Cranefly News

The Dipterists Forum Cranefly Recording Scheme

For Superfamily Tipuloidea & Families Ptychopteridae & Trichoceridae

Newsletter No 38 **Spring 2022**

Editor: John Kramer

Larva of Tipula confusa with friends. Photo. EGH

Another interpretation of the hypopygium of *Paradelphomyia neilseni* (Kuntze) (Limoniidae) - C. Martin Drake

Paradelphomyia are distinctive craneflies quickly recognised to genus but can be awkward to identify to species. The smallest British species is *P. neilseni* whose identification seems easier to me if the genitalia characters in Stubbs (2021) are ignored. Its narrow wing with scarcely any anal lobe and very sparse microtrichia confined to the outer halves of the cells are enough to place it (Fig.1). The reason for ignoring the genitalia is that the figure of the hypopygium, reproduced from Edwards (1938) and perpetuated by Coe *et al*. (1950) and others, shows two features that are not apparent in specimens that I recently collected. These features are the very long backwardly pointing aedeagus and the tiny 'H'-shaped apodeme at the base of the aedeagus.

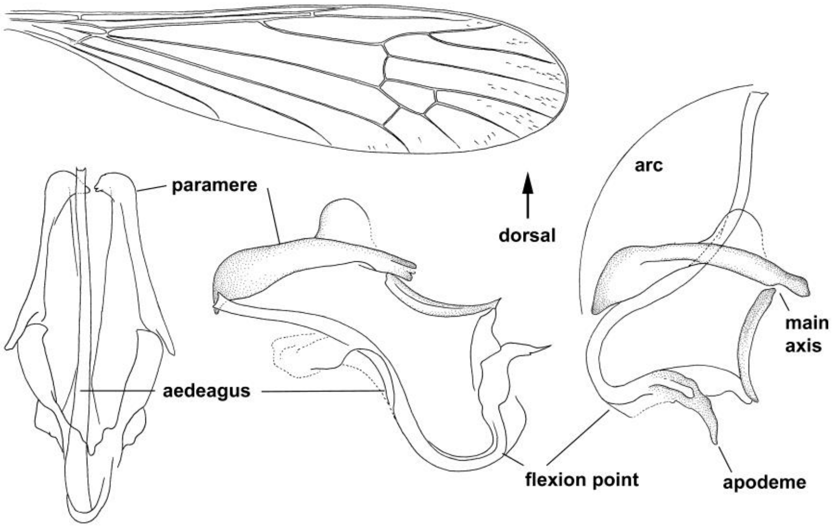

Fig. 1. *Paradelphomyia neilseni* wing (flattened under cover-slip) and aedeagus and parameres in ventral and lateral views showing the aedeagus in its retracted (mid fig.) and extended positions (right fig.), with the arc followed by the tip of the aedeagus. The lateral views show the parameres in their natural position with the dorsal side uppermost.

The apodeme is the easiest to deal with. It is a tiny scrap of chitin that is not easy to make out; in dorso-ventral view, it is nearly rectangular and has no projections, in contrast to other British species in which the apodeme is conspicuous and usually diagnostic. Tjeder (1952) illustrated the apodeme of *P. nielseni* as small polygon with a slightly expanded tip but with no projections, agreeing roughly with my specimens.

The aedeagus is a more interesting structure. It clearly can change position from retracted to extended, as John Kramer (2015) noted. My sample of many males showed all states (Fig 1). In the retracted position, the tip of the aedeagus is level with the tips of the parameres, in a different orientation to that in Edward's (1938) figure, but which looks superficially similar to that of the non-British *P. nigrina* (Lackschewitz), as illustrated by Tjeder (1952) as *Oxyrhiza septentrionalis* and reproduced by John Kramer (2015) and Alan Stubbs (2021). Hence there is a good chance of getting temporarily excited in finding this species, only to be disappointed when the key is followed more carefully. When the aedeagus is extended, it usually points upwards or diagonally backards (dorsally or postero-dorsally) between the parameres, and in dorsal view it does not extend far beyond the paramere tips. In only one example in my sample did it point backwards as Edwards illustrated. To check how the aedeagus moved, I gently manipulated a dissected example in viscous warm glycerine jelly (Ackland 2015). The aedeagus can be made to bend at two points, one being a main articulation where its stout forked base it meets the two parameres, and a second less clearly defined axis just distal to the apodeme where the single duct will bend but quickly spring back to its original position. If this more distal joint is just a weak flexion point and not a true articulation, except perhaps when the whole complex is under some strain during copulation, then movement of the aedeagus is usually limited between the two extreme positions that I illustrate, and the extent of its movement is shown by the arc made by the tip of the aedeagus around the single main axis with the parameres (Fig. 1). John Kramer (2015) suggested that retraction of the aedeagus caused the hair-pin bend but the whole 'hair-pin' is rigid apart from the weak flexion point. So although it is possible to force the aedeagus to point backwards, and thus extend far beyond the parameres, I feel that Edwards often-reproduced figure almost certainly shows an extreme example or even an artefact of his preparation in which he did force it back beyond its normal position. Care is therefore needed when interpreting the hypopygium of *P. nielseni*. However, a protruding aedeagus does seem to be characteristic of this species only, although what happens is *nigrina* remains to be discovered.

Paradelphomyia neilseni has only rarely been recorded in Devon so the 2021 find was particularly interesting because the population was large, with this species being one of the most frequent craneflies in a small patch of possibly slightly acidic hillside seepage under sparse sallow (*Salix cinerea*) woodland. Some of the less common craneflies at this seepage were *Dicranota claripennis* (Verrall), *Lipsothrix remota* (Walker) and *Paradelphomyia fuscula* (Loew). (Devon: Knapp Copse, SY156953, 11 Oct 2021).

I thank East Devon District Council for permission to collect on their local nature reserve, and John Kramer for reminding me of his paper.

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***Paradelphomyia dalei* found in France.**

In a well-presented paper by Pierre Tillier and Clovis Quindroit specimens of *P. dalei* have been reported from five sites in France. This species had been previously recorded only from sites in the UK and was prreviously thought to be endemic here. The habitats reported conform to the 'calcareous wet woodland' described in 'British Craneflies' as typical in Britain.

Reference

Tillier, P. and Qunindroit, C. 2021. Découverte en France d'une espèce de Limoniidae supposée endémique de Grande-Bretagne : *Paradelphomyia dalei* (Edwards, 1939) (Diptera). Bulletin de la Société entomologique de France 2022

Ed.

Vertical movement of *Tipula (Savtshenkia) confusa* van der Wulp, 1883 larvae in response to flooding.

E.G. Hancock,

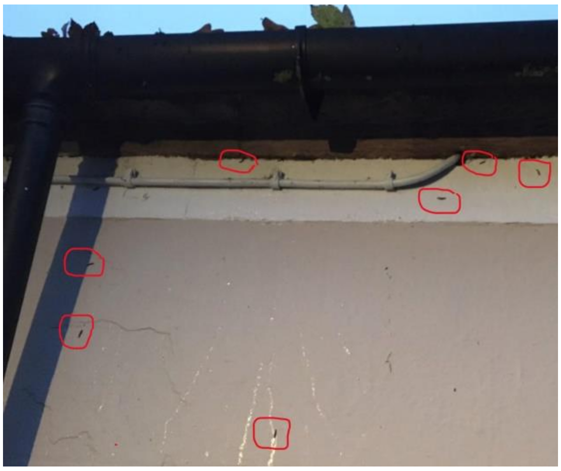

Fig. 1 Position of larvae high up on the house wall.

This behaviour is described in order to ask if any similar observations have been made elsewhere. After some persistent rain *Tipula* larvae (identified as *confusa* from specimens collected) were seen on the wall of my house possibly moving to a drier place to avoid drowning. They had developed in moss covering part of the surface of the concrete yard; samples of which contained many larvae. The yard concrete comes right up to base of the wall, which is rendered and painted, so there is no refuge on the horizontal plane for the larvae if threatened or disturbed. A considerable number of larvae were first noticed on 12th November 2021 (Fig. 1) after two days of rain.

The next day I was cleaning moss and slippery algae from stone steps at the back of the house using a hose pipe and stiff brush. Having soaked the steps and partially completed the job upon returning to finish an hour or so later three larvae were seen crawling up the adjacent wall. Clearly, they had been disturbed by this activity; it was not raining at the time. Having become aware of this 'migration' it has been seen several times since. The temperature was about 10 degrees C., quite mild compared to sub-zero temperature during intervening clear nights. On all the dates it had been raining or drizzling for several hours prior to seeing the larvae. Another sighting on 11th December was in the evening when five larvae were performing this feat in darkness.

I have not seen this behaviour before which may be due to my lack of attention or have been too diligent in previous years in sweeping the yard free of moss earlier in the season. At a natural site it would be difficult to witness such an event, if there is a situation which required such movement, as tree trunks or rank vegetation would conceal any larval activity from view. There are a number of questions to consider. Do they go back down again, and if so when and how soon after it stops raining? The walls get wet from the rain but on drying the larvae would be less able to grip the vertical surface with reduced surface tension. I have not witnessed an entire journey but seen them stop, move sideways or just sit on a windowsill that provides a horizontal ledge as a resting place (Fig. 2). Obviously, there is opportunity for experimentation here. The hypothesis is they avoid temporary flooding by equally temporary vertical movement. The larvae lack abdominal prolegs but appear to move by peristaltic contractions of the body which remains in contact with the wall. The head seems to act as a forward anchorage point, lifting off the substrate to reach out to a suitable part of the surface for gripping. The last segment has lobes ventrally about the anus which in contact with the surface may provide sufficient purchase to assist forward progression during the wriggling (Fig. 3). Video close-up imaging on a glass plate may help with defining movements. Any comments are welcome.

E.G. Hancock, Hunterian Museum, University of Glasgow.

More on *Dicranomyia radegasti*

In Cranefly News #37 there was a description of a specimen of *Dicranomyia radegasti* Starý 1993, caught and identified in Scotland by Kjell Magne Olsen. All of the male diagnostic characters described by Starý in his 1993 paper. were were shown as photographs, apart from the hind tarsal claw. This is shown in Starý's specimens as slightly longer than in *D. chorea*, and slightly undulating, something to look out for in future British specimens.

Kjell Magne sent some more details of the habitat in the Glen Nant NNR, which is shown in the adjacent photo, and which is very similar to that described by Jaroslav Starý.

Fig. 1 Habitat of *D. radegasti*. Photo K.M Olsen

Observations on the phenology and sex ratios of craneflies (Limoniidae) and a few other Diptera found in emergence traps. Robert Wolton

In 2020, I ran four emergence traps in a wet woodland on our farm in Devon between the beginning of May and early October (excepting the month of August), as detailed in Wolton and Field (2021). In addition, the following year I ran a couple of traps in the latter half of April to get some early season data. For those taxa I was able to identify to species or genus level, I recorded the numbers of each sex caught. This information has enabled me to explore both flight times (phenology) and sex ratios, with the outcomes explained below.

The traps captured 30 or more individuals from 15 taxa – I reckon 30 to be the minimum necessary for meaningful analysis. Eight of these taxa are craneflies: *Austrolimnophila ochracea* (30 individuals in 2020), *Dicranophragma adjunctum* (31), *D. nemorale* (37), *Euphylidorea dispar* (46), *Paradelphomyia senilis* (63), *Phylidorea fulvonervosa* (69), The *Erioptera* species emerging into the traps were *fuscipennis* (6 males) and *lutea* (57 males), while the *Molphilus* species were *appendiculatus* (3 males), *bifidus* (6 males), *flavus* (13 males), *griseus* (34 males), *medius* (13 males), *obscurus* (2 males) and *ochraceus* (58 males). *Erioptera* (76) and *Molophilus* (209) (females of the last two genera cannot be identified confidently to species level). (The names of the other seven taxa are given in Figure 2.)

Figure 1 presents phenology charts, using 2020 data. Assuming generations do not overlap seamlessly, three species have one generation (*A. ochracea*, *E. dispar* and *P. fulvonervosa*), four taxa two generations (*D. adjunctum*, *D. nemorale*, *P. senilis* and *Erioptera* spp.), while together the seven *Molophilus* spp. have three, possibly four, generations.
Males emerged earlier than females in each generation for most species In *E. dispar* six males and no females emerged in April 2021., a frequently observed phenomenon in flies (e.g. Buck 2001, Hadley 1969), so no surprise there. However, *A. ochracea* is an exception, the females emerging earlier than the males, as they do in *P. senilis*, at least in the autumn generation. I caught no males of *D. adjunctum* at all in the spring generation, but the probable explanation for this is that trapping in 2020, commencing on 1 May, did not cover the beginning of their season: in 2021 a single male was caught on 19 April (no females were caught that month). Why should females ever emerge before males? Are my results for *A. ochracea* and *P. senilis* anomalous, or is this a real phenomenon in these species? Earlier emergence of females is said to be a rare occurrence in Diptera and insects in general (Buck 2001).

The sex ratios of these craneflies are given in **Figure 2**, again just based on the 2020 data. While those for three cranefly taxa are not significantly different from that expected from a 1:1 ratio of males to females, for *Erioptera* and *Molophilus* significantly more males than females were caught, the converse being true for *A. ochracea* and *E. dispar*.

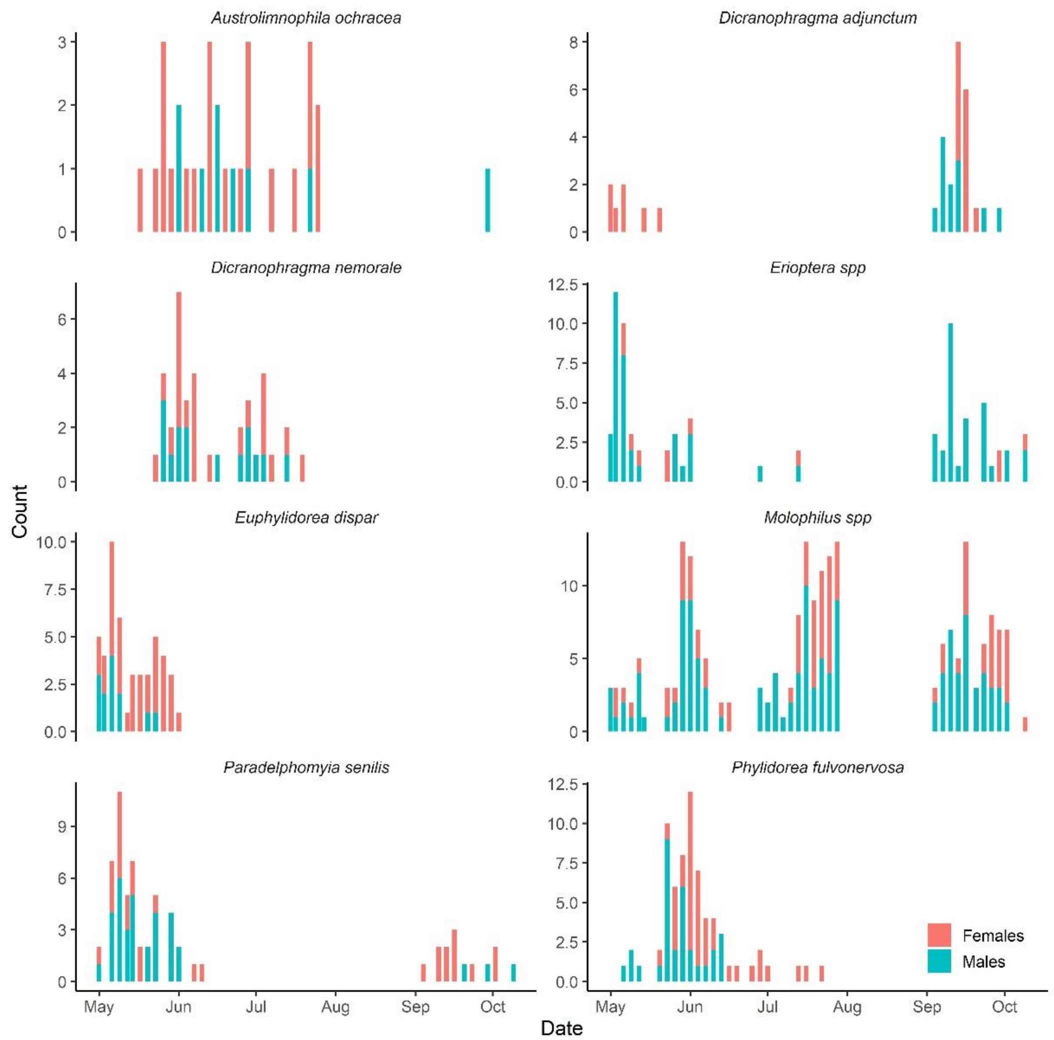

Figure 1. Phenology charts for the eight most numerous cranefly taxa caught in emergence traps in 2020 in a wet woodland at Locks Park Farm, Devon. The traps were operational between 1 May and 9 October, excepting the month of August.

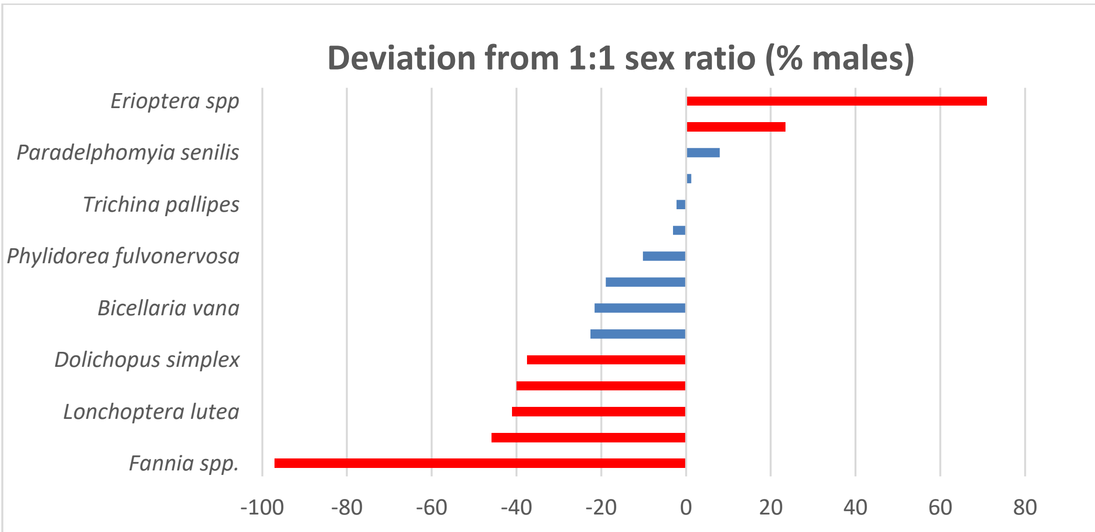

Figure 2. Sex ratios of the 15 taxa identified to species or genus level where 30 or more individuals caught in emergence traps in 2020. Central vertical line is 1:1 male:female ratio. Extreme left line indicates 100% female, extreme right 100% male. Red bars show significant differences from 1:1 at 5% level (χ^2 test).

Some of these variations away from a 1:1 M:F ratio could be explained by trapping taking place only between early May and early October: if more of one sex than the other emerged earlier or later in the season this would skew the results. Examination of the phenology charts suggests that this might be true for *E. dispar*, especially since between 10 April and 1 May 2021 six males of that species were caught, but no females. The strong bias towards females in *A. ochracea* cannot easily be explained in this way and continues to baffle me.

I also remain perplexed as to why more male *Molophilus* and many more male *Erioptera* emerged than females. It is unlikely to be an artefact of the trapping season not starting early enough, since in April 2021 all 18 *Erioptera* (*lutea*) caught were male bar one, and the three *Molophilus* (*griseus*) were all male too. The bias could perhaps be explained by the majority females never flying or crawling up the sides of the emergence traps, so avoiding capture. They may be mated soon after emergence, perhaps even while still teneral, and, finding the surrounding medium suitable for oviposition, never move more than a few centimetres. However, as Alan Stubbs has pointed out to me, in *Erioptera* the males form swarms to attract females, so presumably here the females must usually fly to find mates; at least some *Molophilus* also swarm. Another possible explanation is that the females are more crepuscular or nocturnal than the males, being inactive when I visited the traps. That this too may not be the answer is suggested by an extraordinarily detailed study of *Molophilus ater*, a flightless species, conducted by Malcolm Hadley (1969). He also found a strong male bias: 65% of newly emerged individuals and 55% of those which pupated in the laboratory were males. Perhaps it is a characteristic of the genus that more male than female eggs are laid, or, more likely, that mortality rates differ between the two sexes at larval or pupal stages. Hadley himself was unable to account for the preponderance of males in *M. ater*.

To stray briefly from craneflies, every one of the 69 *Fannia F. aequilineata* (1 individual), *F. genualis* (3), *F. lustrator* (1), *F. serena* (35), *F. similis* (22), *F. umbrosa* (7) (Fanniidae) appearing in the emergence traps was female, the sole exception being the single *F. lustrator*. What happened to the males? If anyone can cast any light on this, I should be pleased to hear from you. Perhaps the most likely explanation is infection by male-killing parasitic microbes. The common bacteria *Wolbachia*, for example, are known to result in extreme female sex biases in some insects and have been found to occur in wild *Fannia*, including *F. serena* (Martin *et al*. 2012). Perhaps they also infect *Austrolimnophila ochracea*!

My thanks to Alan Stubbs for insights and especially to Ben Field for producing Figure 1 using R software.

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Idiocera sexguttata in the New Forest – Paul D. Brock

As a keen photographer, I like to set myself a challenge and survey insect species new to me each year. Having been asked for a photograph of the globally endangered cranefly *Idiocera sexguttata* to use for conservation purposes, also seen the report by Lovegrove et al (2018) referring to the last known record in the New Forest (2000), this species fitted the bill, with distinctive wing spots enabling identification in the field. Contact with Jack Potter (Natural England) established that he had found them at Stony Moors (approx. SZ2199) on 8 June 2018, but only recorded one in June 2019, indicating they may be elusive. As stated in *British Craneflies* by Stubbs (2021), the Forest site is an outcrop of marl (a very calcareous clay). The species also occurs in Wales and there are old records in Dorset c. 1860 and Cornwall in 1912. Recent records from a few areas of Dorset and Wales are listed in Howe (2016).

My first visit to the site was a brief recce of the site on 12 June 2021, when a male *I. sexguttata* was swept. Colin Easton and I visited on the morning of 16 June 2021 and after an hour of methodical searching had found several specimens of both sexes by sweeping and searching vegetation. Although difficult to find at rest, they were observed on bog myrtle *Myrica gale* and bramble, on leaves (including upperside) or branches. If disturbed, the slow ghost-like flight can be carefully followed, the specimen landing on nearby vegetation. Some were photographed in situ, one was brought home for more detailed photos and released on site next day. In order to minimise disturbance of the habitat, this brief survey was concluded on 16 June. The New Forest site mentioned above is small but supports good populations of craneflies in general. If looking for this species, other boggy areas and seepages in the Forest and elsewhere should be surveyed in about mid-June (a permit from Forestry England is required for the New Forest), as there is every probability they will be more widespread but overlooked, due to their small size and short flight period.

Howe, M. 2016. A new Welsh locality for the cranefly *Idiocera sexguttata* (Dale) (Diptera, Limoniidae) in 2015. *Dipterists Digest* 23(1): 47-48.

Lovegrove A., Gillingham P. and Harrison A. (2018). New Forest HLS Scheme Specialist Habitat and Species Surveys: Survey and assessment of Six-spotted cranefly. BU Global Environmental Solutions (BUG) report (BUG2772) to Forestry Commission. Higher Level Stewardship Agreement, The Verderers of the New Forest AG00300016. 19 pp. www.hlsnewforest.org.uk/app/uploads/sites/3/2018/03/Six_spotted_cranefly_survey.pdf

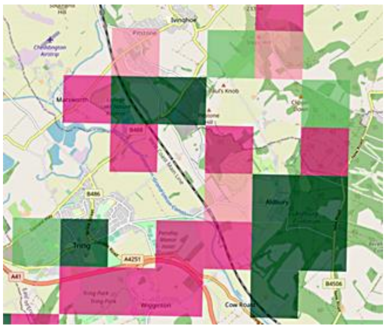
Can you help us with targeting revisit maps for craneflies?

Our friends over at the UK Centre for Ecology and Hydrology (UK CEH) have added craneflies (via the UK Cranefly Recording Scheme) to the list of groups they are collecting data upon under the target revisit mapping project. This is an online mapping tool that is helping to model trend analysis in insect populations but will double as a way of helping us gather more cranefly data for the Recording Scheme. It is also being used by our colleagues over at the Orthoptera Recording Scheme, the Ground Beetle Recording Scheme and the Soldierfly and Allies Recording Scheme.

Here is how it works.

Step 1 – visit the website at https://shiny-apps.ceh.ac.uk/targeting_revisits_craneflies/ or Google 'targeting revisits craneflies'

Step 2 – Decide where you want to survey and zoom in on an area on the UK Map.



Step 3 – You will see a number of differently coloured 1km x 1km squares (monads). Any that are blank are classed as 'unrecorded' as far as the model goes and if you can add any records for here this would be great!

Bright pink squares = targets for revisits. They have records from only one year in the past so if any records can be made in these monads, they can be included in our trend analysis.

Pale pink squares = new this year. These squares have the most recent records and will become targets for revisits next year.

Dark green squares = considered well recorded. These are already being used in the trend analysis as they have records from multiple years so are less important for the model but welcome for the Recording Scheme.

Pale green squares = successful revisits. They used to only have records from a single year but have had records added due to the targeting revisit scheme.

Step 4 – Go out and survey craneflies as you normally would considering access permissions.

Step 5 – Add your records on to iRecord.

Step 6 – Records get verified.

Step 7 – Targeting revisit maps get updated automatically and you should see bright pink squares change colour to pale green, blank squares go pale pink, and more dark green squares.

Step 8 – Sit back knowing you've done a great job and repeat next year!!!

It would be wonderful to get as many recorders adding cranefly records via iRecord to help with the trend analysis models and add new records for us here at the Recording Scheme.

Many thanks!

Pete Boardman

Can you help with the Cranefly Recording Scheme?

Now 'British Craneflies' has been published we anticipate the volume of records to increase that comes into the recording scheme. Our friends over in the Hoverfly Recording Scheme found this and have produced some interesting graphs that demonstrate how the availability of identification resources boost recording and we expect that to be the case with this scheme too.

In the last few years, we've had around 4000 records annually through this scheme. Most come through iRecord, the safest way to submit data, as if one of the current scheme members goes under a bus the data remains and can be picked up by someone else acting on behalf of the scheme. We still do get Excel spreadsheets though, which we have to process and add onto iRecord anyway so that all our cranefly data goes through iRecord one way or another eventually.

About two thirds of our data that comes through iRecord is submitted alongside a photo. Each of these has to be looked at individual to check ID and can be really time consuming, but ultimately really interesting as the quality of digital photography and camera technology has improved.

There are a number of ways people could help with the scheme in a technical or non-technical way – could you help?

1 – Social Media – could you advocate for us? Help spread the news that there is a Cranefly Recording Scheme and that we have a Twitter account (@CRStipula) currently with just over 2000 followers Could you help generate content? We have a Facebook page too with 714 members and always need people to help identify photos on there. Would anyone be prepared to set up and monitor an Instagram page? Maybe put content together for TikTok and get craneflies viral?

2 – Websites - do you have website building skills? We currently have a small presence on the Dipterists Forum website https://dipterists.org.uk/cranefly-scheme/home but it would be great to get more information on here as place for inexperienced cranefly recorders to visit. Maybe species profiles,

3 – Data Handling - Could you commit to convert Excel spreadsheets into iRecord friendly Excel imports?

4 – Cranefly Identification - Are you able to identify craneflies? Could you help with verifying for iRecord – even just common species?
5 – Cranefly training events – are you able to help run events or run events yourself with support from us? Could you host an event? Do you have a venue that we could use? We anticipate the need for more training events over the next few years with the availability of British Craneflies.
If you are able to help with any of these areas (or have other suggestions as to how you could help – please contact Pete or John.
Pete Boardman

The verification of biological records. - John Kramer

In response to an increase in recording we need to be careful in our enthusiasm, not to go for quantity over quality. It is much easier to make a record than it is to check and confirm it . The late Trevor James of the National Biodiversity Network, in his paper 'Improving wildlife data quality' (James, 2006) discussed the process and the purpose of records. He also discusses the need for data verification – 'ensuring the accuracy of the identification of the thing being recorded'. He wrote: *Recording schemes or organisations setting up a survey have a responsibility to take the lead with setting standards for identification. They should define agreed levels of 'difficulty' over the identification of the species being recorded.*

Entomology is a science, and science is an evidence-based activity. We use visual evidence in identification. The level of evidence needed to verify a species record varies from species to species, from common to rare, and from simple characters to complex ones, but sometimes it is reasonable to say 'there is not sufficient evidence on which to base a conclusion.'

We usually accept records of common easily identified species in their usual habitat but if the recorder is a novice or the habitat abnormal we may ask them for the diagnostic character that they observed. However, any claim for a record of a 'difficult', rare or a new species needs the presentation of supporting evidence. This may be for a County (or Vice-County) Recorder, or for the National Recorder. The evidence may be the specimen itself, or it may be a drawing or photograph of the diagnostic features. Important reasons for this are that structures can be missed or misinterpreted by the original observer, or the taxonomy may change and if the evidence is there, the misidentification can be corrected. It goes without saying that any recorder should be able to describe the diagnostic character which led them to their identification, in a similar process to the way that the ornithologists' British Birds Rarities Committee operates. What should we, as a recording community, accept as sufficient evidence? This paper is offered as a contribution to that debate.

Guidance for Validation
The levels of difficulty shown below can be used to sort species into groups. The statements below refer chiefly to males. For many genera a satisfactory key to females has yet to be published and in those cases, where a voucher specimen is female, it should be noted and the site searched further for confirmatory males.

Levels of identification difficulty - Criteria
Level 5. Microdissection of male genitalia necessary to display apodeme or other character. Eg. *Tasiocera, Paradephomyia, Ula mixta.*
Level 4. Some genitalia dissection needed and/or genitalia complicated and/or difficult to see. Eg. *Gonomyia, Idiopyga. Rhabdomastix.*
Level 3. Binocular microscope needed to see small features such as male styles. Eg. *Erioptera, Ormosia.*
Level 2. Diagnostic characters distinct with hand-lens. Eg. *Male Lunatipula, Limonia.*
Level 1. Diagnostic characters distinct with naked eye. Eg. *Acutipula, Limonia nubeculosa*.

Species in Group 5. Voucher specimens, drawings or photos of diagnostic characters necessary to confirm the record. Eg. *Tasiocera jenkinsoni, Paradelphomyia fuscula, P. dalei, Rhabdomastix laeta*

Species in Group 4. Voucher specimens, drawings or photos of diagnostic characters necessary to confirm the record. The genus *Gonomyia* have complex genitalia which can be difficult to make out. Parts change shape or are concealed according to the viewing angle. This means that evidence such as is demonstrated by photomicroscopy is hard-won, and difficult to present.

Species in Group 3. A description of the diagnostic features observed may be requested, especially if the species is rare or in an atypical habitat.

How common or rare a species is another criteria relevant to the evidence required for identification and this can be measured by the National Rarity Indices. If a species is common and widespread (NRI 1 or 2) the record is usually accepted without any anxiety. If however it has only previously been found in a few hectads then it would be necessary to present the full evidence with the record.

The National Rarity Indices

NRI 1	Species found in > 100 hectads
NRI 2	Species found in 30 – 100 hectads
NRI 3	Species found in 16 – 30 hectads
NRI 4	Species found in 6 -15 hectads
NRI 5	Species found in 2 – 5 hectads
NRI 6	Species found in 1 hectad only.

List available from the author.

Some examples of Verification Levels (VL) with the National Rarity Indices (NRI)

	VL	NRI	
Gonomyia bifida	4	4	Voucher
Gonomyia conoviensis	4	4	Voucher
Gonomyia dentata	4	2	
Gonomyia hippocampi	4	6	Voucher
Gonomyia lucidula	4	2	
Gonomyia recta	4	2	
Gonomyia simplex	4	2	
Gonomyia tenella	4	4	Voucher
Gonomyia abbreviata	4	5	Voucher
Gonomyia edwardsi	4	4	Voucher
Hoplolabis areolata	4	4	Voucher
Hoplolabis vicina	4	4	Voucher
Hoplolabis yezoana	4	6	Voucher

There are no hard and fast rules. A species like *Ctenophora ornata* is very distinctive and it appears to be spreading northwards. When it appeared in Sherwood Forest at light, fortunately the Pembertons were able to photograph it and remove any shadow of doubt as to the validity of their record. (CN **26**. 2013). There is a specimen of this species in the Wingate collection in Newcastle, from a site in the north east. The specimen looks authentic and has a layer of soot characteristic of specimens from that time and place. It is simply labelled 'Bishop Aukland, --07, Wingate.' and there are no other details with the specimen. (CN **24** 2012) Did it come from imported timber, or was it a gift from one dipterist in the south of England to one in the north ? So the locality is as important as the species name and despite the presence of a labelled specimen, the presence of *Ctenophora ornata* in Bishop Aukland has not been accepted.

References

James, T. 2006 'Improving Wildlife data quality: guidance on data verification, validation and their application in biological recording. National Biodiversity Network

John Kramer

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Any suggestions for amendments to the book can be made to the author, Alan Stubbs, Pete Boardman or to John Kramer.

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