

ACRIDOLOGY

INSECT PATHOLOGY MANUAL

Section VI





CONTENTS

1. BASIC KIT FOR COLLECTING GRASSHOPPERS	5
2. COLLECTION OF LOCUSTS AND GRASSHOPPERS	5
Collecting grasshoppers	5
Killing grasshoppers - for a reference collection or identification only	5
Field storage of specimens	6
Pinning out grasshoppers	6
Reference collections	6
3. CLASSIFICATION OF LOCUSTS AND GRASSHOPPERS	6
<i>Zoological nomenclature</i>	6
4. GENERAL MORPHOLOGY	7
Head	8
Thorax	10
Legs	12
Wings	14
Abdomen	16
5. HOW TO ASSESS POPULATION DENSITY	18
<i>Introduction</i>	18
<i>Transect counts</i>	18
<i>Imaginary quadrat counts</i>	18
<i>Fixed quadrat and ring counts</i>	18
<i>Cage counts</i>	18
<i>Counting from a vehicle</i>	19
<i>Mean distance between individuals</i>	19
<i>Nymphs</i>	19
<i>Clumps of vegetation</i>	19
6. REARING GRASSHOPPERS AND LOCUSTS	19
<i>Introduction</i>	19
<i>Problems</i>	19
<i>Facilities</i>	19
<i>Feeding</i>	20
<i>Food preparation</i>	20
<i>Temperature</i>	20
<i>Humidity</i>	20
Stages of development	20
<i>Eggs</i>	20
<i>Nymphs</i>	21
<i>Adults</i>	21
<i>Upkeep</i>	21
7. NATURAL ENEMIES OF LOCUSTS AND GRASSHOPPERS	21
Assessing the impact of natural enemies on locusts and grasshoppers	22
<i>Eggs</i>	22
<i>Nymphs and adults</i>	22



1. BASIC KIT FOR COLLECTING GRASSHOPPERS

You must have

SWEEP NETS • must be strong and easily repaired

FIELD COLLECTION CAGES • easy to carry with one hand, to put insects in, as collected

TUBES

SMALL BOXES

PLASTIC BAGS

PAPER ENVELOPES

KILLING JARS • plastic, with plaster of paris in the bottom and sealing lid

LARGE CAGES • for transporting insects from the field to the laboratory

INSECT PINS

PINNING BOX

ENTOMOLOGICAL PINS

NAPHTHALENE

PARADICHLORBENZENE

SILICA GEL

LABELS

STORAGE BOX

TALLY COUNTERS

SQUARE OR ROUND QUADRATS

2. COLLECTION OF LOCUSTS AND GRASSHOPPERS

COLLECTING GRASSHOPPERS

By hand:

This method for catching large, slow moving insects is simple but has the following drawbacks:

- time consuming
- insects can easily be damaged
- must be done in the early morning

By net:

Good catching skills with nets come only with practice. This is the best way to catch insects in flight or from vegetation.

- **Ordinary net** for catching insects in flight; made of light, transparent fabric. This can damage insects when wet.
- **Sweep net** for sweeping through vegetation; made of stout fabric which will not easily tear.

Vacuum method:

Using a D-vac. This method is useful for quantitative sampling, but the equipment is expensive.

Light trap:

This only works well if the light source, location and phase of the moon are right.

- Can attract some grasshoppers.
- Some traps catch the insects live and others kill the insects with insecticide inside the trap.

KILLING GRASSHOPPERS - for a reference collection or identification only

Use a cyanide killing jar with:

- unbreakable plastic
- plaster of paris in the base
- cyanide capsule
- foam sealing pad
- screw lid or cork bung

N.B. DO NOT use a killing jar for insects which will be used for pathology!!

- 1 Put the cyanide capsule in the cavity in the killing jar
- 2 Add a few drops of water to the jar to allow cyanide gas to escape.
- 3 The insect dies very quickly (5-10 minutes).

DO NOT inhale the gas

DO NOT leave open

DO NOT leave in the sun.

N.B. If you cannot get cyanide, use another insecticide.

DO NOT use ethyl acetate or chloroform which can change the colour of the insect tegument.

DO NOT use alcohol, because it can discolour the insect.

If you cannot get cyanide, use another insecticide.

Failing these, plunge the insect into boiling water for a few seconds and dry it in the sun.

Freeze the insect for 30 mins.

FIELD STORAGE OF SPECIMENS

If you can't prepare the specimens you have collected immediately, preserve them temporarily so that they do not deteriorate. Grasshoppers can be kept for a few days in a refrigerator in a sealed jar and then pinned out afterwards. Alternatively, they can simply be put in paper envelopes and kept dry. They will need to be moistened before you can pin them out.

PINNING OUT GRASSHOPPERS

Pin out your grasshoppers as soon as you can after killing the insects.

- 1 **Evisceration:** It is essential to eviscerate large species and recommended for small species. Fill the cavity with cotton wool or filter paper and boric acid.
- 2 **Softening:** Put some damp sand into a hermetically sealed container with (dried naphthalene or carbolic acid to prevent the development of mould. specimens). Leave 1-2 days. The process can be speeded up by putting the container over boiling water.
- 3 **Pinning out:** Spread out the legs of the insect symmetrically. Open out the wings on one side only.

4 **Drying:** Leave the pinned specimens to dry for 1-2 weeks in an air-conditioned room or dry place. Protect from ants.

5 **Labelling:** Name of collector; Date collected; Place collected. Place the label on the body pin, under the insect.

REFERENCE COLLECTIONS

- 1 Identify your grasshopper using a standard text-book such as *Les acridiens des formations herbeuses d'Afrique de l'Ouest*. by J. Mestre, or by referring to an expert or another reference collection.
- 2 Use a glass topped box or entomological cabinet containing paradichlorobenzene or dichlorvos in a glass or cardboard container.
- 3 Arrange the insects systematically with the label under the insect.
- 4 Pin the insects to the box securely.
- 5 Always preserve at least one male and one female of each species, preferably a series.
- 6 Store the boxes in an air conditioned room, or with silica gel or in a dry cabinet with light bulbs.

3. CLASSIFICATION OF LOCUSTS AND GRASSHOPPERS

A brief summary of a general classification for orthoptera:

ORTHOPTERA

ENSIFERAE

(crickets and bush crickets)

CAELIFERAE

TRIDACTYLOIDEA, TETRIGOIDEA,
EUMASTACOIDEA, ACRIDOIDEA

EUMASTACOIDEA

Families: **CHOROTYPIDAE** (1 rare forest species)

THERICLEIDAE (2 forest species)

EUSCHMIDTIIDAE (4 mainly forest species)

ACRIDOIDEA

Families: **PAMPHAGIDAE** (many species in North Africa; 1 species south of the Sahara)

PYRGOMORPHIDAE (about 25 species in West Africa)

ACRIDIDAE over 300 species in 13 sub-families: *Hemiacridinae*
Tropidopolinae

Oxyinae
Coptacridinae
Calliptaminae
Euryphiminae
Eyrepocnemidinae
Catantopinae
Acridinae
Oedipodinae
Truxalinae
Gomphocerinae

ZOOLOGICAL NOMENCLATURE

Living species must be named using precise rules. There is an International Code for Zoological Nomenclature which sets out rules and recommendations for scientific names. If you have any problems refer to this book.

Basic principles

- 1 A species name has two parts.
- 2 The first part is the name of the genus, which always begins with an upper case letter.
- 3 The second part is the species name beginning with a lower case letter.
- 4 A third part may be added to denote a sub-species. This also begins with a lower case letter.

5 Recommended practice is to follow the species or sub-species name with that of the author of the original description and the date of publication of the description.

example:

Acorypha diplectica JAGO, 1967

Chrotogonus senegalensis KRAUSS, 1877

Chrotogonus senegalensis brevipennis KEVAN, 1959

The original name can, be subject to changes due to the development of taxonomic research. In this case, the author's name is given in brackets.

6 All parts of a species name are written in italics or underlined.

7 Sub-families are written in normal script, beginning with an upper case letter and ending in "-inae".

example:

Catantopinae

Oedipodinae

8 Names of families start with an upper-case letter and end in "-idae".

example:

Pyrgomorphidae

9 Superfamilies start with an upper-case letter and end in "-oidea".

example:

Acridoidea

4. GENERAL MORPHOLOGY

Morphological terminology is very variable depending on the author. We have tried to use terminology which is clear and simple.

Body

consists of three parts or tagma: head, thorax and abdomen.

Head

has the main sensory organs: eyes, antennae and mouth parts.

Thorax

is subdivided into the prothorax, mesothorax and metathorax.

Prothorax

has the pronotum (dorsal part covering the sides of the

body) and front pair of legs.

Mesothorax

supports the middle pair of legs and the elytra. The metathorax supports the hind legs and the wings.

Back legs

of locusts and grasshoppers, like other Orthoptera, are very well developed, for jumping.

Abdomen

has the male or female genital organs at the posterior end and this allows easy identification of the sexes.

To precisely identify and classify specimens you will need a more detailed knowledge of morphology.



HEAD

The head can be roughly divided into two parts: ventral, comprising mouth parts articulated on the dorsal part. The cephalic capsule is dorsally constructed from the vertex continuing laterally with the cheeks which are separated from the front part of the head by the sub-ocular suture.

The front has a projecting median band, the frontal side varies in form and can be flat or shaped like a runnel, with parallel keels or not (*Figure 6.1a; 6.1b; 6.1c*). The anterior part of the vertex is the fastigium, defined by the interocular space to the rear (shortest distance between the eyes) and in front by the upper edge of the foveola (if present). These temporal foveola (fastigial foveola) are small depressions between the front of the head and the fastigium which can be present in various forms or absent (*Figure 6.1a; 6.1b; 6.1c*). If absent, the vertex may be continuous until it reaches the frontal side.

The vertex and its fastigium may also have a median carinule (*Figure 6.1d*) or lateral carinules. *Pamphagidae* and *Pyrgomorphidae* are characterised by the presence on

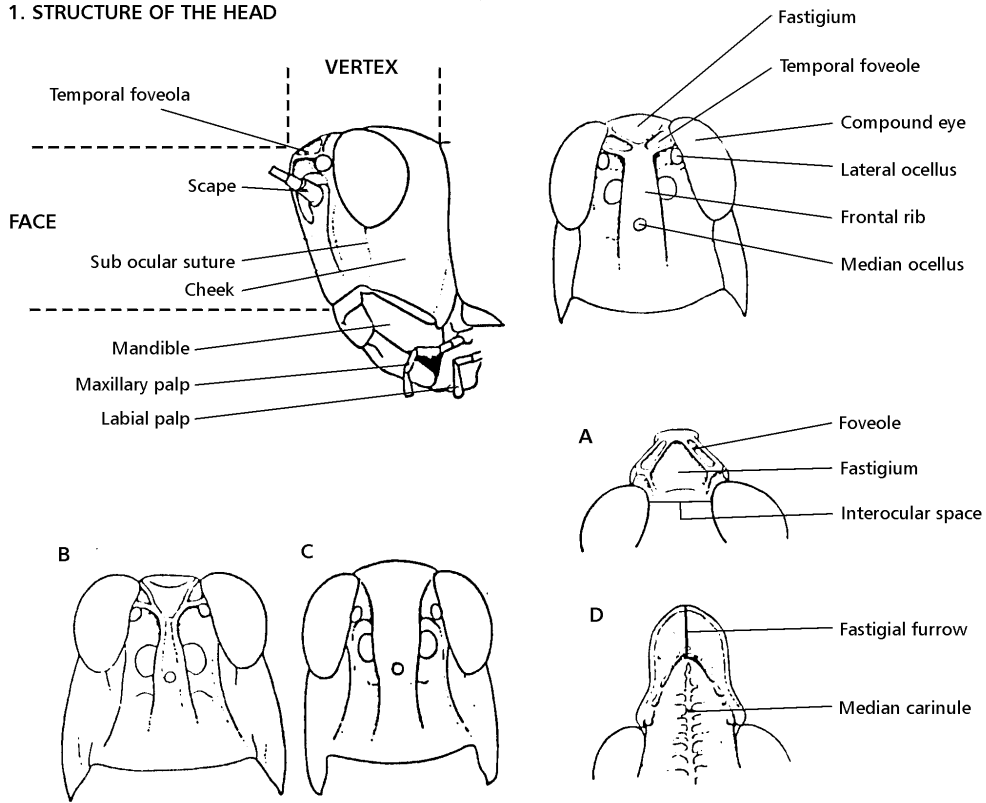
the fastigium of a longitudinal median furrow, more or less pronounced, the fastigial furrow. (*Figure 6.1d*).

From the side, the head may look as though it has a vertical side compared with the body axis (straight profile), rounded, or oblique with a more or less pronounced angle compared with the vertex (conical profile) (*Figure 6.2*). There are many variations between these two extremes. The eyes vary in shape, oval, round, flat or globose. In live insects one can see the coloration and especially whether there are ocular striae, thin longitudinal pigmented bands giving a striped look to the eye.

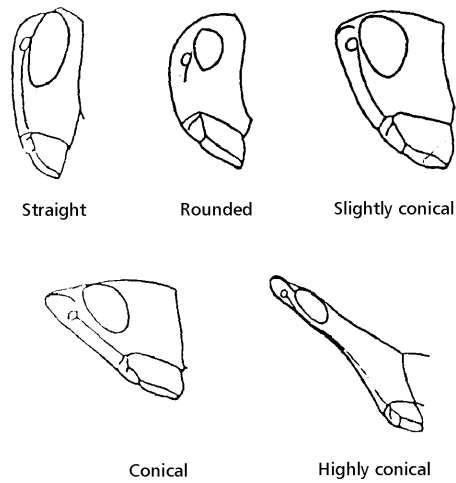
The antennae are in three parts: a basal articulation called the scape, which moves the antenna on the head; the second called the peduncle and the remaining articulations comprising the flagellum. If the articulations of the flagellum are nearly identical and cylindrical this is a filiform antenna; if the articulations are flatt and wide this is an ensiform antenna (*Figure 6.3*).

Figure 6.1

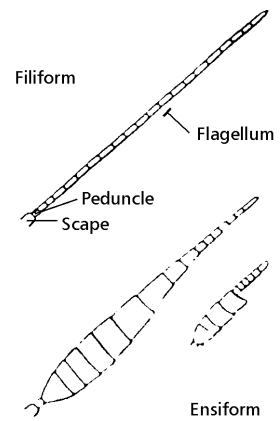
1. STRUCTURE OF THE HEAD



2. EXAMPLES OF PROFILES



3. TYPES OF ANTENNAE



THORAX

The most important part of the thorax for identification is the prothorax, due to the variable shape of the pronotum and of the presence or absence of specific ventral structures.

The pronotum has a dorsal face (pronotal shield) with a median keel and bordered by two lateral keels which divide the pronotum from the lateral faces (lateral lobes) (Figure 6.1). There are four lateral furrows but only three are dorsally visible and the most posterior, called the typical furrow, divides the pronotum into an anterior part - the prozone, and a posterior part, the metazone (Figure 6.1b).

Generally the shape of the dorsal face may be flat, rounded or cylindrical, saddle shaped (selliform) or roof shaped with a prominent median keel (tectiform). Note the disposition of the lateral furrows, if present

(Figure 6.2) and the posterior edge (Figure 6.3).

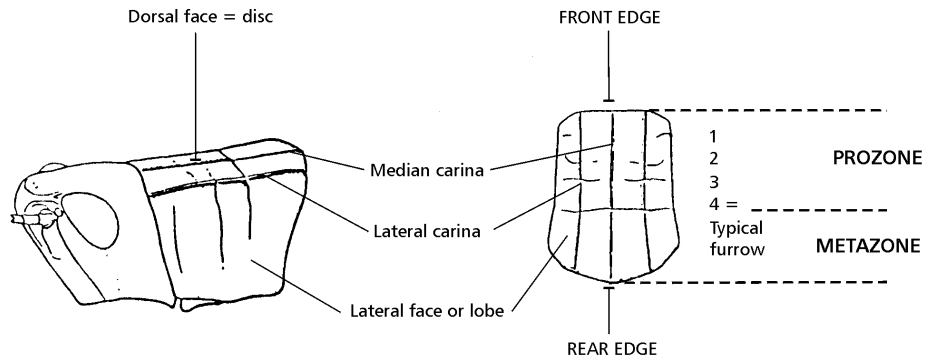
On the ventral face look between the base of the front legs and see if there is a growth which appears in various forms: this is the tubercle or prosterna processus (Figure 6.5a). This can be conical, cylindrical, quadrangular, with a rounded, pointed, flat or bilobial apex. Some species (*Chrotogonus*, *Tenuitarsus*) have a prosternal neckpiece (Figure 6.5b), a laminal growth more or less covering the posterior part of the mouth parts.

There is little of interest in the mesothorax and metathorax.

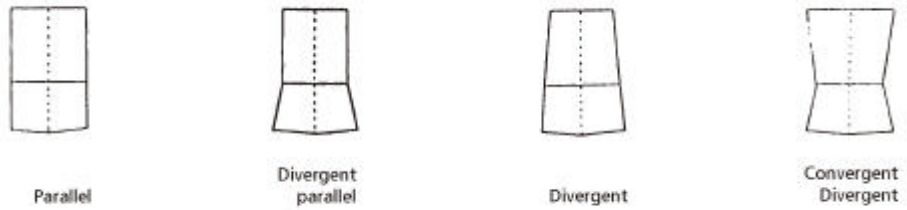
Check the central face and note the shape of the internal angle of the mesosternal lobes and whether the mesosternal space is open (Figure 6.4a) or closed (Figure 6.4b).

Figure 6.2

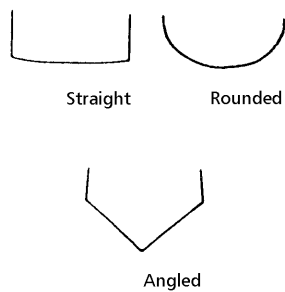
1. GENERAL STRUCTURE OF THE PRONOTUM



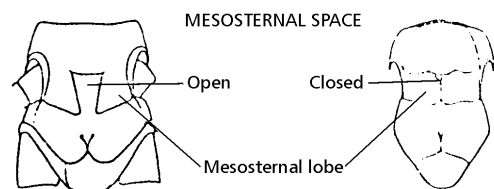
2. EXAMPLES OF THE PLACING OF LATERAL CARINAE



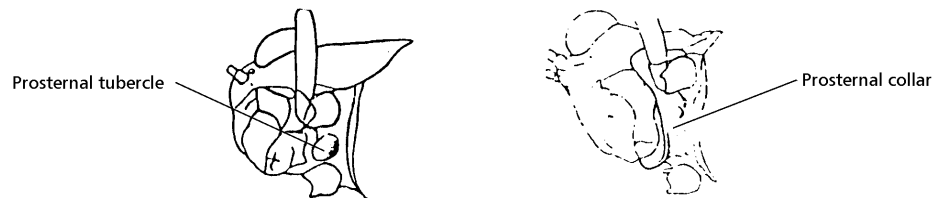
3. REAR EDGE OF THE PRONOTUM



4. PROSTERNUM



5. SPECIFIC PROSTERNAL STRUCTURES



LEGS

The legs can be divided into five parts : coxa (articulated on the thorax), trochanter, femur, tibia and tarsi (three articulations) (*Figure 6.3*). The anterior and median legs hold little interest for identification purposes. The back femur has an external and an internal face (turned towards the body), these two faces are joined dorsally and ventrally at the level of the upper and lower keels (*Figure 6.1*). On each face, keels and carinules outline the upper median and lower areas (*Figure 6.1*)

In the medio-internal portion there is also an internal crest which may have small tubercles or seta making the stridulatory apparatus, stridulation being produced by rubbing the internal crest over the ridges of the elytra.

The geniculate lobes of the posterior femurs are

generally rounded but may be pointed.

There are four faces on the posterior tibia (*Figure 6.5*). The posterior face has two fixed rows of spines on the external and internal sides, which are very variable in size and in number.

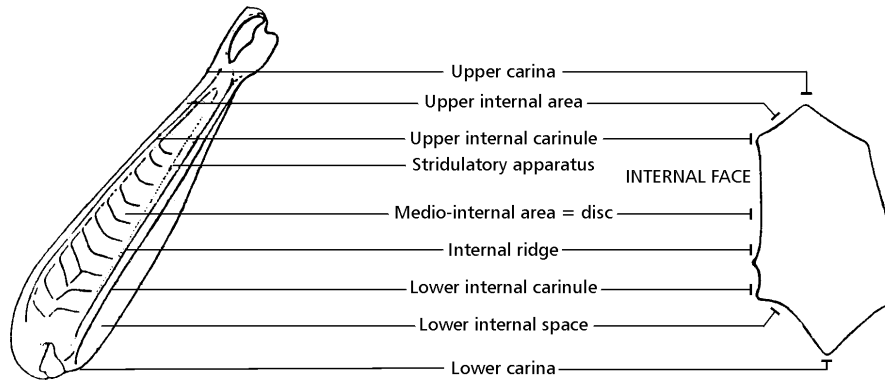
Check the apex of the tibia where it joins the tarsus. On the external and internal sides there may be mobile spurs, which can be quite large in some species.

Look to see whether there is an external apical spine (you may need a magnifying glass).

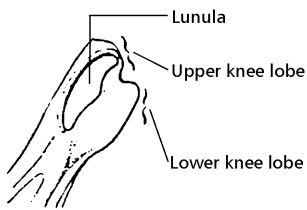
Tarsi have no special characteristics. They are in three parts: basal, median and apical. At the extremity there are two claws surrounding the arolium which are different sizes depending on the species.

Figure 6.3

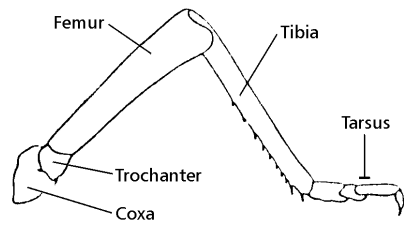
1. INTERNAL VIEW AND TRANSVERSAL CROSS SECTION OF A REAR FEMUR



2. REAR KNEE INSIDE VIEW

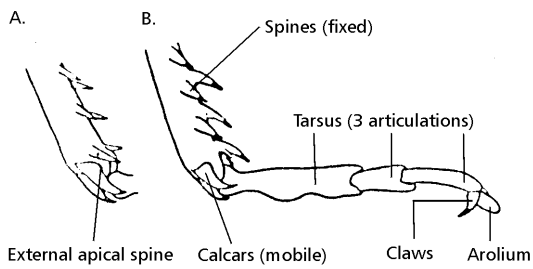


3. MIDDLE LEG FROM THE OUTSIDE

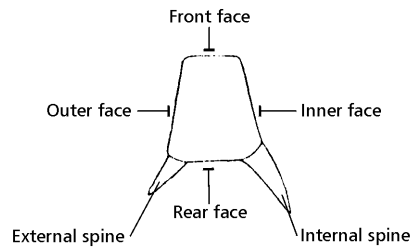


4. APEX OF THE TIBIA AND TARSUS OF A REAR LEG FROM THE OUTSIDE

- A. Typical spine present
- B. Apical spine absent



5. SECTION THROUGH A REAR TIBIA



WINGS

Typically grasshoppers and locusts have two pairs of wings: the anterior wings are narrow and hard; the elytra, which are carried on the mesothorax and when the insect is at rest, cover the posterior wings which are carried by the metathorax.

Grasshoppers can be described as micropterous when the elytra are reduced to two small lobes which do not meet dorsally; brachypterous when they have shortened wings which do not reach the end of the abdomen but do meet dorsally; macropterous when the wings are well developed and they can fly efficiently.

In some species such as *Zonocerus variegatus* and *Hieroglyphus daganensis*, both brachypterous and micropterous forms may be found.

The important characteristics of the elytra are linked to the placing or role of specific veins. *Figure 6.4 1.* gives the terminology for veins and the surfaces of the wings

defined by them. Check also the shape of the apex of the elytra: sharp, rounded or truncated.

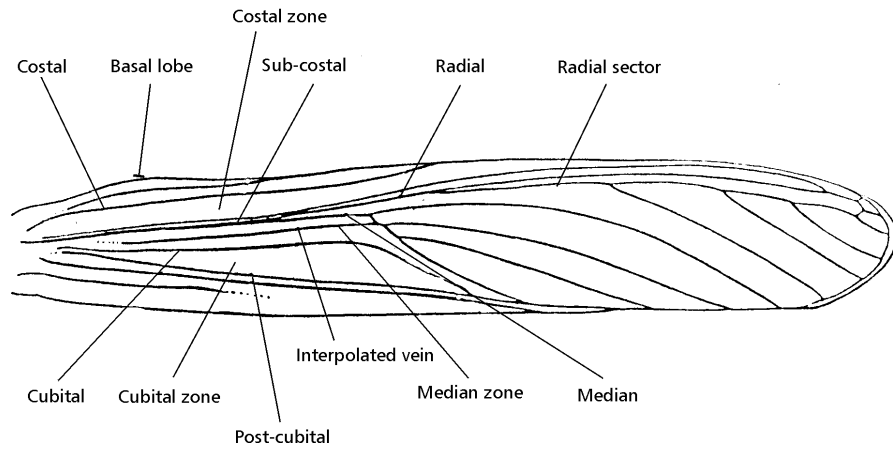
The posterior wings are not morphologically very interesting apart from the presence of a speculum (*Figure 6.4 2.*) corresponding to an enlarged anterior zone.

Colouring however can often be important; overall colour, presence or absence of a fascicule (well defined brown-black band or mark) or total or partial darkening. These colour characteristics are often very constant and specific but do not forget that in adults which have just moulted these peculiarities may not be pronounced, in the same way as in aging they may intensify and darken.

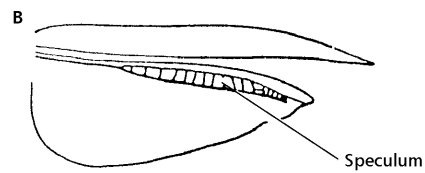
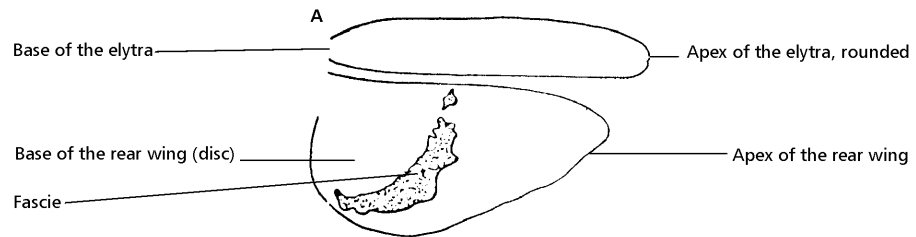
See specific texts if you need more details on this subject.

Figure 6.4

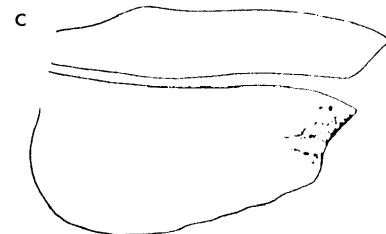
1. ELYTRA OF AN OEDIPODINAE



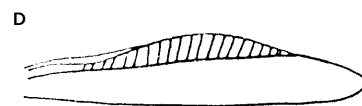
2. SOME SPECIAL CASES



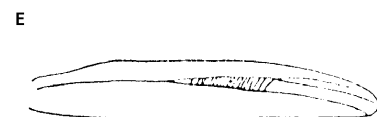
Elytra with pointed apex and speculum.



Wing and elytra truncated and apex of the wing slightly clouded.



Elytra with dilated costal zone.



Elytra with stridulatory veins in the radial zone



ABDOMEN

Most of the abdomen is of no particular morphological interest and we will confine ourselves to the extremity of the abdomen which helps to differentiate between the sexes, and in the males gives a group of characteristics which can be very useful in identifying the insect.

The abdominal extremity of a male locust or grasshopper can be recognised by the shape of the posterior part of the 9th sternite, usually in the shape of a ship's prow (or a clog) which makes up the sub-genital shield and is often very characteristic (*Figure 6.5*). It is often long, becoming conical or flattened like a knife blade (*Figure 6.5 3*). On the dorsal face, the anal

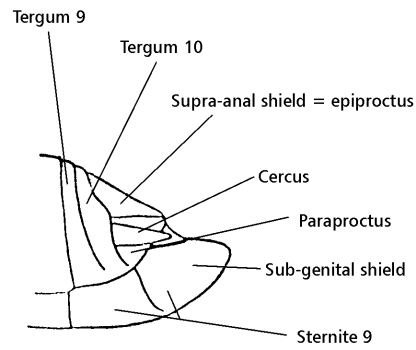
orifice is bordered by paraproctes and dorsally by the supra-anal shield (or epiprocte).

The cerci, small lateral appendages very variable depending on the species (*Figure 6.5 2*) are, together with the sub-genital shield, the most interesting structures morphologically for taxonomy.

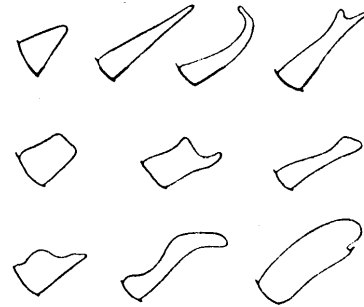
Females can be identified by ventral or inferior valves and dorsal or superior valves (*Figure 6.5 4*). These are used to dig holes in the ground in which to place the eggs. The valves are often very hard and may have small teeth. However, the abdominal extremity in a female has a very homogenous structure and is rarely of much use for identification purposes.

Figure 6.5

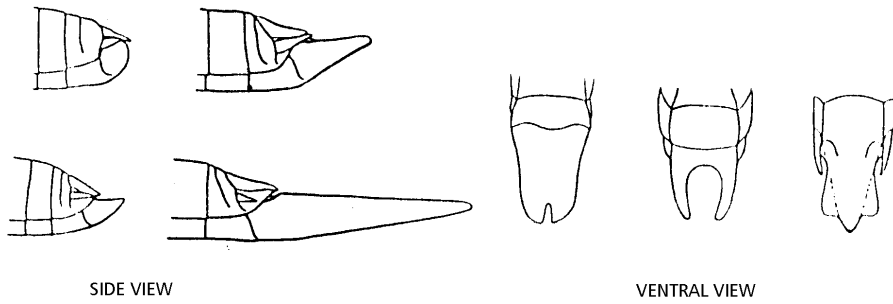
1. ABDOMINAL EXTREMITY OF A MALE



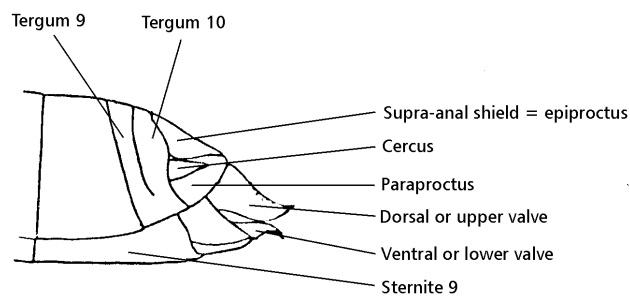
2. EXAMPLES OF MALE CERCI



3. EXAMPLES OF MALE CERCI



4. FEMALE ABDOMINAL EXTREMITY



5. HOW TO ASSESS POPULATION DENSITY

INTRODUCTION

We need to assess population densities of grasshoppers and locusts

- to decide whether to control the insects
- for experimental purposes, to assess the effectiveness of control operations.

It is difficult to assess the density of small populations of grasshoppers. Methods of assessment vary.

Factors which cause variation in population counts

- the operator
- the time of day
- the type of vegetation
- the developmental stage of the insects
- the grasshopper species present
- the degree of aggregation.

If there is too much variation in the counts, the data will be useless.

Assess the quality of your data by taking many counts and calculating the mean and standard deviation. If the deviation is greater than the mean, this means that the insects are aggregated and you must repeat the observations with a greater number of counts.

Use a hand held tally counter for more rapid and reliable counting.

TRANSECT COUNTS

Use this method in difficult terrain, soft sand, cultivated fields and other places where you cannot drive a vehicle. It is useful where there are low densities (<1 per m²) or where vegetation is very dense and when most of the insects to be counted are adult.

- 1 Walk along a 100 m transect, counting the insects which fly up from the line.
- 2 Use a stick to beat the vegetation for a metre on either side of you as you walk. This gives a two (2) metre band.
- 3 Count the insects you see in this band.
- 4 More accurate results can be obtained by putting observers at two (2) metre intervals along the transect.
- 5 **To calculate the population density per hectare:**

$$D = \frac{100G}{2N}$$

where

D = density per hectare

G = mean number of insects over 100m

N = number of observers.

N.B. You can count the flying grasshoppers separately from the nymphs by using a tally counter in each hand.

This method is useful for *Cyrtacanthacris tatarica*, *Diabolocantops axillaris*, *Ornithacris cavroisi*, *Kraussaria angulifera*, *Acorypha glaucopsis* and *Krausella amabile*

IMAGINARY QUADRAT COUNTS

- 1 Use an imaginary quadrat of 1m square starting 5 m from where you are standing.
- 2 Move towards the quadrat.
- 3 Count the insects which jump from the quadrat.
- 4 Stand over the quadrat to count nymphs.
- 5 Use a minimum of 25 quadrats to get a good estimate of populations.
- 6 To calculate the population density per hectare:

$$D = I / ha = \frac{T \times 10.000}{25}$$

where

D = density per hectare

I = individuals

T = total insects from 25m²

N.B. This density includes all species = the total density. If you want to determine the density of particular species you will have to take an appropriate sample of insects and work out the population structure from this sample.

You can use an imaginary quadrat count as a fixed quadrat by marking one corner with a peg or stick.

This method is useful when there are young or nymphal stages and adults which do not move very much - such as *Zonocerus variegatus*, *Pyrgomorpha cognata*, *Oedaleus senegalensis* and *Morphacris fasciata*.

FIXED QUADRAT AND RING COUNTS

Population counts will vary greatly with the kind of vegetation found in the quadrat.

For field trials or to follow population changes through time, use fixed quadrats, marked with pegs or rings.

See section on analysis of population counts. The calculation will depend on the size of the quadrat.

CAGE COUNTS

When the grasshopper population consists mainly of nymphs which do not leave the quadrat as you approach, you can use a cage quadrat to obtain accurate counts. The cage is a wooden frame with mesh on four sides, leaving the top and bottom open.

- 1 Place the cage over the vegetation
- 2 Remove the insects inside the cage
- 3 Count the insects.

This method gives accurate results.

COUNTING FROM A VEHICLE

- Use this method wherever a vehicle can be used.
 - Avoid using this method in the heat of the day.
 - DO NOT use this method when it is windy.
- 1 Drive at about 5-7 km/hr in a straight line (if possible).
 - 2 Use a mechanical counter to count only insects flying in front of the vehicle in a band as wide as the front of the vehicle.
 - 3 DO NOT count insects flying at the sides.
 - 4 Every 100 m note the number of adult insects flying.
 - 5 Do at least 10 transects.
 - 6 **To calculate the density:**

Measure the width of the vehicle (1.20m for a Land Rover).

$$D = \frac{100G}{L}$$

where

D = density per hectare

G = mean of numbers counted over several 100 m transects

L = width of the vehicle

MEAN DISTANCE BETWEEN INDIVIDUALS

This is a rapid method for estimating population density.

- 1 Estimate the mean distance between individuals over 20 pairs of undisturbed individuals.

- 2 The MDI is given in metres to get the density per hectare (D):

$$D = \frac{100x2}{MDI}$$

Workers must learn to estimate distance as quickly as possible. However, it is difficult to obtain accurate estimates of population density using this method.

NYMPHS

In general, nymphs can either be counted using one of the quadrat methods mentioned above, or using the following method:

CLUMPS OF VEGETATION

This method is time consuming, but may be done at any time of day.

- 1 Note the species of plants on which nymphs are found.
- 2 For each plant species count the nymphs in random clumps.
- 3 Some clumps will be empty (0 individuals).
- 4 Stop counting when you have found 25 clumps containing larvae.
- 5 To calculate the mean number of nymphs in each clump of each species: add up the total number of nymphs found in the 25 clumps, then divide by the total number of clumps examined (including empty ones)

Use the MDI method to assess the mean number of clumps per hectare for each species of vegetation.

It is easy to calculate the mean number of nymphs per hectare by adding the number of nymphs per hectare for each plant species.

6. REARING GRASSHOPPERS AND LOCUSTS

INTRODUCTION

We need to rear locusts and grasshoppers because:

- 1 Laboratory reared insects are free of disease, whereas insects from the field are often contaminated with different pathogens.
- 2 A good supply of insects can be maintained all year round without being dependent on field populations.
- 3 It is possible to carry out experiments on insects which can normally only be found in remote places.

If you experience problems with rearing, or want to work with different species, you can use field-caught insects for experiments, provided you keep the insects for a few weeks before using them, in order to see whether they are carrying any disease.

PROBLEMS

There are two main problems in rearing locusts and grasshoppers.

- 1 The appearance of diseases in an insect colony, such as *Malamoeba* and viral infections.
- 2 Some species may lay diapausing* eggs

FACILITIES

- 1 An insect rearing room should have some natural light and ventilation.
- 2 Make space on shelves for insect cages.
- 3 Low humidity is essential for the good health of the insects; this can be maintained in humid climates by using air-conditioners or with light bulbs in the cages, or both.

4 Cages should be about 60 cm square made of wire mesh with a door in one side and a smaller opening inset into the large door.

5 There must be space for a 100 Watt light bulb protected by wire mesh.

6 Install two horizontal dividers in the cage - the higher one of wire and the second in plywood, with two circular holes for putting pots of sand for the insects to lay their eggs in.

Each of these cages can hold 800 nymphs or 400 adults, but it is better to reduce the numbers to about 400 nymphs or 250 imagos in order to reduce mortality during moulting.

FEEDING

Grasshoppers and locusts can be fed on leaves of cassava, maize, millet, sorghum, banana and papaya plants and wheat bran mixed with powdered milk.

All the grasshoppers eat wheat bran and powdered milk, the leaves they eat differ depending on the species.

Schistocerca gregaria : cassava, millet, sorghum, cabbage, cowpeas, groundnuts.

Anacridium melanorhodon: cassava, millet, sorghum.

Ornithacris cavroisi: cassava, millet, sorghum.

Eyprepocnemis plorans: cassava, millet, sorghum.

Locusta migratoria: maize, millet, sorghum.

FOOD PREPARATION

Green leaves

- 1 Pick leaves in the field or from glasshouses.
- 2 Soak in a solution of 2.5% bleach for 5-10 minutes.
- 3 Rinse in running water several times.
- 4 Shake to remove excess water.
- 5 Put the leaves vertically in the cage well away from the light bulb.
- 6 Change the leaves in the cage every day.

Bran, milk powder

- 1 Reduce the bran to a powder.
- 2 Sieve the powder.
- 3 Sterilise in an oven at 200°C for 30 minutes.
- 4 Add 25 g of milk powder to 75 g of bran.
- 5 Put this mixture low down in the cage.

TEMPERATURE

Take into account the number of light bulbs in the rearing room plus the number of fluorescent strip lights.

The ideal temperature of the room is between 32-37°C and that of the cage between 37-39°C. The insects are less active when the temperature is high.

HUMIDITY

The humidity should be kept between 35-65% relative humidity, preferably lower than this. This can be achieved using an air conditioning unit. The insects are less active in a very humid atmosphere.

The lights should be on 14 hours out of 24. This helps to prevent the insects from laying diapausing eggs.

STAGES OF DEVELOPMENT

EGGS

Oviposition

- 1 Wash 1 kg of fine sea or river sand.
- 2 Sieve and sterilise the sand in an oven at 250°C for 30 minutes.
- 3 The sand should have water added in a ratio of 100:15 sand: water by volume to achieve a humidity of 70 - 80% for optimum oviposition.
- 4 Remove and replace the container when it is full of eggs.

Incubation

The length of incubation depends on the species:

<i>Schistocerca gregaria</i>	14 days
<i>Locusta migratoria</i>	12 days
<i>Anacridium melanorhodon</i>	1 month
<i>Eyprepocnemis plorans</i>	1 month
<i>Zonocerus variegatus</i>	3 months

Under normal conditions of humidity and temperature. If the eggs are washed, incubation can take three to four days longer.

Sorting and washing eggs

Eggs are washed to avoid the development of protozoa, bacteria or fungi. This gives a successful hatching rate of 85-95% nymphs or washed eggs as opposed to a rate of 70-85% nymphs for unwashed.

Eggs collected daily

- 1 Wash the eggs two or three days before hatching.
- 2 Wash eggs in a solution of 10% bleach for 5 minutes.
- 3 Rinse several times.
- 4 Place on a layer of paper towel in plastic boxes.
- 5 Pierce the lid of the box.
- 6 Place the box out of the reach of predators.
- 7 Regulate humidity and temperature.

Eggs collected every five days or weekly

- 1 Sort eggs first according to age
- 2 Group according to age.
- 3 Look after them properly so as to allow

development of the embryos to take place until the eggs are washed.

- 4 Wash the eggs 2-3 days before hatching.
- 5 Wash eggs in a solution of 10% bleach for 5 minutes.
- 6 Rinse several times.
- 7 Place on a layer of paper towel in plastic boxes.
- 8 Pierce the lid of the box.
- 9 Place the box out of the reach of predators.
- 10 Regulate humidity and temperature.

NYMPHS

Duration of nymphal stages:

<i>Schistocerca gregaria</i>	18-28 days
<i>Locusta migratoria</i>	18-22 days
<i>Anacridium melanorhodon</i>	25-34 days
<i>Eyprepocnemis plorans</i>	25-34 days
<i>Ornithacris cavroisi</i>	25-34 days
<i>Zonocerus variegatus</i>	3 months and 2 weeks

From the fifth nymphal stage to imago takes about one (1) week, but other stages are shorter.

ADULTS

Adult insects become sexually mature several weeks after the final moult and live for several months according to the species:

<i>Schistocerca gregaria</i>	3-8 months
<i>Locusta migratoria</i>	2-3 months
<i>Anacridium melanorhodon</i>	6-10 months
<i>Eyprepocnemis plorans</i>	4-5 months
<i>Ornithacris cavroisi</i>	6-10 months
<i>Zonocerus variegatus</i>	4-6 months

The life span of insects may be reduced by:

- poor climatic conditions in the cages
- unsuitable food

- accidental damage (during moulting, careless handling, cannibalism)
- predators (ants)
- parasites (nematodes)
- disease (protozoa, fungi, bacteria)

After several oviposition cycles the weight of the insects drops progressively and consumption of food is reduced.

Individuals hatched from washed eggs tend to have a longer life span than those from unwashed eggs.

UPKEEP

Personnel

Workers must wear a lab coat, face mask, hat and gloves.

This clothing should not be worn outside the rearing room and should NEVER enter the rooms where pathogens are cultured.

There should be as few visitors as possible and all objects entering the room must be carefully dealt with so as to avoid contamination.

Workers should respect basic hygiene and disinfect their hands after working with the insects.

Cages

- 1 Clean cages every day using a vacuum cleaner to remove frass.
- 2 Remove large leaves etc. by hand.
- 3 Wipe the surfaces of the cage with a sponge soaked in 10% bleach solution.
- 4 Remove the cages every few weeks and replace with fresh cages.
- 5 Soak the used ones for 20 minutes in a 10% bleach solution or a solution containing hydrochloric acid.

EVERYTHING

SHELVES, TOOLS, WALLS ETC.

MUST BE DISINFECTED REGULARLY

7. NATURAL ENEMIES OF LOCUSTS AND GRASSHOPPERS

Locusts and grasshoppers have many varied natural enemies including predators, parasitoids and pathogens. Records of natural enemies go back as far as the turn of the century and much work on the subject has been done in Africa and in North America.

Different organisms attack different stages of locusts and grasshoppers. Predators of eggs include species of Diptera and Coleoptera. The only true parasitoids of

eggs are from the genus *Scelio sensu lato* and they will attack any species of Acridoidea.

Predators of nymphs and adults include many vertebrates and invertebrates especially when there are locust swarms. Small mammals and lizards are widely reported as predators, but do not seem to have a significant impact. However, predation by birds can be significant especially on small hopper bands and

include passerines, hornbills and rollers and migrating storks. Reptiles, amphibians, scorpions, spiders, praying mantids and beetles may all predate on locusts and grasshoppers but there is no proof that they have any impact on populations. There are several species of Diptera which act as parasitoids to the nymphal and adult stages of locusts and grasshoppers, while mermithid nematodes are well known as parasites. Locusts and grasshoppers can be affected by range of pathogens including protozoa (*Nosema*, *Malamoeba*), bacteria and viruses. However, the most important pathogens affecting locusts and grasshoppers (and the most widely reported) are fungi. See Section 2 Fungi.

Natural enemies can occasionally provide a constraint on the rate of increase of locust or grasshopper populations, but it seems unlikely that they could prevent plagues of locusts from forming. It is most likely that they have most impact on populations which are already declining and so they have some importance.

ASSESSING THE IMPACT OF NATURAL ENEMIES ON LOCUSTS AND GRASSHOPPERS.

EGGS

Abiotic factors:

Hatching is influenced by rainfall.

Predators:

These are found associated with egg pods - larvae of beetles such as Meloidae.

Parasitoids:

Incubate egg pods in the laboratory to check on emerging parasitoids.

Assess egg mortality by periodically taking a sample of

eggs to the laboratory and incubating them. Eggs of species with a dry season diapause often hatch in response to rainfall and wetting the eggs may lead to eclosion of hopper and parasitoids. The impact of parasitoids can be assessed by counting the live hoppers, the parasitoids and the unhatched eggs.

The proportion destroyed by predators can be estimated by keeping any predators found during surveys with egg pods in the laboratory and seeing how many are eaten.

Sources of error:

Once the egg pods are removed from the field as further attack takes place.

Hatching in the field is highly dependant on weather conditions.

NYMPHS AND ADULTS

Abiotic factors and food availability:

Adverse weather and a lack of food can affect the survival of nymphs.

Pathogens:

see Sections 1, 2 and 3.

Parasitoids:

Calliphorid flies such as *Blaesophixa filipjevi* parasitise grasshopper nymphs.

Predators:

Predatory insects (aspid flies, beetles, praying mantids), reptiles and amphibians, birds and mammals all attack locusts and grasshoppers.

The impact of pathogens and parasitoids can be assessed by caging and incubation. The impact of predators is difficult to assess in the field; comparison between caged and uncaged grasshoppers (predator exclusion) provides some clues.