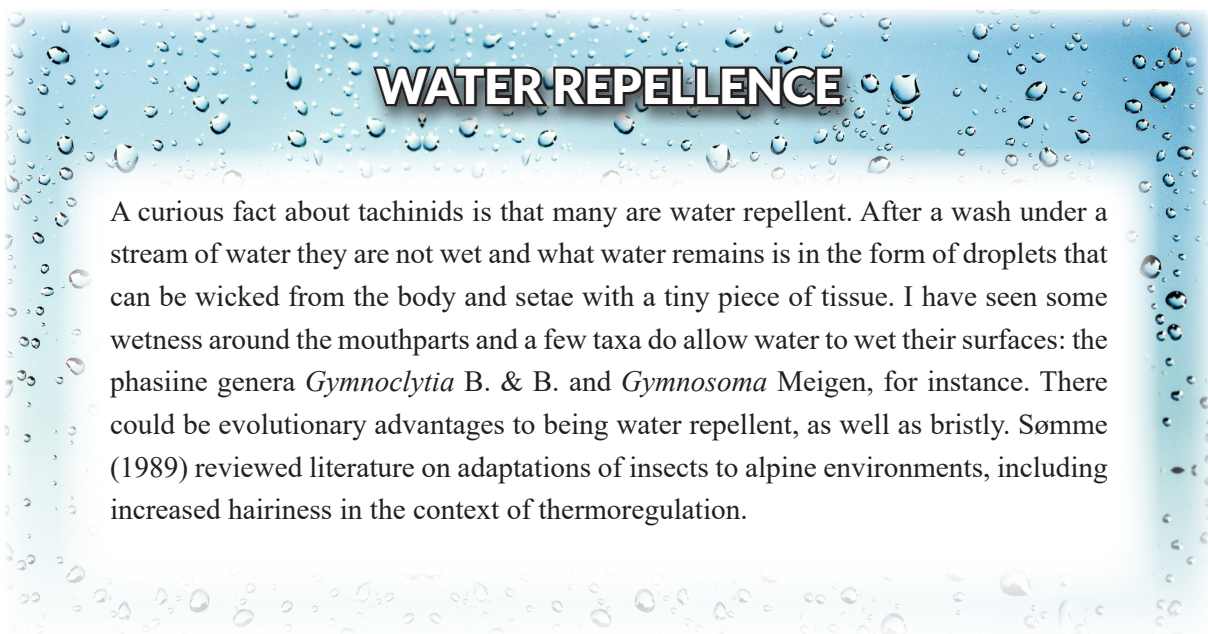
Setae. Some of the larger setae on the head or dorsum of the abdomen (not often on the thorax) are sometimes askew. This is a small matter but they will “click” into position if pushed in the right direction. Do not try this with dried specimens as the setae will break off. Setae on relaxed specimens have a natural position to which they can be returned.

Dirt. The foregoing can be done quickly but removing debris such as pollen, broken setae, lepidopteran scales, etc., takes more time. Forceps are not the right tool for such delicate work. I recommend making a simple “cleaning tool” that bends when it touches setae and body parts. Take a short length of 3mm dowelling (sold at dollar stores in the arts and crafts section) and drill a hole in one end with a 1/32" drill bit, or use an insect pin to create a hole. Insert a 1cm length of hair from a fine paint brush and glue in place (same method as used for terminalia hooks shown Fig. 11).

Pinning boards. Individual preference will dictate where specimens are placed after pinning and before labelling. I like to arrange them on pinning boards made of Styrofoam with a wood backing to add weight. These boards are also handy for labelled specimens if the specimens are not going into unit trays in a collection right away (see insert in Fig. 1). Delicate specimens that are apt to shrivel as they air-dry can be pinned and kept in a freezer for several weeks to slowly “freeze-dry” with less shrivelling.

Bath time

A little-known fact is that most fresh tachinids can be washed. I learned this technique from Bruce Cooper, a long-time technician in the CNC Diptera Unit. This is effective in removing most hairs and lepidopteran scales that are resting on a specimen, as is often the case with fresh material left overnight in a Malaise trap. A *Ptilodexia* sp. pinned from such a moth-filled Malaise sample is shown in Fig. 4. It was held under a gentle stream of water from a tap for about 10 seconds while being rotated to bring all surfaces in contact with the flow of water. The wings were then set in the upright position, some remaining debris cleaned off under the microscope, and legs adjusted (Fig. 5).



Male terminalia

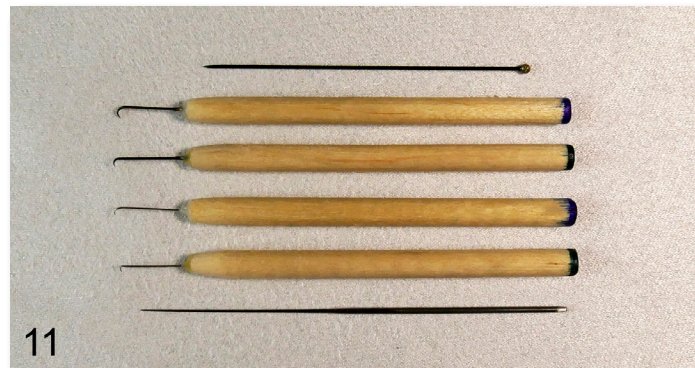
The terminalia of male tachinids represent a wealth of valuable characters for both identification and phylogenetic inferences (e.g., Cerretti *et al.* 2014). A method for removing male terminalia from dried specimens is given further below. Here I will describe quick methods for removing or exposing the terminalia of male tachinids.

Removing male terminalia from a fresh specimen is easy and can take less than 30 seconds. First, make a terminalia hook from a minuten or small-diameter pin (#00, #0). To do this, grab the tip of the minuten or pin with strong forceps and twist with a semi-circular motion to produce a nicely curved hook. Make several using different pin sizes for working with small to large tachinids. I like to embed these hooks in 3mm dowelling (more about the dowelling above under Dirt) (Fig. 11).

Under a microscope, hold the pinned tachinid in one hand and terminalia hook in the other. Turn the fly over so the underside of the abdomen is visible. Insert the hook between the 4th and 5th abdominal sternites, hook the edge of sternite 5, pull out the genital capsule, and wrap the removed parts around the pin below the specimen. Some of the digestive tract will usually come out with the genital capsule and it can be wrapped around the pin to adhere the terminalia to the pin when it dries. If you want to be tidy then shove any remnants of the digestive tract that are dangling from the abdomen back into the cavity of tergite 5. The terminalia can be removed from the pin using very warm water or an insect relaxing fluid. I made a low-quality movie years ago showing the procedure for removing the male terminalia of a tachinid that is now available here:

<http://www.nadsdiptera.org/Tach/WorldTachs/TTimes/tachinid.mov>

Gluing terminalia below specimens will facilitate their examination later on if they are needed for closer study or a taxonomic revision. The downside is that parts of the terminalia are usually hard to see when stuck to a pin and must be removed and cleared for study, then stored in glycerin. (The steps are the same as given in the next section except the terminalia are already separated from the abdomen.) Some species can be distinguished by differences in the male surstylus and cerci and these parts can be exposed on fresh specimens without the need to remove the terminalia. Insert the tip of a terminalia hook into the gap posterior to sternite 5 and twist to catch on to or near the base of a surstylus. Gentle twists and pulls will usually free the surstyli and cerci enough that they can be viewed *in situ* (Fig. 12). There can be greater difficulty in doing this with some taxa than others and there is some risk of damage when the genital parts do not cooperate.



Figures 11–12. 11. Four terminalia hooks, a tungsten steel needle, and a #2 insect pin for scale. 12. A male of *Eribella exilis* (Coquillett) with terminalia exposed (TachImage-00576). This specimen was collected by Monty Wood and its preparation is typical of Monty's style. It was caught at Duncan Lake, Masham Township, Quebec, on 17 July 1997 (Monty's cottage).

Dissecting male terminalia from pinned specimens

I gave a description of how to prepare male terminalia for taxonomic study in a revision of the Polideini (O'Hara 2002). That publication is not easy to obtain online so I will quote the directions here (pp. 13–14):

“The abdomen of a male was carefully removed at its junction with the thorax and placed in 10% hot (not boiling) NaOH^{1,2} until slightly flexible (about 15 minutes). During this time, the abdomen was periodically and gently squeezed against the side of the container to remove trapped air bubbles. The abdomen was then placed in a microdish with 50% acetic acid, held against the bottom of the dish with a curved and blunt pin (inserted into the opening at the base of the abdomen) and the terminalia (sternite 5 to cerci) carefully cut out using a fine tungsten needle³. The abdomen was taken through 70% and 95% EtOH washes (10–20 minutes each) and a final xylene or toluene wash (30 minutes) before being reattached to the thorax in its original position with shellac glue. If the abdomen was placed in position a few seconds before it was completely dry, then the residual xylene would thin the glue momentarily and a smooth bond would often form. Air-drying an abdomen directly from EtOH is not recommended because it almost invariably results in severe curling of the abdomen. After the male terminalia were removed from an abdomen, additional clearing was necessary so that the structures could be separated further for study. Two methods were used for this purpose. The first method, as described by O'Hara (1983), involved additional clearing of the terminalia in 10% NaOH, neutralization of the base using a 20% acetic acid wash, two EtOH washes (70% and 95%), and examination and storage of the terminalia in glycerine. The concentration of the acetic acid wash in the present study was raised to 50% from the 20% used by O'Hara (1983) because some of my dissections performed years ago for other studies have continued to clear in storage⁴.

I began using lactic acid⁵ for terminalic preparations late in the present study to alleviate overclearing of terminalia. Lactic acid has two advantages over NaOH for terminalic dissections: less danger of long-term over-clearing, and sclerotized structures are not cleared. The latter is particularly important for the study of the anterior sclerite of the distiphallus, which is easily over-cleared in normal NaOH preparations. Male terminalia, removed from the abdomen as explained above, were placed in ceramic dishes with 100% lactic acid and heated in a microwave oven for 10 seconds⁶ at a time until soft tissues had cleared sufficiently (generally 3–5 repetitions at intervals of about 5 minutes). The terminalia were then placed in a microdish of glycerine for further dissection. The terminalia proper were removed from sternite 5 and the ring sclerites. Often the dissection would end at this stage, but if further dissection was necessary for more detailed study or for illustrative purposes then the hypandrium (with pregonite, phallus and postgonite attached) was disarticulated from the epandrium and often the phallus would be separated from the hypandrial complex. Terminalic structures were either placed in special trays (O'Hara & McIntyre 1984) with fresh glycerine for periodic study and temporary storage or in microvials (pinned below adult specimens) with glycerine for permanent storage.”

A few notes pertaining to the above (see superscript numbers in text):

1. NaOH (sodium hydroxide) vs. KOH (potassium hydroxide). The latter is more commonly used as a clearing agent. The former is reportedly a little slower acting, which is good if you want to stop the clearing at a precise point, but I have no experience comparing the two.

2. Juan Manuel Perilla López (pers. comm.) has been experimenting with the contact lenses cleaner Ultrazyme™, a digestive enzyme, as an alternative to NaOH/KOH. He does not yet have a protocol for its use. Kanaar (1990) wrote about using Genitase, a proteolytic enzyme, for clearing terminalia.
3. A fine tungsten steel needle is shown in Fig. 11. It can be repeatedly sharpened using super fine emory sandpaper. It is preferable to a minuten or fine insect pin because the steel is stronger and the fine shaft is relatively inflexible. I found several at my place of work early in my career and still use them; I do not know if they can be purchased anymore.
4. Terminalia cleared with NaOH or KOH may continue to clear slowly over time (decades) despite washes of acetic acid that are meant to stop the process. Light-coloured terminalia seem more prone to clearing over the long term. I have a policy of not allowing terminalia of holotypes to be cleared with either chemical unless images or drawings of the terminalia will be published.
5. See Cumming (1992) for more about the use of lactic acid.
6. Microwave ovens are stronger now than when the above was written and heating terminalia in a small ceramic dish for 10 seconds in a modern microwave will likely cause the lactic acid to bubble and/or the terminalia to explode. It is best to experiment with shorter times and expendable terminalia. Another option, albeit slower, is to gently heat a dish of 10% NaOH or lactic acid on a coffee mug warmer. This is my method of choice now.

Save that leg

The legs of tachinids are prone to falling off no matter how gently they are touched during pinning. It is rare to pin 25 specimens without some legs left over. This is more common with the smaller and less robust specimens but no species is immune. I could not be bothered in the past to save these cast-off appendages but with the advent of molecular systematics and DNA barcoding there is more reason to keep them. I simply cut a small rectangle from the sticky end of a Post-it® note and press a leg against it, then pin this below the specimen (Fig. 7). There is not a firm attachment and the leg is easily removed for molecular use. So far I have not seen legs fall off on their own during normal handling but I would not recommend this method for permanent storage.

Comparing two specimens at once

At the beginning of this article I wrote: “Dedicated to the memory of Monty Wood who taught me the importance of well prepared tachinid specimens”. I want to explain what I mean by this. Flies of some families are fairly recognizable based on external features (e.g., Bombyliidae, Syrphidae) or male terminalic features (e.g., Sarcophagidae, Sphaeroceridae). Other families, like the Tachinidae, fall somewhere in the middle with some taxa exhibiting one type of difference or the other, rarely both types in the same taxon, or sometimes only subtle differences in any characters. *Lespesia* R.-D. and *Winthemia* R.-D., for example, fall into this last category. In closely related species, slight differences in the width of the parafacial, length of postpedicel, shades of silver or gold on the fronto-orbital plate, abdominal setation and colouration, are most easily compared between specimens that have all their parts similarly aligned. Differences can be so slight that looking at two specimens at once is the best way to compare them; one pin held below the label in one hand and the other pin held by the pin head with the other hand so the specimens can be moved back and forth in tandem. Trying to do this with badly pinned specimens with heads rotated at odd angles and legs all over the place is frustratingly difficult. DNA barcoding has been confirming that the slightest of differences can be species specific.

Other resources

Museum Specimen Preparation Guidelines.

There are a number of guides to preparing insects. This one is more detailed and comprehensive than most about labelling, pinning and gluing specimens (by Heraty, J., Yanega, D. & Triapitsyn, S., undated):

https://entmuseum.ucr.edu/specimen_preparation/index.html

Note: I agree with the authors that “anything bigger than about 15x7mm is getting too big” for an insect label but I differ with respect to font type, font size and maximum number of lines. They recommend “Times (or maybe New York)”, “4 point lettering” and “5 lines”. They note that letters can bleed together using Arial font. This is true, but I like the uniform thickness of Arial letters and I avoid letting them touch by selecting 105% kerning (this moves the letters apart slightly). With this kerning I can print at 3.5 point with letters sharp and not touching provided the printer is up to the task. I reduce line spacing to 95% to bring the lines closer together. With these adjustments I get 6 lines of data in the same 7mm width recommended by the authors (compared to their 5 lines). In recent years my 6th line is often a database number like “CNCxxxxxxx”. I prefer to have this number on the locality label rather than print a second label for it.

Entomological supplies.

There are suppliers around the world with specialized equipment for entomologists. In North America I recommend *BioQuip Products* (<https://www.bioquip.com>). They are knowledgeable, reliable and professional. Among other things I like their insect pins and label paper because both are high quality (the paper is 100% rag and acid-free, the gold standard for permanent labels). If you can locate the original sources you might save a little money but you also risk buying inferior materials.

Collecting methods for tachinids.

My review paper on the Tachinidae of Gila National Forest in New Mexico, USA (O’Hara 2012) was written for a general audience and briefly outlines common methods of collecting tachinids: handnetting, sweeping, sugaring, Malaise trapping, hilltopping and blacklighting.

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Differences vs. similarities:

Monty Wood's approach to tachinid classification (Diptera: Tachinidae)

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When I was growing up in a small town outside Ottawa in the 1950s and 60s, my view of the world was largely based on what I learned in school, what I saw on two channels of a black and black television set, and what I read in books. We did not have the means to travel much but we had relatives in Montreal and when the world fair opened in that city during the Centennial year of 1967, we visited them and spent a day at Expo 67. For me at the impressionable age of 14, seeing the world on display like that was quite a revelation.

I had no idea at the time that the dipterists at the Canadian National Collection of Insects (CNC) in Ottawa were similarly broadening their horizons in 1967. Their grand plan to coordinate a replacement to Curran's (1934) *Families and Genera of North American Diptera* had been approved the year before and they were hosting German phylogeneticist and dipterist Willi Hennig for three months to discuss phylogenetic principles and dipteran phylogeny. Those early discussions

laid the philosophical foundation for the *Manual of Nearctic Diptera* (MND) that developed into three volumes published sequentially in 1981, 1987 and 1989. Monty Wood (1989: v) noted in the Preface to the final volume: "This third volume ... explains the rationale underlying the classification that was adopted in Volumes 1 and 2, using the methodology of phylogenetic systematics developed by Hennig".



Figure 1. The *Manual of Nearctic Diptera* coordinators with German dipterist Willi Hennig during his visit to the Diptera Unit in the fall of 1967. Back, left to right: Frank McAlpine, Herb Teskey, and Guy Shewell; front, left to right: Monty Wood, Dick Vockeroth, Bobbie Peterson, and Willi Hennig. (Picture and caption from Cumming *et al.* 2011: 548.)

Monty had only been on staff in the Diptera Unit of the CNC for a short time, since 1964, when the MND was approved and Hennigian phylogenetics was adopted as a guiding principle. Monty would go on to coordinate the third volume with Frank McAlpine (McAlpine & Wood 1989) and coauthor the chapter on *Phylogeny and Classification of the Nematocera* with Art Borkent (Wood & Borkent 1989) but here I want to focus on Monty's tachinid research leading up to his *Blondeliini* revision (Wood 1985) and chapter in MND (Wood 1987).

Monty had worked on black flies for his Ph.D. and had used phylogenetic methods during that study. He was also already familiar with tachinids when he was hired by Agriculture Canada to work at the CNC. If he had not known it already, he soon realized as his knowledge of tachinids grew that the most significant work ever written on the Tachinidae and their relatives, the 12-volumes of C.H.T. Townsend's *Manual of Myiology* (1934–1942), was founded on untenable views of tachinid phylogeny. This would not have been so bad if genera were of moderate size and reasonably monophyletic but Townsend restricted most genera to single species and personally described about 1500 genera (Arnaud 1958, Evenhuis *et al.* 2015). The bulk of the new names were given to New World flies and especially Neotropical ones. Townsend's keys to genera are difficult to use and the net effect was summed up in the introduction to Monty's *Taxonomic Conspectus of the Blondeliini of North and Central America and the West Indies*:

“Townsend's generic concepts seldom admitted more than a single species per genus. As more and more species were discovered, he erected new genera for nearly all of them, sometimes relying solely on published descriptions of others. This approach culminated in a vast array of monotypic genera ..., separated on the most insignificant differences, and on occasion, the 2 sexes of the same species separated in different genera. Such excessive splitting at the generic level contrasted with large, undefined, and usually polyphyletic tribes. Subsequent authors, finding it difficult or impossible to assign their new species to Townsend's genera, proposed still more of their own (e.g. Thompson 1968). The result has been the creation of hundreds of what I believe to be unnecessary taxa.” (Wood 1985: 4.)

Monty's solution was to visit museums in the New World and Europe to examine types and rebuild a classification for them, using Hennigian methodology and searching for shared derived character states (synapomorphies). How was this accomplished in an age before computers, spreadsheets and digital cameras? Partly by taking notes but more importantly by committing to memory the features of type specimens:

“It seems to be a matter of training the brain, and my brain has been trained to spot useful details on a tachinid body, and to recognize thousands of profiles and bristle arrangements” (*in litt.* to PHA, 9 February 2000)” (quoted here from Adler & Currie 2021: 8).

The idea of rebuilding tachinid classification from the ground up based on a memory of thousands of described species may sound fanciful; i.e., simply impossible given the diversity of the Tachinidae. Monty was not a boastful man and I never heard him claim to have a phenomenal memory, yet I witnessed it in action time and again. For example, he could often identify a specimen I showed him from his memory of a type he had examined in a collection decades before. Similarly, it was rare to show him a problematic specimen in the CNC that he could not recall having looked at before and wondered about.

At the same time that Monty was visiting collections and memorizing types, he was looking for underlying features that could group them together according to synapomorphies, or at least according to similar characters that might

have a phylogenetic basis upon further study. This seems to have been at the center of Monty's interest in tachinids and he left behind brief notes on the possible synonymy of hundreds of generic names. He was less interested in describing new species—even though he had discovered many—than in placing the old. In his *Blondeliini conspectus* (Wood 1985), for example, he proposed 177 generic-level synonyms, 67 species-level synonyms, and 321 new combinations for species; he described no new genera and only three new species. Similarly, his tachinid key in MND “covertly” proposed almost 200 additional generic-level synonyms for genera mostly from north of Mexico (Wood 1987, reviewed in O'Hara & Wood 1998).

Monty's philosophy was one of searching for derived similarities, not just differences. I will use his own words to illustrate this:

“By an unfortunate accident of fate, early specialists in the Tachinidae, from Robineau-Desvoidy (1863) to Townsend (1934-1942), have been more concerned with differences than with similarities. This perspective has resulted in finer and finer splitting at the generic level. Proliferation of genera by these authors became an end in itself, at the expense of a deeper understanding of higher levels of classification. Because of the superficial heterogeneity in appearance of many adult tachinids, few stable taxonomic characters have been found that may be depended upon to indicate relationships. Instead, Townsend used various permutations and combinations of the presence or absence of bristles but made little use of their relative size or arrangement. Unfortunately, there are only a limited number of possible combinations of presence or absence relative to the many thousands of extant species and most of them do not seem to serve as a basis for understanding relationships or building a stable classification. Descriptions of new taxa, even today, sometimes neglect to mention unique features, but instead, present further combinations or recombinations of the same limited suites of characters. Furthermore, these character suites are usually not correlated with internal structure, life cycle, larval characters, or other character states; instead, they appear, presumably by convergence or parallel development, in various genera that seem unrelated on other characters of terminalia or developmental stages. Such convergence, multiplied many times over because of the vast number of superficially similar species, has resulted in a long history of incorrect identifications and misunderstandings of relationships, a trend that still continues. A classification based on shared derived character states is badly needed but will be difficult to develop and may be long in coming. This need is now being met by research into internal structures, terminalia, larvae, and life cycles. However, much remains to be discovered, and even the most recent classifications still rely mainly on arrangements of bristles and probably contain few monophyletic taxa.” (Wood 1987: 1200.)

The greatest legacy of Monty Wood in the published world of tachinidology, from my perspective, has been his contributions toward consolidating New World genera into more recognizable entities. By so doing, he made possible the keys to genera of America north of Mexico in MND (Wood 1987) and Central America (Wood & Zumbado 2010), and gave those of us following after him a more solid footing from which to conduct our studies.

As we forge ahead with the Tachinidae we would do well to keep in mind this passage Monty quoted from Hennig in the Preface to the third volume of MND:

“... there is no simple and absolutely dependable criterion for deciding whether corresponding characters in different species are based in synapomorphy. Rather it is a very complex process of conclusions by which, in each individual case, ‘synapomorphy’ is shown to be the most probable assumption. [Hennig 1966: 128].” Wood (1989: v.)

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Monty Wood – a life of travel around the world for tachinids sake



by Juan Manuel Perilla López

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Perhaps there is no better way to illustrate the life of the multi-talented D. Monty Wood than in the words of Adler & Currie (2021), “*a naturalist and systematist nonpareil*”. It would be fair to add that he was eager to collect as many tachinids as possible from all biogeographic regions to advance his understanding of the phylogeny of the family. Monty’s travels resemble in some ways those of 19th Century explorers who devoted their lives to exploring remote places, motivated by curiosity about the unknown and collecting as much information and specimens as they could that would later serve as the foundations of their revolutionary ideas.

Admittedly, this comparison may seem a magnification of Monty’s contributions. But his authorship of chapters in the *Manual of Nearctic Diptera* (e.g., Wood 1987), on the phylogeny of the Nematocera (Wood & Borkent 1989), on external morphology of Diptera (Cumming & Wood 2009), on homology of male terminalic characters (Wood 1991, Sinclair *et al.* 1994, Cumming *et al.* 1995, Sinclair *et al.* 2007), and a book on the black flies of North America (Adler *et al.* 2004), among other works, speak for themselves. Such comprehension of the diversity of this insect order that constitutes around 10% of animal diversity was reached through Monty’s involvement as one of the coordinators of the *Manual of Nearctic Diptera*, his opportunities to work on distantly related families like black flies and tachinids, but also through an entire life of collecting expeditions and observations.

Monty was interested in finding zoogeographical connections between the Palaearctic and Nearctic regions through Beringia (Wood 1978, Lafontaine & Wood 1988) and this interest led to multiple expeditions to the Yukon territory in northern Canada early in his career, and later to the eastern regions of the former Soviet Union and Japan. At the same time, Monty’s friend Vera Richter at the Zoological Institute in St. Petersburg had similar interests in species with Holarctic distributions. Their collaborations resulted in a couple of papers on the tachinids of the Yakutia and Kamchatka regions (Richter & Wood 1995, 2004).

Monty’s growing interest in rearranging the classification of the world Tachinidae took him throughout the New World on multiple travels, including most of the countries of Central America and down through the Andes and Patagonia. Monty was especially fond of visiting summits in search of hilltopping tachinids. These hilltops also allowed him to observe, and speculate on, the characteristics of mate-searching behavior in tachinids on summits (Wood 1987, 1996). His enthusiasm for hilltops and their tachinid visitors motivated his colleagues to collect on hilltops as well (O’Hara 1996, 2012, Tschorsnig 1996, Stireman *et al.* 2018).

Monty’s determined spirit took him, and on most occasions also his dear wife Grace, on a myriad of travels over five decades around the world, in many places undoubtedly engaging in constructive discussions with other restless colleagues and friends. I have used the georeferenced records available in the Canadian National Collection of Insects

(CNC) specimen database to plot on the maps below the many locations where Monty collected insects (Figs. 1,2). These records total about 47,000 for specimens of all orders, with tachinids making up over 16,000. However, these records do not fully document all of the tachinids in the CNC that Monty collected because only a portion of his tachinids have been databased. Locality data from all databased insects collected by Monty has been used to help show where Monty collected around the world. Most of Monty's tachinids were collected after his retirement in 1986 and were collected, pinned and labelled at his own expense. They were later donated to the CNC over the course of many years. Such a personal collecting effort not only assisted Monty to revolutionize the classification of the New World Tachinidae, but also made available for future generations of dipterists a huge number of undescribed taxa not otherwise represented in natural history collections. Thanks Monty!

My thanks to Jim O'Hara for his help with the drafting of this note.

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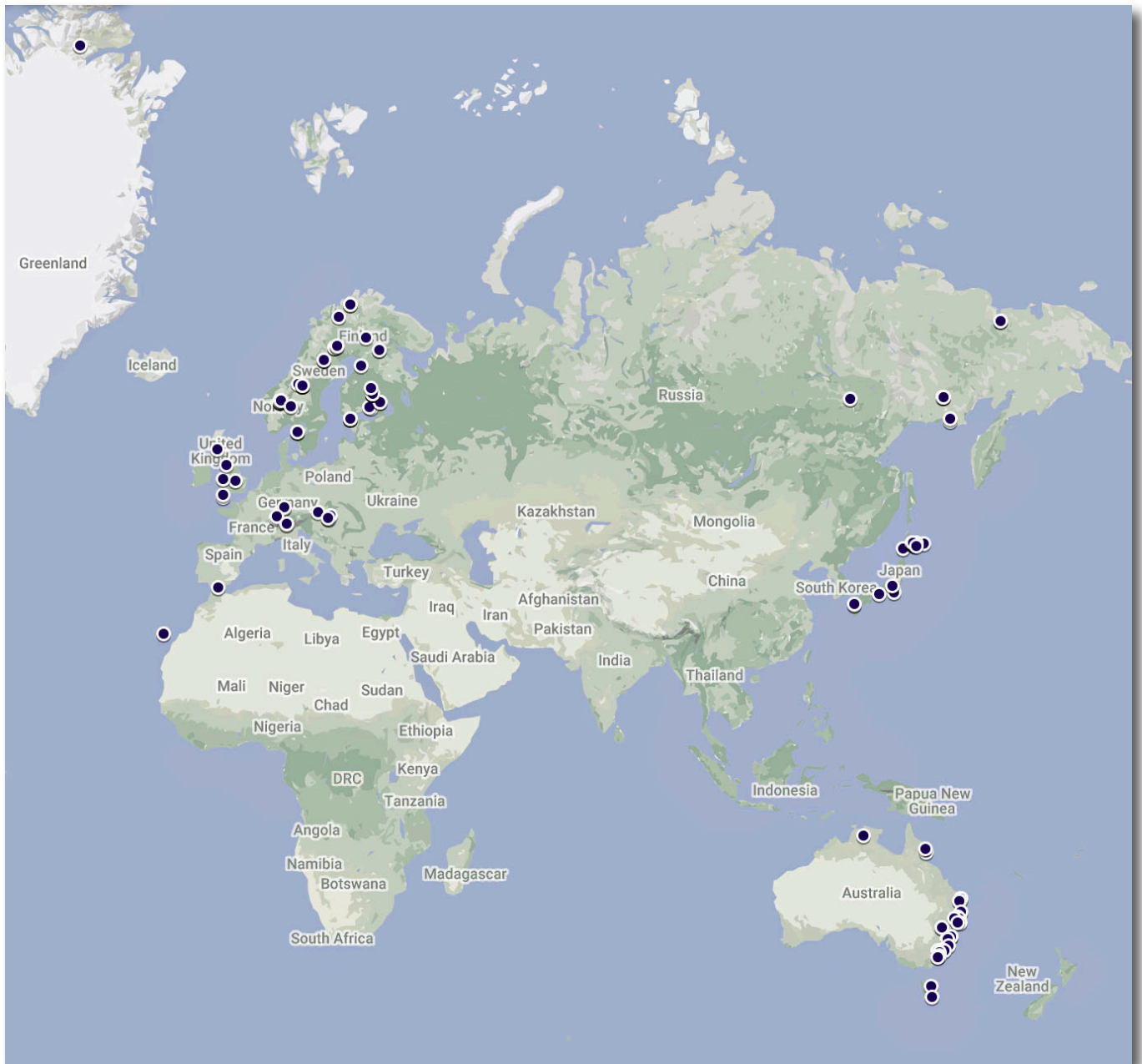


Figure 1. Map of the Old World and Greenland showing places where Monty collected insects.

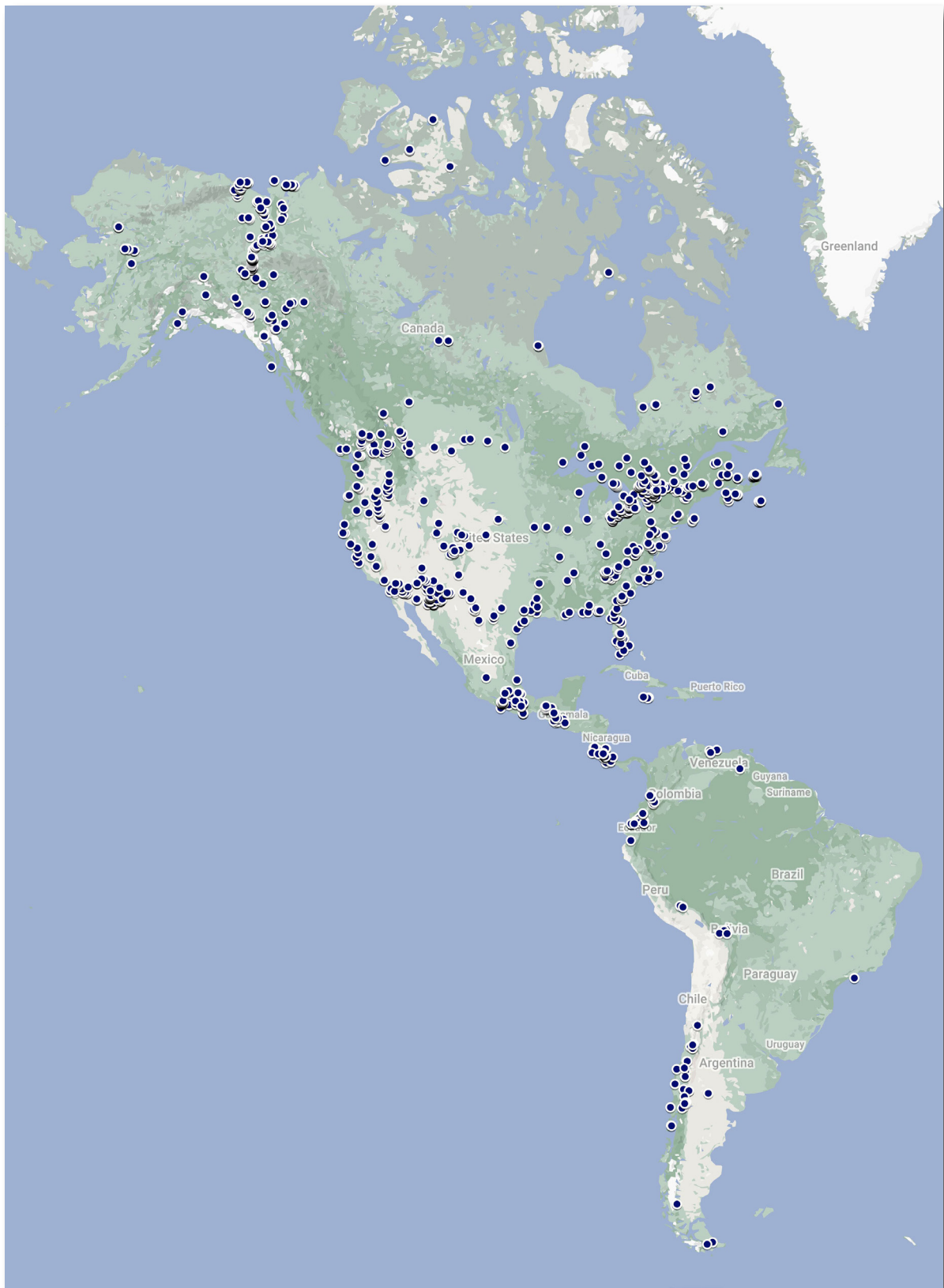


Figure 2. Map of the New World showing places where Monty collected insects.

Remembering Monty Wood (1933-2020)

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Reminiscing about collecting with Monty in Mexico

Chiapas, 1991

“I first met Monty in 1979 when I was a beginning graduate student at Carleton University in Ottawa. Each year, Henry Howden, one of the professors at Carleton, would take a February trip to the American tropics to collect beetles. In 1980 that trip was to Costa Rica and I desperately wanted to go. Monty and Grace were going and I guess Monty found out I wanted to go so he offered to pay for my airline ticket if I would collect him flies. He gave me the appropriate instructions and while there I collected a couple of boxes of big fuzzy tachinids. He seemed pleased with the catch and I hope thought the money was well-invested. In later years our paths would cross again after I got a job in Ottawa at the Canadian Museum of Nature. In those intervening years I had the opportunity to take a few trips to Mexico and had developed some good contacts there. One place I planned to go was the Mexican state of Chiapas, particularly the highlands around San Cristobal de las Casas. Monty approached me and asked if we wanted to undertake a joint field trip. So in September of 1991 Monty and I flew to Tuxtla Gutierrez and rented a car. We drove to San Cristobal and arrived at our destination late in the evening. Through a fellow coleopterist who had worked in San Cristobal we had arranged for Monty and I to stay in a spare room associated with a small local restaurant. When we arrived the restaurant was closed but you could clearly hear people inside so we knocked. No response. We knocked again, louder, with Monty getting impatient. Finally, a guy came to the door and told us they were closed. As he went to close the door Monty stuck his foot in the door effectively holding it open and in a loud authoritative voice said in Spanish we wanted to speak to the owner. He came to the door and upon introducing ourselves we were allowed to enter. Turns out they were having a bachelor party, drinking and gambling, and they thought we might have been hired by some of the wives to check up on them. We were invited to join the party and over the course of the next 8 days enjoyed the owner’s many stories and excellent food.

During that trip the one thing both Monty and I wanted to do was get into these very high elevation patches of cloud forest on the local mountain tops. Two places near San Cristobal had drawn our attention, Cerro Huitepec and Cerro Tzontehuitz, both accessible by decent gravel roads. So, each morning at the crack of dawn we would eat and head up into the mountains. As it usually clouds over in the early afternoon and is raining by mid-afternoon (if not sooner) Monty wanted to get to the tops of the peaks while it was still sunny as he expected these places to be rich in tachinid diversity. These day trips were highly successful for us both with Monty filling vial after vial of tachinids he was collecting in sunny areas laced with his Coca Cola and honey mixture which he would spray liberally over the plants around the clearing. By 4 PM we were usually back at the restaurant with me filling Berlese funnels with leaf litter for my beetles and Monty mounting and dissecting hundreds of flies, most nights finally putting out the lights at 2 or 3 AM.”

(continued on next page)

Remembering Monty Wood (1933-2020)

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(continued from previous page)

Oaxaca, 1992

“During the 1991 trip Monty found out that one of the areas I had visited while I was doing my post doc at Texas A & M University was a place called File De Caballo in Guerrero state, another one of these high elevation patches of wet cloud forest. Monty asked if this was near the important *Biologia Centrali-Americana* type locality of Omilteme. It turns out it was and so in July of 1992 Monty and I returned to Mexico but this time planning to collect up near Filo de Caballo as the primary focus of the trip. I also suggested we might want to try the sites along the road from Valle Nacional on the eastern side of the mountains in the state of Oaxaca up to Oaxaca city as this might be the most species rich area I know of in Mexico for beetles with the road rising from 300m at Valle Nacional up through 3000m then dropping into the dry central valley and Oaxaca City.

We flew into Acapulco and rented a car, a newer model that took unleaded gas. At that time unleaded gas was not common in Mexico and only a few Pemex stations carried it so we had to be careful not to get caught between gas stations with ‘sin plomo’. It took less than a day for us to get stranded. On the main highway south out of Acapulco we made it about 100 kms before we ran out of gas. Monty got out of the car and flagged down a passing truck and shouted “I’ll be back” and off he went. About 2 hours later he was back, in a taxi, with a full jerry can of ‘sin plomo’. Turns out he had to go about 60 kms to find the right gas station. For the rest of the trip we filled up every time we passed a station with the right gas. After heading south along the coast and taking forays up into the mountains down to Puerto Escondido, we turned around and went back up to Chilpancingo in Guerrero from where we would conduct daily trips up to Filo de Caballo – there being no accommodations in the highlands. We carried out 4 or 5 such daily trips having great success finding lots of both flies and beetles. On our last day at the hotel the young front desk clerk finally asked what we were doing. We told her we were entomologists and after she had told us how dangerous it was up there she laughed and said that the staff thought we were from the U.S. Drug Enforcement Agency (or DEA). We laughed as well, but inside were thinking how lucky we were that the people up in Filo de Caballo did not think the same thing. Maybe they did?

Like many great entomologists Monty loved field work. I was fortunate to have taken these two memorable trips with him and to have shared his companionship. He was a wonderful man, generous with his time, his knowledge and friendship.”

Bob Anderson, Ottawa

February 28, 2021

Remembering Monty Wood (1933-2020)

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Monty Wood (centre) with Jim O'Hara (left) and Pierfilippo (right) after a day of collecting during the field meeting of the North American Dipterists Society in Silver City, New Mexico, USA, August 2007.

“People who knew Monty personally agree in recognising him as a great interpreter of the natural world in general and a generous colleague always willing to share his knowledge. Monty’s written contribution to the knowledge of tachinids (which is my main scientific interest and the reason why I met him in 1998, during the 6th ICD in Oxford, UK) is remarkable, but what makes Monty special, not to say mysterious and fascinating, is that what he wrote is nothing compared to his actual knowledge on this subject. Much of his knowledge was thus delivered personally in a Socratic fashion. While talking, Monty could spin together anecdotal stories with great pieces of knowledge that could change your point of view once and for all. And one of my favourite places where I conversed with him was in the aisles among the cabinets of the CNC tachinid collection in Ottawa, where Monty had a chunk of the immense complexity of tachinid diversity at hand. He liked to pick out and show me ‘tricky’ specimens; those that puzzle any taxonomist by rejecting easy answers about taxa circumscriptions, and I liked that too! For each of the ones he had collected himself, he remembered the place of collection and circumstances (i.e., weather conditions, time of the day, place of sitting), with details on behaviour and personality of the fly. So, a single fly turned often into a trigger to talk about nature in general. — Monty, you had a deep impact on my way of thinking as a tachinid taxonomist, thank you very much, I will miss you!”

Remembering Monty Wood (1933-2020)

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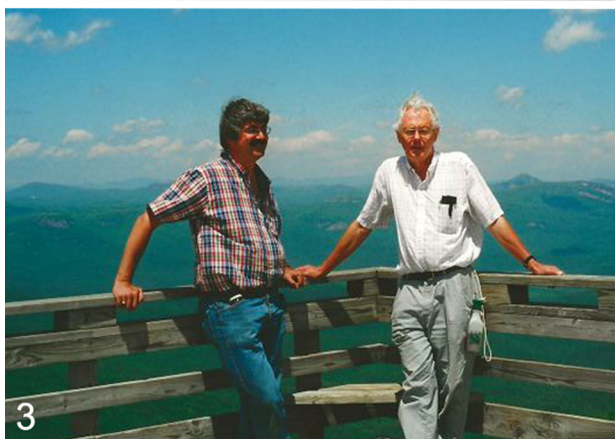


“By 1999, the National Institute of Biodiversity (INBio) in Costa Rica had an excellent group of expert collaborators who came and went. Some of them became well-known and well-loved faces. One was Monty, a kind and placid man. Always with a smile on his face, always listening patiently before answering. He didn’t miss an opportunity to practice his Spanish. He loved what he did. I first met Monty in 1992. The day I met him, a colleague informed me that he was an expert on Tachinidae and I did not waste a second in going to him and introducing myself: ‘Hello! My name is Jorge Corrales and I work with Limacodidae’. To which he replied with a smile: ‘Limacodidae? mm ... *Austrophorocera* food!’ From there we became friends and he frequently came to my office to talk about anything but Tachinidae. At that time we drew caricatures of all the staff in the Entomology and Botany department and dear Monty was included as one of only two non-staff to be granted such an ‘honor’. This drawing was based on sketches I made of Monty in the field and at lunch times. I remember him fondly.”

Remembering Monty Wood (1933-2020)

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1. Monty, Grace and Civil War cannon at summit of Kennesaw Mountain, Cobb County, Georgia. **2.** Monty and Grace at Rabun Bald lookout tower, Chattahoochee National Forest, Rabun County, Georgia. Good tachinid and sarcophagid collecting! **3.** Greg and Monty at Rabun Bald lookout tower. **4.** Grace and Monty at Rabun Bald lookout tower.

Greg, a sarcophagid specialist, has a passion for collecting sarcs on mountain summits where males congregate to “hilltop” and mate with more transitory females. Tachinids also “hilltop” and Monty sought out summits on every continent he visited, with great success. Harold Dodge and John Seago collected sarcs on hilltops in Georgia (USA) in 1952* and Greg suggested to Monty and Grace that they should revisit some of the hilltops to see what interesting species they might catch there nearly 50 years later. Monty and Grace jumped at the chance and the three of them set forth on a tour of six Georgia hilltops in May of 2000. The pictures above are from that trip.

An account of the trip is given here: <https://www.nku.edu/~dahlem/GA%202000/gatrip.htm>

* Dodge, H.R. & Seago, J.M. (1954) Sarcophagidae and other Diptera taken by trap and net on Georgia mountain summits in 1952. *Ecology*, 35, 50–59.

Remembering Monty Wood (1933-2020)

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Pictures from the field meeting of the North American Dipterists Society, Silver City, New Mexico, USA, 13–16 August 2007. Above: Monty Wood, Jeff Cumming and Grace Wood take a break from collecting at Cherry Creek campground area in Gila National Forest ca. 17kms north of Silver City. Left: Jim O'Hara and Monty pose for a picture while hilltopping on the summit of Eighty Mountain in Gila N.F. ca. 7kms north of Silver City.

“Monty and Grace spent a lot of time travelling the world in search of tachinids after Monty’s early retirement in 1986. For trips within Canada and United States they frequently took their truck camper and spent their nights in national forests and campgrounds close to good collecting spots. I was pleased when they decided to drive down to New Mexico in 2007 to attend the only NADS field meeting I ever organized. It was held in Silver City just south of Gila National Forest. The southern portion of the national forest is one of the best spots in southwestern United States for tachinid collecting and a personal favorite of mine. Although the meeting was held in the middle of the monsoon season (the best time for tachnids), we had uncommonly clear skies and perfect temperatures. Monty and Grace were very pleased with the number and diversity of tachinids they caught and with the comradery of fellow dipterists. They returned to Silver City in their camper in 2011 to collect in the area again.”

More about the NADS field meeting here:

O’Hara, J.E. (2007) Field meeting of the North American Dipterists Society, Silver City, New Mexico, 13-16 August 2007. *Fly Times*, 39, 3–5.

Remembering Monty Wood (1933-2020)

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“I have one photo to share, This picture was taken in Canberra in August 2012 when Monty and Grace last visited Australia. Brian Wiegmann was visiting on sabbatical and the fourth person in the photo is my wife Francoise Berlandier. Monty spent a number of weeks in ANIC sorting and identifying our tachinids, and sampling our red wine in the evenings. I remember him saying that there were many tachinid groups in Australia that looked superficially like Northern Hemisphere groups, but were not related. He was always a very knowledgeable and approachable colleague, and his contributions to the Manual of Nearctic Diptera stand as testament to his global impact, vision, passion and expertise.”

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Included here are references on the Tachinidae that have been found during the past year and have not appeared in past issues of this newsletter. This list has been generated from an EndNote 'library and is based on online searches of literature databases, perusal of journals, and reprints or citations sent to me by colleagues. The complete bibliography, incorporating all the references published in past issues of *The Tachinid Times* and covering the period from 1980 to the present is available online at: <http://www.nadsdiptera.org/Tach/WorldTachs/Bib/Tachbiblio.html>. I would be grateful if omissions or errors could be brought to my attention.

Please note that citations in the online Tachinid Bibliography are updated when errors are found or new information becomes available, whereas citations in this newsletter are never changed. Therefore, the most reliable source for citations is the online Tachinid Bibliography.

I am grateful to Shannon Henderson for performing the online searches that contributed most of the titles given below and for preparing the EndNote records for this issue of *The Tachinid Times*.

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