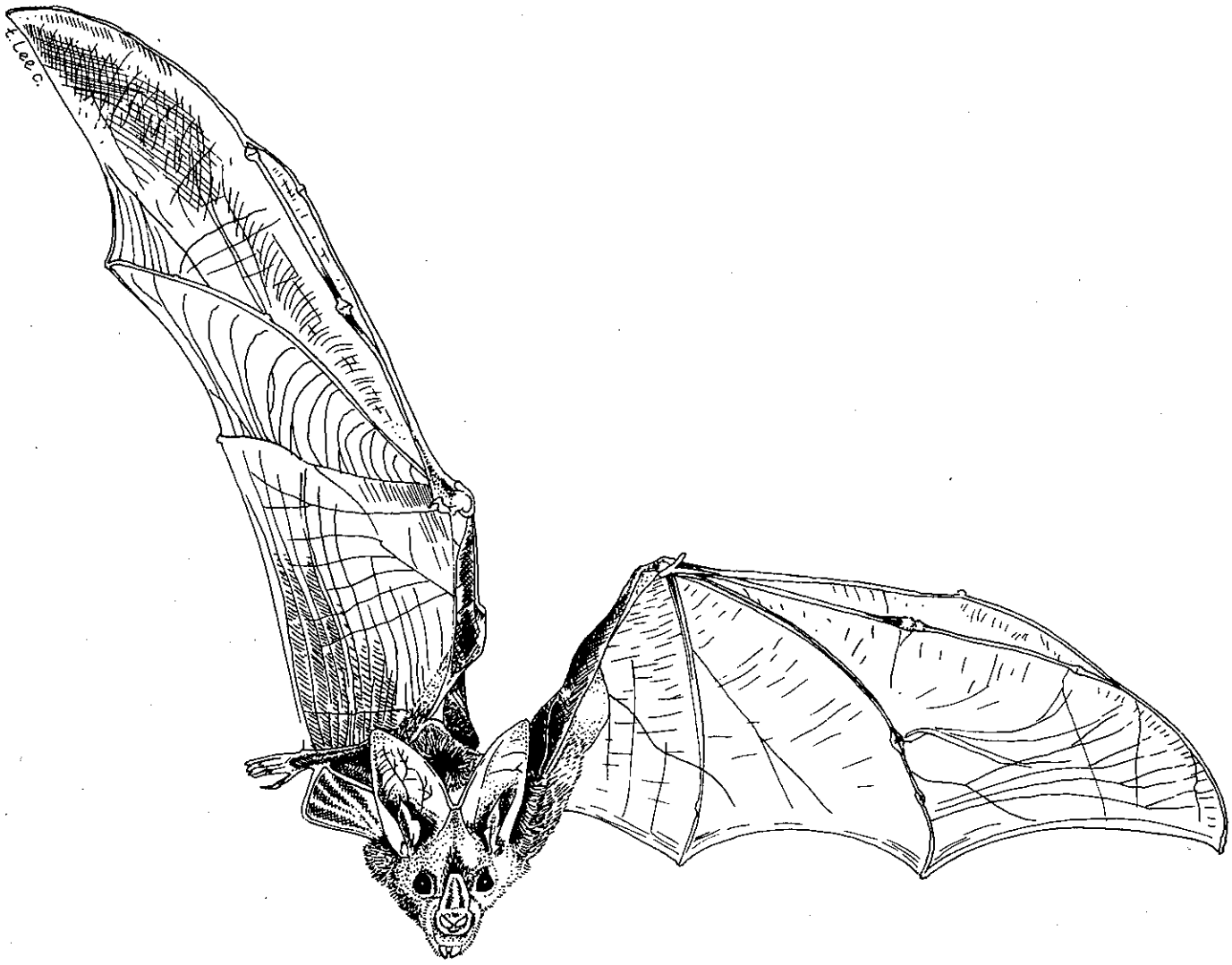
Macroderma

Volume 4 Number 1 March 1988



ISSN 0815-2152

Macroderma Volume 4, Number 1, March 1988

Contents

Macroderma

Macroderma is a journal of bat research in Australasia published twice yearly in March and September by an Editorial Group comprised by:

All correspondence and manuscripts should be addressed to:

Macroderma
P.O. Box 859
Civic Square
ACT 2608

Correspondent: Kunwar Bhatnagar, Editor, Bat Research News,
Department of Anatomy, University of Louisville,
Kentucky 40292, U.S.A.

Printed in Canberra by Union Offset Printers
Typing by Elizabeth Jackson
Cover design by Sue Craven from a drawing by Terry Clayton

P A P E R S

FIXATION-PRESERVATION OF BATS IN THE FIELD

Kunwar P. Bhatnagar

Department of Anatomical Sciences and Neurobiology,
University of Louisville, School of Medicine,
Louisville KY 40292, USA.

Abstract

This note encourages field biologists to collect, prepare, and preserve bat specimens in a manner so as to be maximally useful for future investigators. To achieve superb fixation of cellular detail, intracardiac perfusion procedures are described in addition to other less effective procedures such as stick injection, and immersion techniques. The fallacy of the traditional collecting method, wherein the animal body is simply "dropped" in the fixative, is pointed out. Detailed lists of equipment, chemicals, formulae for solutions, and suggested entries in the logbook are provided.

Introduction

During expeditions and field trips bats are preserved routinely either in freshly made 10% formalin or in 80% ethyl alcohol. Frequently the abdomen is slit for better and quicker penetration of the fixative. While such specimens serve the taxonomists and external morphologists, they are capable of providing only worthless histological detail. This note points out the necessity and usefulness of careful fixation - preservation of bats in the field so as to increase the utility of the specimens for a wider variety of investigative approaches.

A comparative morphologist's dream is to be able to study bats spanning the limits of the taxonomic range including not only widely distributed taxa but even those species which are extremely rare. Only a few choices are available to the investigator for obtaining such study materials. Either he can travel to all those exotic places collecting specimens himself, an unlikely possibility, turn to a museum, or even request specimens from a colleague. Very often, specimens obtained from the last two sources are simply not adequately fixed at least for histological purposes. Well preserved specimens, with minimum expenditure of resources and following simple and easily learned techniques, provide maximum benefit not only to the collector but to several generations of investigators. Animal sacrifice is minimized thereby achieving conservation of species.

Fixation-preservation of animal tissue is carried out either by perfusion (flooding of tissue under pressure) or by immersion. Numerous versions of both methods exist. Only those procedures are discussed here which are readily applicable to field conditions. An appendix at the end provides a list of major equipment, accessories and chemical formulae for making solutions.

Figure 1:

Table set-up for perfusion. A-D, perfusion apparatus consisting of a three-way stopcock to which a Luer-lok syringe at A, the intake tubing at B, the outflow tubing at C, and the injection cannula at D are attached. Bottles marked E and F contain the flushing solution and the fixative respectively. The bat is pinned on corkboard G, with thoraco-abdominal cavities opened and the cannula inserted into the apex (left ventricle) of the heart. Note the use of the table edge as a lever against which the syringe plunger is used to force out fluids. A cloth bag, and a syringe for anaesthesia are nearby.

Figure 2a:

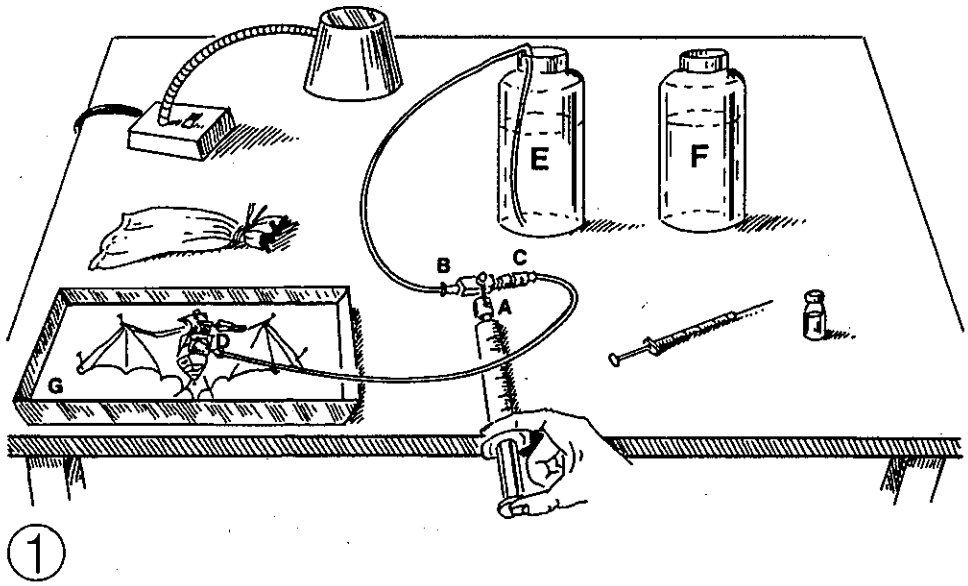
Diagrammatic sketch of the ventral aspect of the animal to be perfused showing the incisions. A-B, vertical incision from the genitals to just below the diaphragm; B-C and B-D, lateral incision on the right and the left abdominal musculature respectively below the diaphragm; C-E and D-F, vertical incisions on the right and left lateral aspect of the rib cage. XP, xiphoid process. See text for further explanation.

Figure 2b:

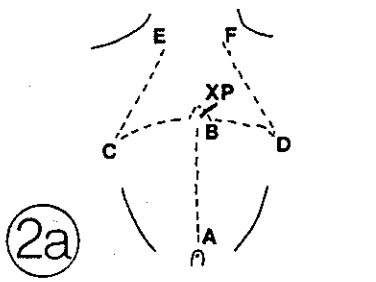
Ventral aspect of the animal after incisions and reflection of flaps.

Figure 3:

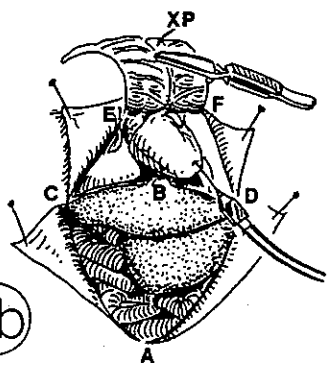
Gravity method for perfusion. The bottles E and F hanging upside down or separatory funnels contain the flushing solution and the fixative respectively. A Y-connector with clamps (c) leads to the tubing to which a cannula is attached. All air-bubbles are first removed from the system by allowing the two solutions and finally the flushing solution to flow through the cannula. After the cannula is placed in the apex, perfusion is started by appropriate loosening of the clamp on bottle A. Other details are similar to the ones described for use with perfusion apparatus. Sometimes only one separatory funnel can also be used first for the flushing solution followed by the fixative.



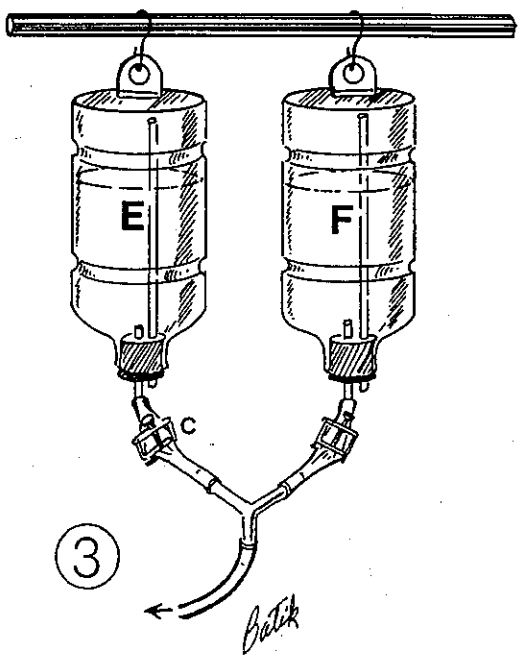
1



2a



2b



3

Batik

Perfusion fixation

Perfusion of the whole body utilizing the intracardiac approach is one of the most valuable procedures for obtaining the highest levels of fixation-preservation. This method is so superior to any other that it is utilized both for light and electron microscopy, and is advantageous for the tissue that requires rapid fixation but is inaccessible for rapid removal. The major advantage is that the blood is replaced by the fixative under pressure which is distributed to all organs and tissues in a uniform manner. The step-by-step procedure is as follows:

- Weigh each animal as soon as possible after capture, record its sex and bag individual separately. Attach a tag which lists weight and sex.

- Check the entire set-up for perfusion by arranging all items on the dissecting table (Fig. 1).

- Make the flushing solution and the fixative; filter, and fill the respective bottles.

- Draw the flushing solution into the syringe, change valve direction and push the solution out through the cannula (injection needle) removing all air-bubbles from the system. Leave the cannula end immersed in a puddle of flushing solution.

- Anaesthetize the animal by injecting it intraperitoneally (i.p., anywhere in the lower abdomen) with appropriate dosage (0.1 ml/10 g or 0.07 mg/g body weight) of diluted Nembutal. Undiluted Nembutal can also be used but one must work fast because the animal may die within minutes after injection. Chloroform and ether are unsuitable and are not advised for use.

- Record measurements after anaesthesia takes effect (5 to 10 minutes; a second dose of anaesthetic might be needed); pin the animal on the corkboard (kept in a dissecting tray). With practice one can work without a corkboard.

- Make a midline ventral abdominal skin incision extending from the genitals to the neck. Reflect and pin the skin.

- Make a V-shaped cut in the midline abdominal musculature extending it vertically from the genitals to immediately under the diaphragm (Figs 2a,b). Extend the abdominal incision to the right (B to C), and the left (B to D) and straight up (C to E and D to F) on both sides. Hold the xiphoid process (XP) in an artery clamp or a haemostat and fold the breast-plate over the neck thus exposing the heart.

- Using fine forceps remove the thin pericardium covering the heart.

- Now gently hold the still beating heart in a blunt pair of forceps and insert the cannula (injection needle) to a depth of some 3-4 mm only straight in the APEX (left ventricle) of the heart. As soon as this is accomplished, make a small incision in the right ventricle for blood to escape. INSERTION OF THE CANNULA and INCISING THE RIGHT VENTRICLE (or right atrium) are the two most critical steps in the procedure.

- Holding the syringe firm against the edge of the table begin perfusion by gentle but continuous pressure on the plunger. Some clear fluid should begin to escape from the cut end of the heart. If necessary, draw more flushing solution into the syringe and continue.

- Transfer the intake tubing to the fixative bottle and fill the syringe. Turn valve and continue perfusion until syringe is empty. The animal responds to the fixative by stretching, a sure sign of proper perfusion. Also check the bat's neck. It should be rigid and not limp as before perfusion. Moving the bat's head and neck from side to side several times and keeping the head straight down helps greatly in the perfusion.

- This next step is CRITICAL and must be followed. REFILL the syringe with fixative and REPEAT the PERFUSION. This precaution assures perfusion of the body with undiluted fixative, since the first run of the fixative was diluted with the flushing solution as it was being replaced. The animal's neck should now be very rigid, if not, check the cannula in the heart and repeat perfusion using fixative.

- Check the liver and the intestines; blood colour should have disappeared. If Bouin's fluid (a superb fixative for general histology) is used, the yellow colouration due to picric acid is reflected in the organs, wing veins, mouth and other regions.

- Remove cannula. Any excess fixative can be returned to the bottle. Clean the perfusion apparatus several times with water in preparation for the next animal.

- Check for pregnancy; if positive, remove the gestation sac, weigh and record. Prepare a tag recording necessary data and attach it to the specimen.

- Ideally the bat should be immersed in the fixative for 1-2 days and then transferred to 80% alcohol. However, appropriately perfused animals can be directly transferred to 80% alcohol.

- Once the initial steps are practiced, it takes 10-15 minutes to perfuse one animal.

- As described above, placement of the needle and incising the right heart are the most critical steps in the entire procedure. The needle should be aimed straight through the apex to a depth of some 3-4 mm only, otherwise it is likely to enter other chambers of the heart and no perfusion will result. The needle must remain in the LEFT VENTRICLE. To avoid incorrect placement of the needle, some workers exteriorize the ascending aorta by blunt dissection, make a V-shaped incision, insert the cannula and clamp it above and below the incision by sutures. This procedure is more delicate for smaller animals such as bats and therefore not highly recommended. Faulty incision in the heart will also result in unsuccessful perfusion.

- It is possible to perfuse selected parts of the body. This is done not only to conserve fixatives (some of which are expensive) but also to ensure greater success with fixation. Major arteries are clamped or ligated at a point beyond which fixation is not desired. For example, to selectively perfuse the head and neck region, a ligature is placed in the descending aorta beyond the origin of the left subclavian artery.

- The procedure described above can be used for all animals (big or small) with appropriate modifications in the cannula size, perfusion apparatus, tubing dimensions and volumes of solutions to be circulated, etc.

- On an average, for a 30-40 g bat, 25 ml of flushing solution and 25 x 2 ml of fixative are sufficient for achieving optimum fixation.

- Intracardiac perfusion can be made utilizing any fixative. Those desirous of further details should consult the selected references provided.

Intracardiac perfusion with other devices

Excellent fixation can also be achieved without a perfusion apparatus. Two of such procedures are described here.

1. *Two-syringe Method*

This procedure requires two Luer-lok syringes and adequate practice of handling. After anaesthetizing the animal, fill the marked syringes, one with flushing solution, the other with fixative. Prepare the animal as described above and when ready insert the needle (attached to the syringe filled with flushing solution) to the appropriate depth in the apex of the heart, incise the right ventricle and flush out all blood. THE NEXT STEP IS DELICATE AND DIFFICULT IN OPERATION! With a steady hand remove only the syringe, leaving the needle in place (use of Luer-lok syringes allows this feat to be accomplished easily). Let the other syringe, filled with fixative, be handed over to you without the needle.

Lock this syringe on the needle and complete the perfusion. Refill and repeat. Follow with other steps as above.

This procedure might appear clumsy, but in expert hands it is accurate, quick and reliable. The author has been following this two-syringe method for over 25 years with excellent results. However, the advantages of using a perfusion apparatus over the two-syringe method are numerous and cannot be over-emphasized.

2. *Gravity Method*

The flushing solution and the fixative are kept in two bottles hanging upside down at a height of approximately 0.5 meters from the animal. Figure 3 illustrates the basic set up. After the cannula is placed in the apex and the right ventricle is incised, perfusion is started by removing the clamp on the flushing solution. Delivery of perfusate depends on the hydrostatic pressure which is increased by gradually raising the bottles to about 1.5-2 meters above the animal. An adjustable clamp (c) controls the flow. Other steps are basically similar to those described. The gravity method is a frequently used procedure, but it takes much longer for perfusion than the other two methods.

Preservation-fixation of bats without perfusion

1. *Stick-injection Method*

In principle, the anaesthetized animal is injected with the fixative and stored in the same fluid. Separate injections are made in the abdomen, right and left ribcage separately, dorsally under the neck, at the base of the brain, in the nostrils, into the mouth etc. The bat is then immersed in the fixative.

While some organs get poorly fixed by this procedure, brain and other parts of the central nervous system and organs of special sense are not fixed. Overall, the quality of fixation is very poor, yet decidedly better than the method which follows. Stick injection is recommended only when intracardiac perfusion is either not possible or is impracticable.

2. *Immersion Fixation*

After slitting the abdomen of the anaesthetized animal, it is "dropped" into the fixative and considered fixed and preserved for a long time to come! One can imagine the quality of fixation in this case compared to all others described above. Unfortunately, this type of immersion fixation has been the age old and traditional method for collecting and storing specimens.

Removal of body parts or organs and then dropping them in the fixative is a respectable method of fixation provided the pieces to be fixed are very small. This type of fixation, however, still remains inferior to the superb quality of fixation achieved through perfusion. Immersion fixation of pieces of organs is frequently utilized in electron

microscopy, but is to be restricted only to those organs which can be dissected and finely "chopped" in the fixative within about one minute of sacrifice. Immersion fixation is ill advised where tissues cannot be reached quickly. Oftentimes, if the desired tissue can be quickly exposed, it is flooded with the fixative, dissected, and transferred to the fixative.

In summary, for superb fixation of cellular detail the only recommended procedure is intracardiac perfusion. Other procedures can be followed in the field, but with compromised fixation. The practice of immersing the entire animal in the fixative should be avoided since it yields little advantage to anyone else other than the collector himself.

Acknowledgements

The author is grateful to his peers, especially Dr Frank C. Kallen, Buffalo, New York, and Dr Heinz Stephan, Frankfurt A.M., for the opportunity of learning and observing these basic, yet highly useful, techniques. Thanks are due to Dr Fred J. Roisen and Dr Charles E. Wagner for critically reading the manuscript, to Susan Hodge for secretarial assistance, and to George Batik for the illustrations.

Recommended Reading

Bhatnagar, K.P. (1980) Notes on preserving bats in the field. *Bat Research News* 21, 8-10.

Fox, C.H., Johnson, F.B., Whiting, J. and Roller, P.P. (1985) Formaldehyde fixation. *J. Histochem. Cytochem.* 33, 845-853. Review article.

Humason, G.L. (1972) *Animal Tissue Techniques.* Freeman and Company, San Francisco, 641 pp.

Lillie, R.D. (1965) *Histopathologic Technic and Practical Histochemistry.* McGraw Hill, 715 pp.

Palay, S.L., McGee-Russell, S.M., Gordon, S. and Grillo, M. (1962) Fixation of neural tissues for electron microscopy by perfusion with solutions of osmium tetroxide. *J. Cell Biol.* 12, 385-410.

Williams, T.H. and Jew, J.Y. (1975) An improved method for perfusion fixation of neural tissues for electron microscopy. *Tissue & Cell* 7, 407-418.

Appendix A: Formulae for Making Solutions

10% NEUTRAL BUFFERED FORMALIN (pH 7.0)

37-40% Formaldehyde solution[1]	100 ml
Distilled water[2]	900 ml
Acid sodium phosphate, monohydrate	4 g
Anhydrous disodium phosphate	6.5 g

Continued ...

SALINE IN NITRITE

Distilled water[2]	500 ml
Sodium chloride (table salt)	4.5 g
Sodium nitrite	2.5 g

PHYSIOLOGICAL SALINE

Distilled water[2]	1000 ml
Sodium chloride (table salt)	9 g

AQUEOUS BOUIN'S FLUID[3]

Picric acid (saturated aqueous solution)	75 ml
Formalin (37-40%)	25 ml
Glacial acetic acid	5 ml

NEMBUTAL SODIUM[4] (sodium pentobarbital)

Nembutal (50-60 mg/ml)	3 ml
Physiological saline, 0.9%	27 ml

DILUTION OF 95% ALCOHOL

Use 95% alcohol. Fill the graduated cylinder with 95% alcohol to the mark desired (for 70% alcohol, 70 ml; for 80% alcohol, 80 ml etc) and then add distilled water up to the 95-ml mark. The density for 95% and 90% alcohol at 15°C is 0.816 and 0.834 respectively.

1 Fixation with unbuffered formalin produces the risk of the formation of "formalin pigment" since formaldehyde oxidises quite readily producing formic acid (see Fox *et al.* 1985, p. 847). This is specially so for the blood-rich tissues. Despite this problem, use of unbuffered formalin may still be inconsequential, and therefore the step for buffering can be eliminated.

2 Distilled water can be readily substituted with the one available in the field, specially from a natural source. It should be filtered.

3 To be made freshly each time. Keep ingredients separate. Mix when needed. Bouin's provides superb fixation of tissues. Picric acid should be removed while dehydrating. Add few grains of lithium carbonate to the jar containing specimen when in 70-80% alcohol. Keep changing alcohol until no more yellow colouration is perceived.

4 Of the above dilution use only about 0.1 ml/10 g (or 0.07 mg/g) body weight of the bat, intraperitoneally, for light anaesthesia. If failure occurs, a second small dose may be necessary. Nembutal is also available in powder form.

Appendix B: Equipment

i. Perfusion apparatus, in at least three sets (Fig. 1)
Luer-lok syringes, 10-30 ml (plastic ones are lighter and unbreakable, but need frequent replacement)

ii. Other equipment
Corkboard in a metal or plastic pan
Dissecting instruments including artery clamps, haemostats, a metal ruler

Continued ...

Three-way stopcocks fitted with polyethylene tubing (PE240, 2 mm diameter) and adapter to take injection needles of different sizes
Two widemouth bottles with tight-fitting screwtop caps
Injection needles (cannula) in several sizes
Tuberculin syringe, 1 ml, for anaesthesia
Table lamp
Graduated cylinder (100-500 ml)
Funnel and filter paper
Scale for weighing
Tags, India ink, pens, pencils, markers
Thin leather gloves
Mist nets, butterfly nets etc
Cloth bags (with drawstrings)
Logbook
Flash lights
Camera and films
Vials of several sizes
Travel clock
Dissecting microscope

Appendix C: Chemicals

Acetic acid, glacial
Acid sodium phosphate, monohydrate
Anhydrous disodium phosphate
Ethyl alcohol, 95%
Formalin (37-40%)
Nembutal sodium
Picric acid, saturated aqueous solution
Sodium chloride (or table salt)
Sodium nitrite
Lithium carbonate

Appendix D: Suggested Logbook Entries

Serial number, Genus & Species, Sex
Adult/Subadult/Juvenile
Date and time of collection
Place/Biotope
Body weight (g)
Pregnant uterus weight (g)
Head & body length
Forearm length
Tail length
Ear length
Hindfoot length
Maxillary tooth row length
Wing span
Time of anaesthesia
Time of perfusion
Fixative
General comments

ON THE PROBLEM OF "RABIES" IN BATS*

Ulrich Jüdes

IPN/University of Kiel, Olshausenstr. 62,
D-2300 Kiel 1, Federal Republic of Germany.

Abstract

The identification of rabies-related rhabdoviruses in European bats has resulted in exaggerated and in some instances incorrect reports in the mass media, and has increased general hysteria directed against bats. In this article, virological, clinical and epidemiological aspects of rabies and rabies-related viruses (genus lyssa viruses) are described, the data on the occurrence of lyssa viruses in bats collected, open questions discussed and practical hints given for personal behaviour with regard to bats.

In summary it can be stated that the occurrence of lyssa virus serotype 4 (Duvenhage) has hardly any medical relevance for man. Minimal potential risks for bat workers and other exposed persons can easily be removed by prophylactic innoculation with rabies vaccine. Direct interventions against bats are - as latest findings show - neither justified nor effective, besides conflicting with the needs of bat protection.

Introduction

In 1986 the West German tabloid press caused widespread concern with a spate of sensational headlines: "Rabid bats bit pupils and teachers", declared one Hanover newspaper, "Diseased bats fell from the sky", disclosed a TV magazine. A south German newspaper called a "Vampire Alarm", and a Hamburg daily issued a "Warning" against the "animals who bear death and disease".

But quite apart from the exaggeration and false information issued by much of the press, several questions arose which at that time could not be answered due to the lack of relevant data:

- How real is the danger for those who often come into contact with bats?
- Is there a genuine risk for the population as a whole from rabid bats?
- To what extent should activities be modified which set out to protect bats and encourage them to roost in buildings near human habitations?

Although the flood of alarmist reports abated towards the end of 1986, it is to be expected that concern will grow once more when the bats come out of hibernation.

The WHO recommends that a bat expert should be consulted whenever rabies-control measures are taken which affect bats[53]. This is to

---

* (A German version of this article appeared in: *Myotis (Bonn)* 25, 1987).

ensure that pointless steps are avoided, such as the setting up of rabies quarantine areas and uncontrolled intervention in bat colonies. The bat experts must, however, themselves be fully familiar with the rabies problem.

I am, therefore, pleased to take up the editor's request to collect the most important data on rabies, as far as they are accessible and relevant for bats, as well as to discuss open questions and investigate the problems created by the, at times, conflicting interests of epidemic control and bat conservation.

1. *Medical and Virological Aspects of Rabies*

Rabies (lyssa) is an acute virus infection of the central nervous sytem in warm-blooded animals[45], though susceptibility to the virus differs considerably from species to species. The infection is transmitted in the saliva of an infected animal, generally as a result of a bite or contact with broken skin. The possibility of infection by aerosol inhalation, e.g. in the laboratory or in bat caves, has been discussed in the USA[10] and explored in experiments[11].

The morbidity rate in humans bitten by a rabid animal is by no means 100%. In fact, even without vaccination the average rate is around 15%, depending on the location of the bite or infected wound (Table 1) and the species of animal which gave the bite[22,57].

Table 1: Morbidity depending on the type of wounding (from wolf, fox or dog).

Type of wounding	Morbidity
Serious bite wound in the eye or nose region	almost 100%
Serious bite wound on neck or hands	15-20%
Lighter hand wound	5-10%
Light wound on legs or trunk	2%

The incubation period of the disease is usually around 2 to 4 weeks in foxes, 2 to 8 weeks in dogs and 3 to 12 weeks in humans, although this can vary greatly (up to 5 months in foxes, 5 days to a year in dogs, 10 days to over 15 months in humans)[30,56]. Thanks to this relatively long incubation period, vaccine treatment (active immunization) can be administered after infection (see section 6).

If infection leads to an outbreak of the disease, it is generally fatal (in animals within 10 to 14 days, in humans within 3 to 7 days following the appearance of the first symptoms)[30]. Only two humans are so far known to have survived an attack of rabies[23].

Healthy virus carriers are unknown. Although vampire bats were long thought to be able to survive a rabies infection whilst still transmitting the virus via their saliva over a longer period of time (in some cases life-long)[56], recent investigations have been unable to prove

this theory[47]. The relatively long incubation period of the rabies virus can lead to the deception that the host does not suffer an acute outbreak of the disease. Vampire bats are "able to act as a symptomless carrier for up to 5 months"[57]. In general it can be said that animals, including vampire bats, which suffer an attack of rabies do not survive the disease[30].

It should be mentioned that an infected animal usually begins to secrete the virus about 2 to 3 days before the first symptoms of the disease appear. As wild animals do not bite without motivation at this stage, the fact is of little practical significance[56].

The rabies virus, having been transmitted through an animal's saliva, infects the cells of the tissue around the bite or wound. Liquid saliva remains infectious for 24 hours at room temperature, dry saliva for up to 14 hours[14]. The virus replicates itself in striated muscles, and frequently leads to local symptoms in the area of infection, where it remains for up to 72 hours. The virus then spreads towards the brain via the peripheral nervous system (neuromuscular junctions - axons) and the ganglion cells of the spinal cord at a rate of up to 7 cm per day. In the brain virus colonies, called Negri bodies, of varying size are formed. As greater virus localization occurs in the limbic system, the host tends to undergo quite noticeable behavioural changes. From the brain the virus continues to propagate, spreading via the nerve fibres to peripheral organs such as the salivary glands, the papillae of the tongue and the nasal mucosa[30].

As the virus spreads only through the central nervous system and not the blood, a massive immune reaction of the body sets in quite late, when large quantities of the virus particles are finally released from the infected nerve cells[23,45].

The rabies virus itself belongs to the Rhabdoviridae (rhabdos = rod). It is 180 to 210 nm in length, typically bullet-shaped (i.e. almost cylindrical with one rounded end) and consists of a lipid-containing bilaminar outer membrane and the nucleocapsid with the viral genome (RNA). The virus is inactivated at temperatures in excess of 56°C, and by ultra-violet and solar rays, so it cannot long survive outside the host cells. It is not killed by low temperatures (e.g. by freezing infected animals).

The virus has specific antigenic properties which are of practical significance when identifying the virus by immunofluorescence, a neutralization test or by using monoclonal antibodies. On the basis of such antigenic properties on the surface of the virus and in the nucleocapsid, various strains of rabies virus can be identified[23].

- Street virus strains: wild strains of full virulence (e.g. DR19B: isolated from a vampire bat, *D. rotundus*, in Rio de Janeiro; R-205: isolated from a badger).

- Fixed virus strains: adapted in laboratory animals with partial loss of pathogenicity (e.g. CVS - Challenge Virus Standard).

As all these strains show serological cross-reactions when neutralization or haemaglutination tests are applied, they are grouped under serotype 1 of the lyssa viruses.

Besides these rabies viruses in the narrow sense there are at least five rabies-related virus serotypes which are grouped together with the former as lyssa viruses[23,38,45,47]. These various serotypes have common structural and immunological properties, but can be clearly distinguished by means of neutralization tests and by using monoclonal antibodies[36]. Table 2 shows the lyssa viruses.

Table 2: Rabies and rabies-related viruses.

Serotype	Virus type	Distribution	Hosts
1	Rabies virus prototype	almost worldwide	all warm-blooded animals, particularly carnivores regionally bats (America) Man
2	Mokola prototype	Nigeria	Shrews (*Crocidura flavescens* and *C.* spp.)
		Cameroon	Man
		Zimbabwe	Dogs Cats
3	Lagos bat prototype	Nigeria Cent. Afr. Rep. South Africa	Bats (*Eidolon helvum*, *Micropteropus pusillus*, *Epomophorus wahlbergi*)
4	Duvenhage prototype	South Africa Europe	Bats (unknown African species, in Denmark and FRG almost exclusively *Eptesicus serotinus*) Man (1x South Africa, 1-2x USSR)
-	Obodhiang prototype	Sudan	Insects (*Mansonia uniformis*)
-	Kotonkan protype	Nigeria	Insects (*Culicoides* spp.)

The viruses of the various serotypes do not all necessarily cause rabies-like illnesses[45]. The Duvenhage virus and the Mokola virus under certain circumstances can cause a serious, fatal attack of encephalitis or myelitis in humans[39].

The **Mokola virus** has been identified in humans in two cases, and otherwise in Cameroon shrews[39]. More recent findings indicate its possible occurrence in dogs and cats[30,47].

The **Lagos virus** was first isolated in frugivorous megachiroptera in Lagos (Nigeria), near Bagui (Central African Republic) and near Durban

(Natal). On the other hand, the viruses found in dogs in the same area of Natal belonged to serotype 1, which confirms that the Lagos bat virus has a mode of dispersion independent of domestic animals[38]. In experiments, however, the Lagos bat virus is pathogenic in dogs and monkeys; nothing is known to date of its pathogeny in humans[39]. A man bitten by a fruit bat showed no pathological symptoms following vaccination[27], although the normal rabies vaccine is not known to stimulate immunity to the Lagos virus (or to the Mokola virus)[2,52].

The **Duvenhage virus** was first identified in South Africa in 1970, in a human who did not survive the infection[26]. A connection with a bat bite was suspected. Bats subsequently examined were shown to be carrying a virus closely related to the Duvenhage strain[39]. In the Soviet Union a Duvenhage-related virus was identified in a young girl following her death[55]. A probable connection exists between the illness and a bat bite.

Viruses found in Hamburg (1968), Stade (1979) and Bremerhaven (1982) were judged "closely related, but not identical, to the Duvenhage virus"[53] on the basis of tests with monoclonal antibodies. They showed different reactions in the nucleocapside reaction with various antibodies, but can clearly be assigned to the heterogeneous serotype 4, which indicates an African origin[36; see also section 6]. Further viruses in bats recently located in Denmark, Poland and the Federal Republic of Germany also belong to Lyssa serotype 4. The Danish and German viruses isolated are serologically identical[16], but whereas Danish scientists speak consistently of a Duvenhage-related virus in their bats[12,16], the viruses found in Poland, Denmark and the Federal Republic of Germany in 1985 and 1986 are termed by Schneider both as Duvenhage viruses[52 and personal comment] and as viruses very similar to the Duvenhage strain[53].

In laboratory tests mice, dogs and cats were infected with the Danish virus (by intramuscular or intracerebral injection)[7,16]. This, however, says nothing about the possibilities of natural infection.

The virus types **Obodhiang** and **Kotonkan** are something of a curiosity, as they are present only in stinging/biting insects. No evidence of human pathogeny has been found so far; artificially infected monkeys and dogs did not contract a disease[39].

Although the five rabies-related virus types are serologically quite different, certain immunological cross-reactions can be observed. Tests carried out on mice indicated that the usual rabies vaccine offers no protection against the Mokola and Duvenhage viruses[52]. Differences in the effectiveness of rabies vaccine have also been proved for variants of virus serotype 1. For example, vaccines of the PM strain provide only partial protection to laboratory mice against rabies strains from Thailand, Iran or Madagascar[39].

Further experiments with the Stade bat virus strain ascertained a reduced, but adequate production of antibody titres for both pre-exposure and post-exposure innoculation with rabies vaccines (RABIPUR

produced by the Behring Company, used in Germany)[24,53]. These findings are confirmed by Danish experiments[7]. Protection should, therefore, be adequate if the WHO vaccination guidelines are observed.

Neurological complications have been reported for all vaccines used to date, apart from cell culture vaccines (particularly those from human diploid cells (HDC))[6]. The vaccines used in Europe, which are produced by the companies Behring and Merieux, belong to the latter group. At present work is going on to produce a vaccine by genetic engineering[28].

2. *Epidemiology of sylvatic (wild animal) rabies*

Rabies is a disease with virtually world-wide distribution, which is recorded as early as 2300 BC in the Babylonian Empire[45]. Rabies-free areas are primarily geographically isolated regions (in Europe: Spain and Portugal, Norway, Sweden and Finland, as well as a large number of islands such as Great Britain, Ireland, Iceland and Malta; outside Europe: Japan, Australia, New Zealand). The areas of distribution spread in relation to the dynamics of population and the migration of the carrier species. In Europe, for example, the progression is westwards (in 1939 Poland was the westernmost area, by 1968 it had spread to France[22]). In South America the disease is presumed to be spreading along the Transamazonian motorway[42].

Rabies caused by virus serotype 1 is mainly carried by carnivores[30]. Two general modes of dispersal can be distinguished:

> The predominant type in the developing countries of Asia, Africa and Latin America is the **urban form (domestic animal rabies)**. The most frequent carriers are stray dogs[6,45]. In a country such as Nigeria, for example, 98.8% of all rabies cases in humans result from contact with rabid dogs[15].
>
> The immunization of dogs (and in some cases cats) has led to a reduction in urban rabies in the more developed countries. Here the predominant type is the **sylvatic form (wild animal rabies)**, which is spread by various species of wild animal depending on the region (see Table 3). Of the 55,078 cases of animal rabies ascertained in Europe between 1960 and 1975, 80.3% were in wild animals. Of these, 83.3% were in foxes, with a high rate among badgers and other carnivores[23].

In Europe the rabies virus has also been found in the following species of wild animals: hedgehog, shrew, wild rabbit, brown hare, squirrel, common dormouse, fat dormouse, hamster, field mouse, musk, house mouse, brown rat, stoat, weasel, polecat, raccoon, marten, wild boar, moufflon, red deer, European bison, as well as in kites, falcons, little owls and magpies[21,32,54]. These appear to serve as hosts only, if the main carrier species are infected with rabies[47].

In the Federal Republic of Germany, a total of 15 human deaths from rabies were registered between 1951 and 1970, of which 7 cases

(= 47%) were due to infection contracted whilst abroad. We have no record of any further human cases since 1970[23].

Table 3: The major rabies-transmitting species in continents (after 5, 27,40).

Region	Main hosts
Europe	Red Fox (*Vulpes vulpes*) Badger (*Meles meles*) Raccoon dog (*Nyctereutes procyonides*) Domestic dog (*Canis familiaris*) Domestic cat (*Felis catus*)
North America	Skunks (*Mephitis mephitis*, *Spilogale putorius*) Raccoon (*Procyon lotor*) Red Fox (*Vulpes vulpes*) Grey Fox (*Vulpes cinereoargenteus*)
Central and South America	Vampire bat (*Desmodus rotundus*) Mongoose (*Herpestes auropunctatus*) Domestic dog (*Canis familiaris*) Domestic cat (*Felis catus*)
Africa	Domestic dog (*Canis familiaris*) Domestic cat (*Felis catus*) Jackals (*Canis aureus, C. mesomelas*) Mongoose (*Cynictis penicillata*)
Asia	Domestic dog (*Canis familiaris*) Domestic cat (*Felis catus*) Wolf (*Canis lupus*) Arctic fox (*Alopex lagopus*)

World-wide the situation is rather more serious: around 1000 deaths from rabies are reported to the WHO each year. This does not, however, adequately reflect the true figure: in India alone some 15,000 cases of rabies are reported annually[23].

3. *Rabies and rabies-related viruses in bats*

The first case of lyssa virus transmittal by bats to be reported was in 1921[40]. The following paragraphs outline the incidence of rabies and rabies-related viruses, continent by continent, according to information available to date.

(a) The Americas

The lyssa viruses ascertained in American bats all belong to serotype 1. In Central and South America and in the Caribbean, the common vampire bat (*Desmodus rotundus*) is an important source of

infection, particularly for cattle. In these regions almost half a million cattle die each year from rabies transmitted by bats[23].

In North America (including Canada) insectivorous bat species are also carriers of rabies viruses, a fact ascertained for the first time in 1953[32]. In Texas the Mexican freetail bat (*Tadarida brasiliensis*) is thought to be particularly affected[30]. Rabies was identified in a total of 30 of the 39 bat species in North America, which belong to the following families: *Diphylla, Tadarida, Dasypterus* (e.g. *D. floridianus*), *Lasiurus, Mollosus, Myotis, Eptesicus* (e.g. *E. fuscus*), *Artibeus* and *Uroderma*[13,29,31,45]. Despite the wide variety of affected species and the widespread discovery of rabies-positive animals in almost all USA states and half of the Canadian provinces, bats are still a relatively minor source of infection in North America, except in limited areas of the north-west[29,57]. Of the 269 cases of human rabies in the USA between 1946 and 1983, only 11 (= 4.1%) could be traced to bats, whereas 173 (= 57.1%) could be traced to dogs[29].

In Canada only 4.1% of all animal rabies cases were in bats (69 individuals), although a relatively large number of animals were examined in 1984. Exact figures are available for three provinces:

Ontario	713	bats	examined/	51	virus carriers	(7.2%)
Alberta	206	"	"	/6	" "	(2.9%)
British Columbia	131	"	"	/6	" "	(4.6%)

In 1983 and 1985 only 2.8% (= 63) and 2.5% (= 56) of all rabies cases recorded were in bats[29,49].

The frequency of virus-positive individuals among the dead, diseased or otherwise suspect bats discovered in North America is between 3 and 10%[46]. However, in random samples from natural populations, the frequency of virus-positive animals is much lower than 0.5%[46].

In South America *Phyllostomus* sp. and *D. rotundus* are the main carrier species of rabies (Pawan, see 5).

(b) Asia

In the Asian part of the Soviet Union there have been two discoveries of lyssa-positive bats: a *Vespertilio murinus* in Omsk in November 1984, and a *Myotis daubentoni* in the Novosibirsk area in 1985[55].

For the rest of Asia only two examples of lyssa viruses could be found in 79 frugivorous bats (*Cynopeterus brachyotis*) in Kanchanaburi/ Thailand[40]. No other tests on bats have yet identified further cases of lyssa viruses[45].

(c) Africa

In 1956 a virus isolated from a flying-fox (*Eidolon helvum*) on Lagos Island in Nigeria was described as Lagos bat virus[38]. Later this virus was also found in a dwarf epauletted flying-fox (*Micropteropus*

pusillus) 110 km north of Bangui in the Central African Republic, as well as in a common epauletted flying-fox (*Epomophorous wahlbergi*) in Pinetown near Durban in Natal[27].

In 1971 a new virus - the Duvenhage virus - was described from a man who had been bitten on the lip by a bat in South Africa[26]. In the course of further investigations in this region another virus strain, very similar to the Duvenhage virus, was also discovered[39].

The findings point to a considerable heterogeneity in the lyssa viruses present in Africa, both within the genus and within the various serotypes[36].

(d) Australia

The continent of Australia is free of rabies[30].

(e) Europe

Reports on the occurrence of lyssa viruses in bats are not new to Europe. Most experts agree, however, that the insectivorous European bats cannot be regarded as major carriers of lyssa viruses[7,21,22,32,45, 48].

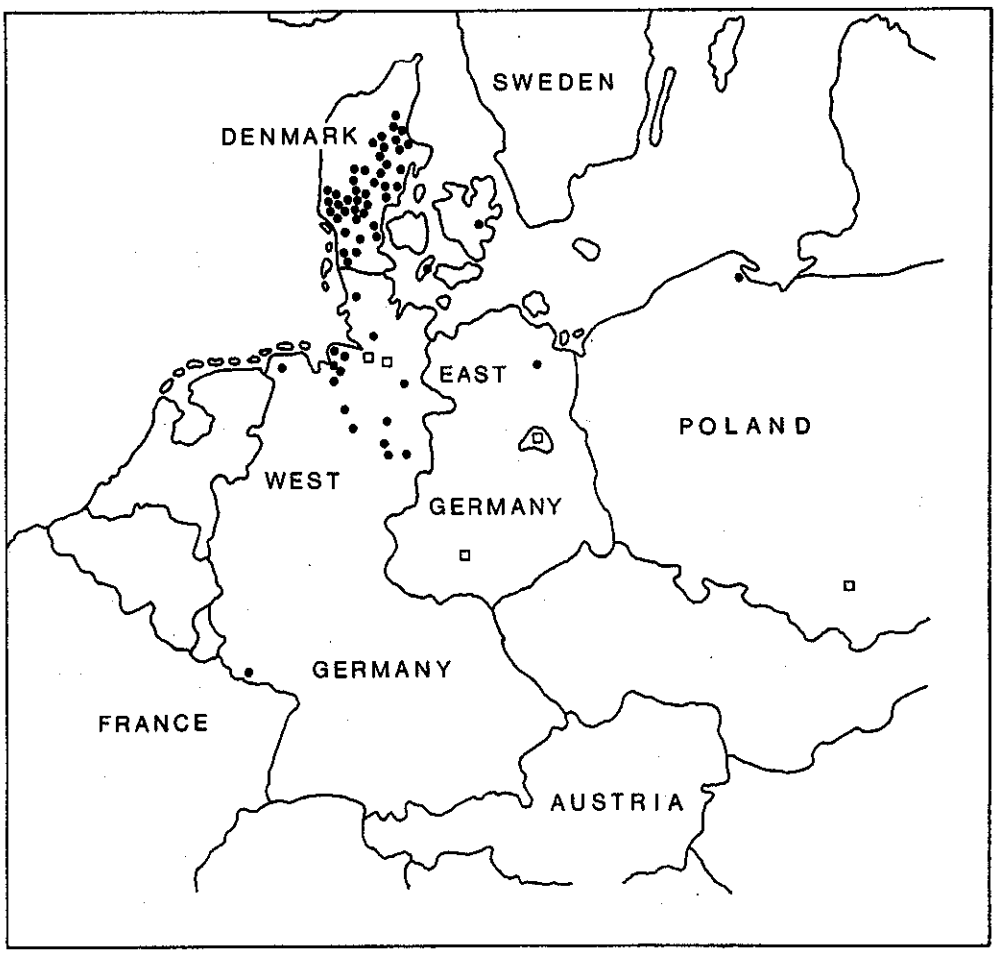


Figure 1: Geographical distribution of Lyssa-virus findings in bats of North and Central Europe (status: end of 1986).

□ record before 1980 • record after 1980

Schoop (see 33) describes a few cases in Yugoslavia and one in northern Germany, where the bats could not be identified. According to WHO reports (see 34), virus-positive bats have also been discovered in Turkey, Yugoslavia and East Germany. The following species were affected[32]:

Nyctalus noctula (Yugoslavia 1954 and 1956)
Rhinolophus ferrum-equinum (Turkey 1956)
Eptesicus serotinus (German Democratic Republic: Jena 1963).

Investigations on 421 bats of different species (including 405 *Myotis myotis*, 6 *Plecotus auritis*, 6 *Barbastella barbastella* in the north of the Federal Republic of Germany and in the German Democratic Republic yielded only negative results[32]. Further virus-positive findings for the Federal Republic of Germany in the years 1968, 1970 and 1973 are displayed in Table 5.

In the German Democratic Republic a lyssa-positive *E. serotinus* was discovered in Jena in July 1963[32]. In the third quarter of 1986 a further virus-positive bat (species unknown) was recorded in the district of New Brandenburg[54].

In Poland (Krakau) in 1972, a bat of unknown species was registered as being suspected of having rabies. In May 1985 a rabies-related virus of the Duvenhage type was discovered in a bat (*E. serotinus*) found near Danzig[50,52].

In October 1964 a man was bitten by a bat (*E. serotinus*) in Kiev in the European part of the Soviet Union. A lyssa virus was later identified in the bat. The man was treated with rabies vaccine and did not contract the disease[55]. In 1977 and 1985, however, two people, in Voroshilovgrad and Belgorod (Ukraine), died of rabies traced to bites from bats of unidentified species. Neither victim was vaccinated. The incubation periods in the two cases were estimated at 3 weeks (from a bite on the lower jaw) and 4 weeks (from a bite on the hand). In the latter case a virus related to the Duvenhage type was identified immunologically[55].

In Denmark bats were first identified as lyssa virus carriers in 1985[12,51], after rabies had been eradicated in three non-endemic localities between 1964 and 1982[47], where cats had been the only animals affected, and these in very few cases[52]. No further cases of rabies had been registered in Denmark since 1983[54]. In the third quarter of 1985, however, a lyssa-positive bat was found near Ribe; in the final quarter of that year 9 out of 23 bats examined were positive, all of them *E. serotinus*, found when either sick or dead[51]. The investigation results for 1986 are presented in Table 4 below. Discoveries of the virus were most frequent during August[16].

On October 29, 1985, a biologist died of rabies in Helsinki. He had been bitten by bats during his work in Finland, Switzerland and, 4 1/2 years earlier, in Malaysia. This was the first case of human rabies in Finland for more than 50 years. Since 1959 the country had been

completely free of rabies[25]. The origin of this new infection is unclear, particularly as *E. serotinus* is not known to exist in Finland. Immunological tests showed that the Finnish virus was identical neither to the Danish viruses nor to the Duvenhage strain[7].

Table 4 shows the data obtained in European countries. No data are available for the countries not mentioned.

Table 4: Lyssa virus studies in European bats including Turkey (as at January/February 1987).*

Country	No. of bats	Virus-positive bats	Species (No./pos. iden.)
Denmark**	574	105	*E. serotinus* (366/103) *P. pipistrellus* (52/0) *P. nathusii* (2/0) *N. noctula* (7/9) *M. daubentoni* (30/1) *M. dasycneme* (5/1) *M. brandti* (3/0) *M. mystacinus* (2/0) *M. nattereri* (1/0) *Pl. auritus* (30/0) *V. murinus* (59/0) *B. barbastellus* (1/0)
Finland	183 (rabies-free)	0	*E. nilssoni* (?/0) *M. daubentoni* (?/0) *M. mystacinus* (?/0) *M. brandti* (?/0) *Pl. auritus* (?/0)
Great Britain	28 (rabies-free)	0	*E. serotinus* (2/0) *P. pipistrellus* (15/0) *N. noctula* (1/0) *M. nattereri* (1/0) *M. mystacinus* (2/0) *M. daubentoni* (2/0) *Pl. auritus* (3/0) unident. spec. (2/0)
Federal Republic of Germany	(59)***	19	*E. serotinus* (14/13) *N. noctula* (1/0) *P. pipistrellus* (8/0) *P. nathusii* (2/1) *M. daubentoni* (1/1) *Pl. auritus* (1/0) *Pl. austriacus* (1/0) unident. spec. (?/4)
Democratic Republic of Germany	?	2	*E. serotinus* (?/1) unident. spec. (?/4)

Continued ...

Table 4 continued...

Country	No. of bats	Virus-positive bats	Species (No./pos. iden.)
Poland	?	2	*E. serotinus* (?/1) unident. spec. (?/1)
Sweden***	274	3	*E. serotinus* (?/1) *M. daubentoni* (?/1) *V. murinus* (?/1)
Switzerland**	74	0	11 different species
France	0	0	---
Yugoslavia	?	3	*N. noctula* (?/3)

* Data from central research laboratories in the respective countries (see Acknowledgements) and from literature.

** Data refer to 1986 only.

*** The total number of bats examined comprises only those examined at the WHO Rabies Collaboration Centre at Tubingen. There is a lack of data on the virus-free bats examined by the regional veterinary authorities, which forward only virus-positive animals to Tubingen.

In Table 5 the localities of all virus-positive bats found in the Federal Republic of Germany are listed. Most of the finds were in the north, in Lower Saxony (see Figure 1). Very little data are available so far for Schleswig-Holstein.

Table 5: Findings of Lyssa-virus-positive bats in the Federal Republic of Germany (after Muller, in lit. and Wernery/Judes, unpubl.).

Location	Year/month	Species	Evidence
Hamburg	1954, Oct	unknown	Mohr (1957)
Hamburg	1968, Jul	unknown	Werschning and Schneider (1969)
Stade	1970	unknown	WHO Rabies Centre
Berlin	1973	*M. myotis*	Hentschke and Hellman (1975)
Bremerhaven	1982, Aug	unknown	WHO Rabies Centre
Aurich	1983, Sep	*E. serotinus*	"
Bremerhaven	1985, May	*E. serotinus*	"
Rodenberg/Hann.	1985, Jun	*E. serotinus*	"
Nienburg/Weser	1985, Oct	*E. serotinus*	"
Langwedel/Verden	1986, May	*E. serotinus*	"
Ottweiler/Saarbr.	1986, Jun	unknown	"
Fallingbostel	1986, Aug	*E. serotinus*	"

Continued ...

Table 5 continued...

Location	Year/month	Species	Evidence
Luneburg	1986, Aug	unknown	WHO Rabies Centre
Nienburg/Weser	1986, Aug	*E. serotinus*	"
Peine	1986, Aug	*E. serotinus*	"
Wedemark/Hann.	1986, Sep	*E. serotinus*	"
Hagen/Bremen	1986, Sep	*P. nathusii*	"
Nienburg/Weser	1986, Sep	*E. serotinus*	"
Dorum/Cuxh.	1986, Sep	unknown	"
Loxstedt/Bremhv.	1986, Sep	*E. serotinus*	"
Steinau/Cuxh.	1986, Sep	unknown	"
Luneburg	1986, Sep	*M. daubentoni*	"
Mildstedt/Husum	1986, Oct	*E. serotinus*	Werney (pers. acc.)
Itzehoe	1986, Oct	*E. serotinus*	"

The findings for Schleswig-Holstein and Lower Saxony are listed separately in Table 6.

Table 6: Number of bats examined in Schleswig-Holstein and Lower Saxony.*

State	Total no.	Species (Total/virus-positive)
Schl.-Holst.**	39	*M. daubentoni* (20/0) *M. nattereri* (2/0) *E. serotinus* (16/2) *N. noctula* (1/0)
Nieders.***	198	*M. brandti* (3/0) *M. mystacinus* (5/0) *M. brandti/mystac.* (3/0) *M. nattereri* (1/0) *M. daubentoni* (39/1) *E. serotinus* (65/11) *N. noctula* (11/0) *Pl. auritus* (6/0) *V. murinus* (1/0) *P. pipistrellus* (41/0) *P. nathusii* (3/1) Art unident. (20/3)

* For other states there is no information on how many bats have been examined by the regional veterinary authorities (23 offices in FRG).

** After Wernery and Judes (unpubl.).

*** After Siedler *et al.* (1987) and Muller (in lit.).

The Danish findings up to the end of 1986 are concentrated in the south and east of Jutland (see Figure 1). A number of virus-

positive bats have also been found more recently on the islands of Fyn and Zealand (Baagoe, in lit.).

The percentages of virus-positive bats among those believed suspicious and investigated in Denmark, Schleswig-Holstein and Lower Saxony are as follows:

Denmark	18.3.% (1986 only)
Schleswig-Holstein	5.1%
Lower Saxony	8.1%

Tests on several *P. pipistrellus* and *E. serotinus* in the area around Aachen produced no evidence of lyssa viruses (Roer, in lit.).

According to the relevant literature[12,37,48,51,53] it appears that all evidence of lyssa viruses in bats in Europe can be traced to animals found in a diseased or weakened condition, generally sitting more or less paralyzed on the ground.

4. *Open questions concerning bat rabies*

The findings to date regarding the incidence of lyssa viruses in European bats still leave a number of questions unanswered.

Where does the lyssa virus found in European bats originate?

The bat virus is presumed to have originated in Africa. This assumption is based on two facts:

(a) The European bat virus is closely related to the African Duvenhage virus[36].

(b) The variety of lyssa genus serotypes identified in Africa is greater than in any other region of the world (see 38), a fact which points to this area as the evolutionary centre of the genus.

It is not yet clear when and how the virus spread to Europe: whether it reached this continent via non-European bats[35,36] or whether it was able to propagate naturally over such distances.

The unidentified bat found in Hamburg in 1954 and registered by Mohr could have belonged to an African species[35]: Erna Mohr, a mammalogist, could safely be expected to have recognized any native species.

On the other hand, it should be remembered that the area of distribution of *E. serotinus*, the bat with the highest incidence of rabies virus in Europe, includes North Africa and a number of Mediterranean islands. This could have led to contact with affected areas in Africa.

Evidence of rabies virus in a *N. noctula* in Yugoslavia[32] and a *V. murinus* in the Soviet Union[55], both of these being species which travel long distances, provides further support to the theory that

the virus can be transported over vast areas. The distribution area of *N. noctula* also includes North Africa.

It is to be expected that tests on the European lyssa viruses using monoclonal antibodies will shed some light on the question, as this method shows up antigen characteristics of individual virus strains, which may have developed during the spread of the virus[36,47]. In America, however, these characteristics have been shown to demonstrate considerable heterogeneity within an area, which indicates an intermixing of the bat populations[29].

Is the virus long established in Europe or was it introduced more recently?

Here, too, we can do no more than postulate at present. Although certain factors (findings in Hamburg, a sea-port, in 1954 and 1968; concentration of virus incidence in coastal areas and in one bat species) indicate that the virus is a fairly recent import, there is other evidence to show that the virus has been present in Europe for some considerable time, but was discovered by chance only recently, i.e. in the last few decades. Baagoe (in lit.) considers the latter explanation the more probable.

As *E. serotinus*, the main species affected in Denmark and North Germany, tends to stay in limited areas and does not undertake any major seasonal migrations, the virus, once introduced into a region, would take a relatively long time to spread farther afield (for example, from Hamburg to Denmark or Poland). The widely dispersed virus findings in Yugoslavia, Turkey and the German Democratic Republic, made as early as the 1950's and 1960's, could be proof of a longer existence of the virus in Europe, but also of multiple introduction (e.g. by migratory bat species). This brings us to the next question.

How widespread is the virus in Europe today?

The findings of virus-positive bats available to date presumably reflect the collecting activity of bat researchers rather than the true distribution of the virus.

All the data we have point to *E. serotinus* as virtually the exclusive virus carrier in northern and central Europe. This species is found in almost all regions of Europe, apart from the northern and central areas of Great Britain, Norway, Sweden (found only in southern Sweden; Baagoe, in lit.) and Finland. As the epidemiology of a virus depends on the density and dynamics of the host population, the virus can only be expected to thrive in those areas where the host is well represented. In the case of *E. serotinus* this applies to the plains of Denmark, western and northern Germany and Poland.

The fact that virus-positive bats have been found in many parts of Europe is no indication that the virus is equally distributed over such a wide geographical area, as several instances concerned migratory bats - a fact which complicates the epidemiology of the virus in Europe.

How is the virus transmitted from bat to bat?

Due to the low density of bats in the caves and buildings of central Europe, the danger of aerosol virus transmission demonstrated in American experiments can be regarded as non-existent in this geographical area.

As far as transmittal from bat to bat via social contact without biting (e.g. licking of the snout region with mucosa contact) is concerned, we can only hypothesize. This factor would be of particular significance in the few days before symptoms of the disease (accompanied by abnormal behaviour) become apparent, when, as findings in carnivores and laboratory animals show, the virus can already be present in the saliva.

Throughout the year, *E. serotinus* roosts in buildings (caves only in exceptional cases) and sometimes associated with other species, such as *P. pipistrellus, M. dasycneme, P. auritus* (Baagoe, in lit.). Until now, investigations of species other than *E. serotinus* give no reason to assume that they are to any considerable degree carriers of lyssa viruses.

How does the virus behave in hibernating bats?

It is presumed that virus propagation slows down or stagnates in hibernating bats, unless virus manifestation has previously progressed far enough to ensure that the animal does not survive hibernation[37].

Does the recent increase in the findings of virus-positive bats indicate a real increase and spread of the disease?

The true cause of the recent "increase" in lyssa findings in bats is more probably increased research activity rather than a real epidemic. A comparable effect was observed when investigations were carried out on voles. In the early 1970's, 2162 small rodents from rabies-free and rabies-infected areas were examined over a period of four years and 28 virus strains isolated. Another study of 635 clinically healthy mice from southern Germany yielded 8 virus strains of serotype 1[22]. These, too, are increases conditioned by the intensity of investigation and are not in conformity with long-term rabies statistics.

How does the lyssa virus affect bat populations? Can it be expected to endanger bat numbers?

Wild animal rabies of serotype 1 appears to fluctuate over a period of several years. In Central Europe the fluctuation is dependent on the main vector, the fox, but also influences the dynamics of the fox population[21,22]. Bats do not show a population development comparable to that of foxes. If, however, the lyssa virus should succeed in spreading massively within a serotine bat colony, a population collapse is to be expected, which would curb the spread of the virus, yet have such a detrimental effect on bat numbers that the colony, due to the low rate of reproduction in bats, would take a long time to recover.

How great is the risk of infection for martens, cats or dogs which come into contact with bats? Can the bat virus be transmitted to humans via other wild or domestic animals?

Experiments with sylvatic rabies virus (serotype 1) have furnished enough evidence that carnivores cannot be infected via the alimentary canal[22]. Transmittal by parasites such as ticks is likewise impossible[3]. Even the transmission of the rabies virus to other animal species via the insectivorous American bats was shown by experiments to be extremely unlikely[47]. Schneider writes[51], that in Europe "natural transmission of rabies from insectivorous bats to other terrestrial animals ... has not been observed to date". Transmission would have to be through a bite from the bat. As martens, cats or foxes can be expected to have more frequent contact with diseased bats than humans, virological examinations were carried out in Denmark in 1986 on 52 cats, 52 foxes, 17 dogs and 31 specimens of other terrestrial animal species, in particular from areas with a high proportion of virus-positive bats. The bat virus could not be identified in any of these animals (16 and Grauballe in lit.). Despite the numerous findings of virus-positive bats in Denmark (in the four most severely affected areas of Jutland, 32% of all *E. serotinus* found weakened or dead in 1986 were virus carriers), no cases of rabies in either wild or domestic animals have been reported in the country since April 1983[54], which indicates that transmittal of the bat virus to other animal species very seldom occurs, if at all. In the Federal Republic of Germany no correlation can be ascertained between findings of virus-positive bats and areas affected by wildlife rabies.

Schneider[54] and Shope[38] are of the opinion that lyssa viruses of the Duvenhage type in African and European bats have a cycle and mode of dispersion independent of other animal species including humans, as the ecology of bats is quite different from that of carnivores. The epidemiology of the virus depends on the host[22,31].

The possible - though highly improbable - risk for humans of infection via domestic animals can be easily removed by prophylactic vaccination of the latter. The risk to humans is, in any case, significant only in the case of house-dwelling bats. If it is assumed that the virus has been present in Europe for decades at least, the virtual absence of human rabies traceable to bat bites (just two cases in the Soviet Union) points to an extremely low risk of infection and disease for humans. North Germans who have had a colony of *E. serotinus* in their house for years report that they have been repeatedly bitten when attempting to touch bats sitting on the ground, yet without contracting disease. This, too, supports the assumption of minimal risk, even with direct contact with bats.

5. *Bat conservation - epidemic control: a potential conflict?*

World-wide eradication of wildlife rabies is considered technically and economically unfeasible[6]. One has only to remember the mass slaughter of foxes in Europe (at least one and a quarter million foxes were killed each year) which merely succeeded in slowing down rather than halting the spread of the disease. The European average for human deaths from rabies over the last two decades lies at 1 to 4 per year[1].

Checks on the bat population by means of similar control measures are impossible for several reasons (lack of local knowledge on and irregularity of bat habitats and, above all, reasons of conservation). Bats tend, moreover, to undertake seasonal migrations far more often than most carnivores, which renders local control measures useless.

A comparison of the situation in America with that in Europe from the point of view of livestock epidemic control is pointless, because the bat population density in the affected areas of Europe is far lower than in America, and even there the danger represented by bats is grossly exaggerated in the media[17].

The significance of the rabies danger to humans must be measured according to the number of deaths, the suffering caused by the disease and the level of public hysteria; the significance for wildlife can be seen in the steps taken to control it, the losses of domestic animals and the influence on the population of endangered species[28]. The number of human deaths where European bats are presumed to be the virus transmitters is, at 2, extremely low, whereas public hysteria on the subject of "bat rabies" is considerable. Yet very little has been done to curb the spread of the virus in the way of, for example, vaccination. No losses of domestic animals have been reported and probably have not occurred. The current danger to humans and their domestic animals, therefore, seems negligible, particularly if they are compared to risks from other sources (traffic accidents, nuclear power, pollution, etc.). The influence of the virus on the bat population itself cannot be gauged at the present time.

The simplest, cheapest and most effective method of prevention is, for the vast majority of the population, avoidance of bodily contact with bats. This is generally also in the interests of bat conservation! The most effective control method for potential risk persons and domestic animals is vaccination[8,28], particularly as more than 90% of human rabies cases throughout the world are caused by dogs and cats[45].

If the livestock epidemic control law, which is totally unsuitable for bats, is sensibly used (i.e. not used), there is little likelihood of a conflict between epidemic control authorities and conservationists, especially in view of the fact that the data available to date would in no way justify any measures which would further destroy the already endangered bat population. Cooperation between veterinary surgeons and bat conservationists is to be recommended in order to discuss questions still open.

6. *Practical suggestions*

Personal precautionary measures

The WHO recommends that all those who have regular contact with bats should be immunized against rabies[48]. As innoculation with one of the five vaccines commonly available in Europe (e.g. *Rabipur* or in case of egg albumen allergy *Rabivac* from the Behring Company) has none of the dangers associated with older vaccines and only very minor side effects, it is strongly to be recommended if there is a risk of being bitten.

Prophylactic vaccination with *Rabipur* consists of one intramuscular injection on days 0, 28 and 56, or in accelerated form on days 0, 7 and 28. Reinforcing doses are advisable every one to three years[47].

Those who do not belong to the risk group mentioned above are advised, wherever possible, not to touch bats. If contact cannot be avoided, it is advisable to wear gloves or use a firm piece of cloth for protection.

What to do if bitten

If a person is bitten by a bat suspected of carrying lyssa virus, the first move should be to wash the wound thoroughly with soap or detergent and water. The virus particles, with their high lipid content in the outer membrane, are thus inactivated. 70% alcohol or other skin disinfectant should then be applied.

Post-exposure vaccination against rabies is still advisable which, in the case of *Rabipur*, consists of a course of 6 injections on days 0, 3, 7, 14, 30 and 90. If bites have occurred on or near the head (neck, face), or if there has been mucosa contact with the animal's saliva, rabies hyperimmunoglobulin should also be administered as passive immunization[47].

If the animal which gave the bite is later proved virus-negative the course of vaccination may be stopped.

Examination of suspected lyssa-carrying bats

Bats suspected of carrying lyssa virus may be examined by the veterinary authorities (information should be provided on enquiry). It ought to be pointed out, however, that as a rule only bats found dead or in a weakened condition should be examined for the virus. If an animal is proved virus-positive following a fluorescence test, it should be sent to the appropriate WHO rabies centre (e.g. the Federal Republic of Germany in Tubingen), where the virus can be specified by means of monoclonal antibodies. It is important to identify the species of bat involved in each case.

Capturing healthy bats or bat colonies (even if lyssa virus has been established in one member of the colony) is a completely pointless step which clearly infringes existing nature protection laws. According to the new nature protection law in the Federal Republic of Germany (species protection amendment of 10 December 1986), not only the bats themselves are protected, but also their habitats (20d/1/3), which means that the destruction of bat habitats or the prevention of access to them is not permissible without due cause.

Public awareness

Efforts at public enlightenment should be made without any hint of sensationalism and in cooperation with bat researchers and veterinary

surgeons. The need for bat conservation should be emphasized, particularly to the press. All data so far available provide no grounds for epidemic control measures out of keeping with the requirements of bat conservation: one-sided "enlightenment" is therefore unjustifiable.

In the Federal Republic of Germany, an information sheet clearly summarizing the most important points for the lay reader was prepared for the press (see Appendix 1). Concise information of this type could be handed out, for example, upon enquiry by newspaper reporters.

Acknowledgements

I would like to thank the following for the contributions in my search for information: P.C. Grauballe (Copenhagen, Denmark); Dr Hoppe (Kiel); Dr A. King (New Haw, UK); A. Mare (Malzeville, France); Dr W. Muller (Tubingen, FRG); Dr E. Neuvonen (Helsinki, Finland); Dr H. Roer (Bonn, FRG); Dr M. Seidler (Hanover, FRG); Professor Dr M.A. Selimov (Moscow, USSR); Dr D. Seroka (Warsaw, Poland); Dr I. Wandeler (Berne, Switzerland); Dr Wernery (Neumunster, FRG). Special thanks are due to Professor D. W. Muller-Ruchholtz (Kiel, FRG) and Dr H. Baagoe (Copenhagen, Denmark) for their critical comments on the manuscript. Mrs Horton-Kruger (Kiel, FRG) prepared the English translation of the German text.

References

[1] Anderson, R.M. (1986) Vaccination of wildlife reservoirs. *Nature, London* 322, 304-305.

[2] Anon. (1983) Rabies. *Lancet* 30 April, 967.

[3] Aubert, M.F.A. (1982) Essais de transmission de la rage au furet par und ixodide: *Pholeoixodes rugicollis* (Schulze et Schlottke, 1929). *Acarologia* 23, 125-132.

[4] Baagoe, H.J. (1986) European bats and bat rabies. WHO Informal Discussion on Bat Rabies in Europe Marburg, May 5-6. Manuskript.

[5] Barnard, B.J.H. (1979) The role played by wildlife in the epizootiology of rabies in South Africa and South-West Africa. *Onderstepoort J. vet. Res.* 46, 155-163.

[6] Bektimirov, T.A. and Assaad, F. (1984) Vaccination status of human viral diseases. In: 'Applied Virology' (Ed. E. Kurstak), Academic Press, Orlando. pp. 131-158.

[7] Bitsch, V. *et al.* (1986) Bat rabies - Europe. *MMWR* 35, 430-432.

[8] Bundesgesundheitsamt, Ed. (1985) Tollwut. *Bundesgesund hbl.* 28(5), 150-155.

[9] Clark, H.F. and Wiktor, T.J. (1972) Rabies virus. In: 'Strains of Human Viruses' (Eds M. Majer and S.A. Plotkin), Karger, Basel-Munchen. pp. 177-182.

[10] Constantine, D.G. (1962) Rabies transmission by non-bite route. *Publ. Hlth. Rep. (USA)* 77, 387-389.

[11] Constantine, D.G. (1966) Transmission experiments with bat rabies isolates: responses of certain carnivora to rabies virus isolated from animals infected by non-bite route. *Amer. J. Vet. Res.* 27, 13-15.

[12] Danish State Veterinary Service (1986) Information on bat rabies in Denmark. Manuskript. 11 S.
[13] Editors (1962) Transmission of rabies without biting. *J. Bomb. Nat. Hist. Soc.* 59, 283-285.
[14] Eichwald, C. (1965) 'Die Tollwut'. Hirzel, Leipzig.
[15] Fagbami, A.H. *et al.* (1981) Hospital records of human rabies and antirabies prophylaxis in Nigeria, 1969-1978. *Trans. R. Soc. Trop. Med. Hyg.* 75, 872-876.
[16] Grauballe, P.C. *et al.* (1987) Bat rabies in Denmark. *Lancet* 14, Febr. 379-380.
[17] Greenhall, A.M. (1982) House bat management. *Res. Publ. No. 143. US Fish and Wildl. Serv.* Washington DC.
[18] Harry, T.O. *et al.* (1985) Rabies in Nigeria. *Lancet* 14, September, 601-602.
[19] Hummeler, K. (1971) Bullet-shaped viruses. In: 'Comparative Virology' (Eds K. Maramorosch and E. Kurstak), Academic Press, New York-London. pp. 361-386.
[20] Kaplan, M.M. and Koprowski, H. (1980) Rabies. *Sci. Amer.* 242, 120-134.
[21] Kauker, E. (1966) Die Tollwut in Mitteleuropa von 1953 bis 1966. Sitzungsber. Heidelb. Akad. Wiss., Math.-nat. Kl. 1966, 205-232.
[22] Kauker, E. (1975) Vorkommen und Verbreitung der Tollwut in Europa von 1966 bis 1974. Sitzungsber. Heidelb. Akad. Wiss., Math.-nat. Kl. 1975, 1-44.
[23] Kuwert, E. and Scheiermann, N. (1984) Rhabdoviren. In: 'Lehrbuch der Medizinischen Mikrobiologie' (Eds. H. Brandis and H.J. Otte), G. Fischer, Stuttgart-New York. pp. 653-658.
[24] Lafon, M. *et al.* (1986) Human rabies vaccines induce neutralising antibodies against the European bat rabies virus (Duvenhage). *Lancet* 30, August, 515.
[25] Lumio, J. *et al.* (1986) Human rabies of bat origin in Europe. *Lancet* 15, February, 378.
[26] Meredith, C.D. *et al.* (1971) An unusual case of human rabies thought to be of chiropteran origin. *S. Afr. Med. J.* 45, 767-769.
[27] Meredith, C.D. and Standing, E. (1981) Lagos bat virus in South Africa. *Lancet* 11, April, 832-833.
[28] Murphy, F.A. *et al.* (1984) Genetic engineering in animal vaccine production. In: 'Applied Virology' (Ed. E. Kurstak), Academic Press, Orlando. pp. 17-30.
[29] Pacer, R.E. *et al.* (1985) Rabies in the United States and Canada, 1983. *MMWR* 34, 11-27.
[30] Pattison, J.R. (1983) Rabies - a growing threat. Van Nostrand Reinhold, Wokingham.
[31] Perry, B.D. (1987) The control of rabies. *TREE* 2, 30-31.
[32] Pitzschke, H. (1965) Tollwut bei einer Breitflugel-Fledermaus (*Eptesicus serotinus*) in Thuringen. *Zbl. Bakteriol. Parasitenk. Infektionskrh. Hyg.* 196, 411-412.
[33] Roer, H. (1964) Ist eine Ubertragung der Tollwut durch Fledermause in Europa zu befurchten? *Myotis* 2, 17-18.
[34] Roer, H. (1967) Zur Frage der Tollwutubertragung durch insektenfressende Fledermause. *Myotis* 5, 12-14.

[35] Roer, H. (1969) Zur Frage der Tollwutubertragung durch insektenfressende Fledermause. *Myotis* 7, 17-18.
[36] Schneider, L.G. (1982) Antigenic variants of rabies virus. *Comp. Immun. Microbiol. infect. Dis.* 5, 101-107.
[37] Seidler, M. *et al.* (1987) Tollwut bei Fledermausen in Niedersachsen. Manuskript.
[38] Shope, R.E. (1982) Rabies-related viruses. *Yale J. Biol. Med.* 55, 271-275.
[39] Shope, R.E. (1984) Rabies enigma: human and animal disease control. In: 'Applied Virology' (Ed. E. Kurstak), Academic Press, Orlando/Florida. pp. 231-240.
[40] Smith, P.C. *et al.* (1968) Isolation of rabies virus from fruit bats in Thailand. *Nature, London* 216, 384.
[41] Stougaard, E. (1983) Denmark again free from rabies. *Rabies Bull. Europe* 1, 4-5.
[42] Sturchler, D. (1981) Endemiegebiete tropischer Infektionskrankheiten. Huber, Bern-Stuttgart-Wien.
[43] Tignor, G.H. and Shope, R.E. (1972) Vaccination and challenge of mice with viruses of the rabies group. *J. Infect. Dis.* 125, 322-324.
[44] Tignor, G.H. *et al* (1977) Duvenhage virus: Morphological, biochemical, histopathological and antigenic relationships to the rabies serogroup. *J. Gen. Virol.* 37, 595-611.
[45] Turner, G.S. (1984) Rabies. In: 'Virology. Vol. 4. Principles of Bacteriology, Virology and Immunity' (Eds F. Brown and Sir G. Wilson), E. Arnold, London. pp. 472-486.
[46] Tuttle, M.D. and Kern, S.J. (1981) Bats and public health. *Contrib. Biol. Geol.* 48, 1-11.
[47] WHO (1984) WHO Expert Committee on Rabies. Seventh Report. WHO, Genf.
[48] WHO Collaborating Centre for Rabies Surveillance and Research (O.J.): Informationen zur Fledermaustollwut in Europa. *Merkblatt* 7 S.
[49] WHO Collaborating Centre for Rabies Surveillance and Research (1985). *Rabies Bull. Europe* 1/85.
[50] WHO Collaborating Centre for Rabies Surveillance and Research (1985). *Rabies Bull. Europe* 2/85.
[51] WHO Collaborating Centre for Rabies Surveillance and Research (1985). *Rabies Bull. Europe* 4/85.
[52] WHO Collaborating Centre for Rabies Surveillance and Research (1986). *Rabies Bull. Europe* 1/86.
[53] WHO Collaborating Centre for Rabies Surveillance and Research (1986). *Rabies Bull. Europe* 2/86.
[54] WHO Collaborating Centre for Rabies Surveillance and Research (1986). *Rabies Bull. Europe* 3/86.
[55] WHO Collaborating Centre for Rabies Surveillance and Research (1986). *Rabies Bull. Europe* 4/86.
[56] Wiesmann, E. (1974) Medizinische Mikrobiologie. Thieme, Stuttgart.
[57] Woodruff, A.W. (Ed.) (1974) Medicine in the Tropics. Ch. Livingstone, Edinburgh-London.

Appendix 1: Press Bulletin

Bats are the most endangered mammal species in Central Europe. Bat conservation is therefore one of the most urgent tasks of wildlife protection.

All native bat species had to be entered in the Red List of endangered flora and fauna in the Federal Republic of Germany. The bats (including their roosts) are therefore now under protection by regional and national laws.

Bat conservation measures must take into consideration the differing habitats and requirements of the animals throughout the year: summer and winter roosts and feeding grounds.

As all native bat species feed exclusively off insects, extensive green areas in villages/suburbs, forests and fallow land are also important for their existence.

In Europe bats have no significance as pests or carriers of disease. In recent months, however, a new type of virus has been found in a few bats, mainly in northern Germany and Denmark, for which there is as yet very little scientific data.

The virus has a certain similarity with a South African virus strain of the Duvenhage type, which is in the rabies-related group of viruses. It is not, however, identical with the virus which causes wild and domestic animal rabies in this part of the world.

In Central Europe there has been no case of rabies in either domestic animals or humans which could be connected with the bat virus - although the virus was identified in several bats as early as the 1950's.

On the basis of the available data it is presumed that the virus is present in less than 0.5% of all native bats. Examinations to date (of more than 1000 bats) have found the virus only in the serotine bat, one of 22 species found in this region of Europe.

The World Health Organization recommends the following precaution: Anyone who has regular contact with bats or who is bitten while handling a bat should be vaccinated for safety's sake.

The only relevant source of infection for humans is the handling of bats found on the ground. There is no risk from bats in flight hunting insects, or from bat colonies.

FRUIT LOSSES FROM FLYING-FOXES IN QUEENSLAND

G.I. Jamieson

Horticulture Branch, Queensland Department of Primary Industries,
GPO Box 16, Brisbane, Qld 4001.

Abstract

Flying-foxes cost Queensland tree fruit growers 3 per cent of their production, valued at around $20m, over the three years from 1984/85 to 1986/87. Fruit losses varied widely between seasons, crops and localities. Flying-foxes were most destructive during the 1986/87 fruit season, causing 3.6 per cent crop losses, worth about $8.3m. Lychees suffered the heaviest losses (22.3 per cent) over the three-year survey period, and lowchill stonefruit losses reached 24.6 per cent in 1986/87. The vine fruit crops, grapes, kiwifruit and passionfruit were completely ignored by flying-foxes.

It is noteworthy that fruit-eating birds were almost as damaging to the fruit industry as were flying-foxes. Over the three-year survey period, growers estimated that birds caused fruit losses averaging 2.1 per cent of production, worth around $15m. All fruit crops were attacked by birds and there was little variation in the extent of damage from one year to the next.

These were some of the major findings of a comprehensive survey questionnaire mailed to 550 randomly selected Queensland fruit growers in 1987. Over 73 per cent of contacted growers responded to the survey.

Introduction

Flying-foxes, otherwise known as fruit bats, have always been a thorn in the side of Queensland's fruit growers. They feed most actively on commercial fruit crops in those years when seasonal conditions reduce flowering and fruiting of the bush trees that are their normal food source.

However, to date, the only serious study of the problem was reported by Ratcliffe (1931). He concluded from his study that fruit losses from flying-foxes were generally not very severe, in spite of significant losses in some localities in some seasons.

Since Ratcliff's study, complaints from fruit growers have continued and have probably become more vociferous. Fruit losses caused by flying-foxes could have increased over the years through:

- the clearing of natural food trees in the development of grazing and farming lands;
- expansion of the fruit industry into new areas;
- the introduction of new fruit crops.

In desperation, fruit growers have tried a wide range of procedures in attempts to protect their crops from flying-foxes. Some of

the methods tried have been partially successful; some have been unethical and illegal; some of the special equipment they have tried has proved to be an expensive white elephant. But no generally economical and reliably effective method of protecting fruit crops from flying-foxes has yet been devised.

Responding to fruit growers' pleas for help, the Queensland Government in 1985 removed the four fruit-eating species of flying-foxes from the protected list. These species are *Pteropus scapulatus* (little red), *P. poliocephalus* (grey-headed), *P. alecto* (black) and *P. conspicillatus* (spectacled). Animals of these species may now be destroyed without a permit, subject to laws prohibiting cruelty and the dangerous use of the various means of destruction employed.

Obviously, it was necessary to establish the extent and severity of flying-fox damage in the fruit industry. It was also necessary to relate fruit losses caused by flying-foxes to losses caused by other vertebrate pests. These data would be useful in determining the need for further study; they could also be applied to cost/benefit analyses of potential control measures.

Methods

The study was conducted by means of a mailed survey questionnaire. The questionnaires were mailed in June 1987 to 550 growers selected at random from lists of 3054 commercial banana, deciduous fruit and "other fruit" growers from Queensland. The lists were provided confidentially by the Committee of Direction of Fruit Marketing (COD), a cooperative of all of Queensland's commercial fruit and vegetable growers. Citrus, pineapple and vegetable growers were not included in the survey as it was considered that their crops were not susceptible to damage by flying-foxes.

Reminders were mailed to non-respondents one month after the first mailing.

The questionnaire covered the previous three fruit seasons - 1984/85, 1985/86 and 1986/87. Among other objectives, its design allowed:

- an estimation of the relative importance of various vertebrate pests;
- calculation of percentage fruit losses caused by these pests to all tree and vine fruit crops on a regional and state-wide basis;
- determination of the type of crop damage caused by flying-foxes.

Calculations of fruit loss percentages on a regional or state-wide basis were based on (a) the stated or calculated value of fruit produced by each grower and (b) the grower's estimate of percentage crop losses caused by flying-foxes, birds, possums and other vertebrate pests. As participants in the survey were randomly selected from Queensland's total population of commercial banana, deciduous and "other fruit"

growers, conclusions from the survey results could be ascribed on a state-wide industry basis.

Figure 1 shows the boundaries of the regions of Queensland which were used to interpret the results of the survey: far north, north, central and south Queensland and the Granite Belt. Because of its elevation and latitude, the Granite Belt is Queensland's only pomefruit producing district.

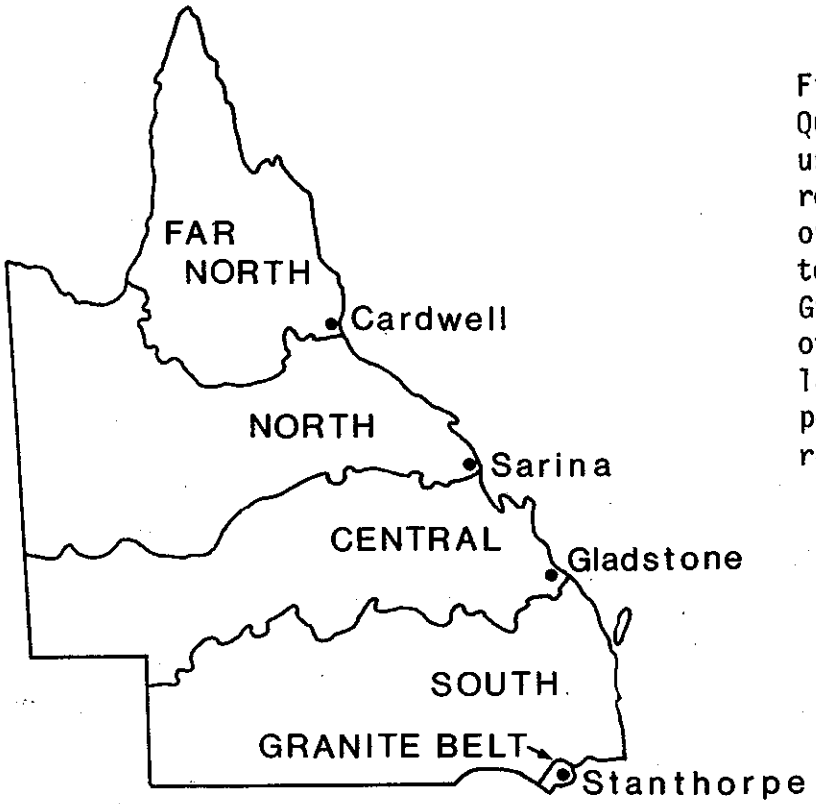


Figure 1: Regions of Queensland which were used to interpret the results of the survey of flying-fox damage to fruit crops. The Granite Belt, because of its elevation and latitude, is the only pomefruit producing region of Queensland.

Results and Discussion

Over 73% of contacted growers responded to the survey, and many wrote covering letters expanding on the data they had supplied.

Table 1 shows production losses caused by vertebrate pests to the most common crops. Losses caused by flying-foxes are shown for each of the three years covered by the survey; for birds and other vertebrate pests (including possums), only the total losses over the three-year period are shown. Where differences were apparent in percentage fruit losses caused by flying-foxes, data are shown for the various regions of the State.

Table 2 summarises the reported frequency of various types of damage caused to fruit crops by flying-foxes.

Table 1: Fruit production losses from vertebrate pests - major crops 1984-87.

Crop	No. of Reports	Region	Production Losses (%)					
			Flying-foxes				Birds	Other Vertebrates Pests
			1984-85	1985-86	1986-87	1984-87	1984-87	1984-87
All crops	459	Queensland	2.5	2.5	3.6	3.0	2.1	0.3
Avocado	62	Queensland	0.2	0.3	0.1	0.2	0.6	0.2
	44	South	0.3	0.3	0.3	0.3	1.2	0.4
	18	Other	0.0	0.3	0.1	0.1	0.1	0.0
Banana	72	Queensland	0.9	1.2	1.0	1.0	1.3	0.4
	33	South	4.6	4.3	6.2	5.1	3.8	0.7
	39	Other	0.1	0.1	0.2	0.2	0.8	0.4
Citrus	8	Queensland	0.9	2.4	2.3	1.8	2.2	0.0
Custard apple	20	Queensland	1.3	1.3	1.5	1.4	0.8	0.0
Grape	16	Queensland	0.0	0.0	0.0	0.0	9.6	0.8
Kiwifruit	6	Queensland	0.0	0.0	0.0	0.0	0.3	0.0
Lychee	24	Queensland	25.9	20.0	20.3	22.3	1.0	2.0
	5	South	1.0	25.6	12.3	19.2	1.1	1.0
	8	Central	0.5	17.9	29.1	12.7	1.5	0.0
	6	North	39.4	46.1	20.3	35.3	0.0	0.0
	5	Far North	5.7	4.9	0.0	4.8	2.8	10.0
Mango	46	Queensland	7.4	5.8	6.2	6.4	0.7	0.0
	7	South	0.4	1.2	1.7	1.5	1.1	0.2
	12	Central	5.4	12.7	5.7	8.0	1.8	0.0
	19	North	9.6	4.4	7.7	6.8	0.5	0.0
	8	Far North	2.2	2.0	3.1	2.4	0.1	0.0
Papaw	39	Queensland	7.1	8.8	9.1	8.5	11.4	0.4
	20	South	2.7	7.1	0.5	3.6	7.9	0.2
	12	Central	8.6	9.8	15.4	11.4	14.3	0.0
	1	North	25.0	33.3	3.3	20.6	0.0	5.3
	6	Far North	0.2	0.1	3.5	1.8	0.3	0.0
Passion-fruit	4	Queensland	0.0	0.0	0.0	0.0	2.0	6.0
Persimmon	7	Queensland	0.0	0.2	26.8	0.8	0.2	0.0
Pomefruit	61	Queensland	3.8	2.3	10.8	6.9	3.4	0.0
Stonefruit	104	Queensland	5.7	3.9	9.1	6.6	1.0	0.4
	66	Granite Belt	5.7	4.0	7.5	6.0	1.1	0.5
	38	Other	5.9	3.5	24.6	12.6	0.3	0.0

Flying-foxes cost Queensland's fruit growers an annual average of almost $7 million (3 per cent of their production) over the three years from 1984/85 to 1986/87. However, losses varied considerably between seasons, crops and localities, with the heaviest losses of 3.6 per cent in 1986/87 (Table 1).

Table 2: Type of flying-fox damage to fruit crops.

Crop	Reports		Percentage of all damage reports (%)						
	Number received	Percentage of crops damaged (%)	ate ripe fruit	ate green fruit	punctured fruit	stained fruit	damaged blossom	broke branches	knocked fruit down
All crops	459	55	25	15	24	16	3	15	2
Avocado	62	21	38	23	23	15	0	0	0
Banana	72	50	20	12	30	25	8	2	1
Black sapote	1	0	0	0	0	0	0	0	0
Carambola	1	0	0	0	0	0	0	0	0
Citrus	10	50	40	0	20	10	0	0	0
Custard apple	20	35	20	20	40	20	0	0	0
Fig	2	100	33	33	33	0	0	0	0
Grape	16	0	0	0	0	0	0	0	0
Guava	1	100	50	50	0	0	0	0	0
Kiwifruit	8	0	0	0	0	0	0	0	0
Lychee	24	75	34	22	24	10	0	10	0
Mango	46	63	33	19	26	11	6	5	0
Papaw	39	74	26	7	23	17	8	17	2
Passionfruit	6	0	0	0	0	0	0	0	0
Peachpalm	1	100	50	0	50	0	0	0	0
Persimmon	7	86	30	10	20	0	10	20	0
Pomefruit	61	72	23	10	23	19	1	19	5
Stonefruit	105	72	22	17	22	16	1	20	1
Tamarillo	2	50	50	0	0	0	0	50	0

Table 1 also shows that fruit-eating birds were only slightly less damaging than flying-foxes, costing growers 2.1 per cent of their production over the three-year survey period. Each year, the level of bird damage was more consistent than the damage caused by flying-fox, which tended to vary widely between seasons.

Table 2 shows that 55 per cent of all fruit crop plantings suffered at least some damage from flying-foxes during the three years surveyed. Flying-foxes most commonly ate ripe fruit and punctured fruit, but they also ate green fruit, stained the fruit and broke branches. Less frequently, they knocked fruit down and damaged blossoms.

Lychees were the fruit crop that was hardest hit by flying-foxes, with 75 per cent of the State's crops attacked and average annual fruit losses of 22 per cent. In contrast, birds caused only 1 per cent fruit losses over the three-year survey period.

Flying-fox damage was most severe in north Queensland with 35 per cent fruit losses. South Queensland growers lost 19 per cent of their fruit and there were 13 per cent losses in central Queensland. Far north Queensland growers reported only 5 per cent losses during the three-year survey period.

Most commonly, flying-foxes ate and punctured lychee fruit, but they also stained the fruit and broke branches.

Stonefruit. Flying-foxes were also very damaging to stonefruit crops. They attacked over 72 per cent of orchards causing State-wide fruit losses of almost 7 per cent. Birds caused only 1 per cent fruit losses, with most damage suffered on the Granite Belt.

Flying-foxes attacked stonefruit crops heavily in 1986/87, especially in districts other than the Granite Belt. While losses in that season were almost 8 per cent on the Granite Belt, flying-foxes cost stonefruit growers in other districts almost 25 per cent of their production. Twenty-one of these respondents specified that they grew peaches and nectarines; they reported that flying-foxes cost them 80 per cent of their fruit production in 1986/87, but less than 4 per cent in the other two seasons surveyed.

Growers reported that flying-foxes ate both green and mature stonefruit, punctured and stained the fruit and also broke branches.

Papaws were also hard hit by flying-foxes, with 74 per cent of plantings attacked and State-wide fruit losses of 8.5 per cent over the survey period.

Birds were more damaging to papaw crops than flying-foxes, causing over 11 per cent losses to the State's crops. The most severe losses were recorded in central Queensland (14 per cent) and South Queensland (11 per cent), but losses from birds were insignificant in other papaw producing regions.

Flying-foxes caused consistently heavy losses in central Queensland papaw crops. Losses averaged over 11 per cent over the three-year survey period and over 15 per cent in 1986/87. The only report from north Queensland showed average fruit losses of almost 21 per cent over the survey period, including relatively minor losses in 1986/87. Losses reported from far north and south Queensland were far less severe, although fruit losses in south Queensland exceeded 7 per cent in 1986/87.

Most commonly, flying-foxes ate ripe fruit and also punctured and stained the fruit. They also snapped off the leaves, which exposed the fruit to sunburn. Many growers commented that fruit harvested green-mature was not attacked by flying-foxes or birds.

Pomefruit. Queensland's pomefruit production is centred on the Granite Belt. The 1987 apple harvest was reduced by 11 per cent as the result of attacks by flying-foxes, with a three-year average of 7 per cent. Pear crops were not as badly affected, averaging only 2 per cent loss.

Birds caused a consistent annual loss of 3 per cent of apple and pear production over the three-year period.

The most common damage caused by flying-foxes to apple and pear crops was the eating of ripe fruit, puncturing and staining fruit and breakage of branches. One grower commented that his close-planted dwarf apple trees suffered no damage from flying-foxes, whereas he had heavy losses in the past, when he grew tall wide-spaced trees.

Mangoes. Flying-foxes attacked 63 per cent of all mango orchards during the three seasons covered by the survey, and cost the industry over 6 per cent of fruit production.

Flying-foxes caused heavy fruit losses in central Queensland (8 per cent), with a peak of almost 13 per cent in 1985/86.

Most frequently, flying-foxes ate and punctured the mangoes, but preferred ripe fruit. This suggests that frequent harvesting may reduce the losses. One grower commented that he had to harvest early to avoid damage by flying-foxes, causing him to miss the late high-priced market.

Production losses from birds were generally light to minor, except in central Queensland, where birds caused 2 per cent losses in 1984/85 and 3.3 per cent loss in 1985/86.

Bananas. South Queensland banana growers suffered over 5 per cent losses from flying-foxes, but losses in the rest of the State were negligible. Probably the southern losses were largely in Lady Fingers which are not grown in other parts of the State; but few growers reported the variety they grew.

Puncturing and staining fruit were the most frequently reported types of damage, but flying-foxes often ate the fruit, especially when it was ripe.

Comments were made that the most serious losses were due to down-grading of the quality of spoiled fruit. A number of growers also complained that flying-foxes damaged the bunch covers.

Birds caused fruit damage in all banana growing regions of the State. This was most evident in south Queensland with fruit losses of almost 4 per cent over the three-year survey period. Most of this damage was due to scratching before the bunch covers were applied.

Persimmons. Persimmon growers reported heavy losses of 27 per cent in 1986/87, following negligible losses in the two previous seasons. Mostly, flying-foxes ate the ripe fruit, punctured fruit and broke branches.

Birds caused further fruit losses of 9 per cent in 1986/87, but no damage was reported during the two previous seasons.

Figs. Only two fig growers were in the survey, but their losses from flying-foxes averaged around 50 per cent. Flying-foxes ate and punctured the fruit. Reported bird damage was negligible.

Lightly damaged crops. Avocados suffered negligible losses from flying-foxes. Birds were generally only a minor problem, but they caused over 1 per cent crop loss in south Queensland.

Flying-foxes cost custard apple and rollinia growers over 1 per cent of production and similar losses were caused by birds.

Although apricot growers reported that flying-foxes cost them less than 1 per cent of their production, one grower commented that he would expect losses of around 25 per cent if the fruit were left to ripen on the trees.

On average, flying-foxes cost pear growers 2 per cent of their production, including 3 per cent in 1986/87. Another 3 per cent of their crops were destroyed by birds during each of the three years surveyed.

Avocado, custard apple, rollinia, apricot and pear crops all suffered similar types of damage from flying-foxes; they ate the fruit, both green and ripe, and caused puncturing and staining. Commonly, they also broke the branches of pear trees.

Although citrus growers were not specifically surveyed, eight respondents reported that flying-foxes cost them almost 2 per cent of their citrus fruit production over the three-year survey period. Usually, flying-foxes ate ripe fruit (but not green fruit) and punctured the fruit. Occasionally they caused staining. Birds consistently caused further losses of around 2 per cent during each of the three years surveyed.

The following comments on the remaining lightly damaged crops cannot be considered very reliable, as only one or two growers of each responded to the survey.

No significant damage by flying-foxes was reported by guava, carambola, black sapote, cherry or quince growers. One tamarillo crop suffered 0.3 per cent fruit loss after flying-foxes were attracted to fruit prematurely ripened by hail damage. One casimiroa grower responded, reporting 11 per cent losses from flying-fox damage in 1986/87.

Undamaged crops. No flying-fox damage of any kind was reported by growers of the vine fruit crops, passionfruit, grapes and kiwifruit. However, birds caused heavy losses to grape growers, averaging almost 10 per cent of annual production. Birds destroyed 2 per cent of the passionfruit crop over each of the three years surveyed. Bird damage cost an average of 0.3 per cent of the kiwifruit crop, including 1 per cent loss in 1984/85.

Reference

Ratcliffe, F.N. (1931) The flying-fox (*Pteropus*) in Australia. *Bull. Counc. Sci. Ind. Res. Aust.* 53, 1-80

SHORT COMMUNICATION

PARTIAL ALBINO *MORMOPTERUS PLANICEPS* (MOLOSSIDAE)

W.N. Holsworth

13 Nabilla Crescent, Bendigo, Victoria 3550.

On 4 December 1987 a pole replacement crew of the State Electricity Commission found two Little Mastiff-bats, *Mormopterus planiceps*, under the metal cap of a pole near Strathfieldsaye, Victoria (about 7 km east of Bendigo). One of the bats was a normally coloured male; the other, a female, had all the hair on the body, wings, interfemoral membranes and feet, pure white. The hairless parts of the body, that is, the ears, face, feet, tail and the membranes, had the normal dark grey colouration. Both animals have been deposited with the National Museum of Victoria, Melbourne (registration numbers: ♂ C27329; ♀ C27330).

Setzer (1950) noted that albinistic bats were rare but had been recorded from every continent except Australia. McCoy (1960) reported the first records of true albinos in the genus *Tadarida* (closely related to *Mormopterus*, but notes that Glass, in 1952 reported on several Mexican free-tailed bats *Tadarida brasiliensis mexicana* which had scattered patches of colourless hair). McCoy, collecting in the same area as Glass, collected two individuals that completely lacked pigment in the body hair but had normally pigmented skin. In 1957 and 1958 they collected two pure albinos and one with only pigmented eyes. Herreid and Davis (1960) examined over 47,000 Mexican free-tailed bats in Texas in 1957 and found 149 with white patches of fur on the back, one with white spots over most of the body, and five with outstanding marks on the abdomen. They caught one true albino in 1959 in a different area.

The only reference to an albino bat in Australia that I am aware of is a note by Swanson (1980) of an albino common sheath-tailed bat *Taphozous georgianus*. However, colour variations in *Mormopterus* are not uncommon. Together with members of the Bendigo Field Naturalists Club, I caught and tagged 250 *M. planiceps* in Bendigo (Holsworth 1986) and recorded some variations in the colour pattern of 17 individuals. Our tagging notes refer to 14 (13 females and 1 male) as being light coloured, light grey or light brown. One male was recorded as silver grey, another was recorded as having a white belly. One female had a very pale grey back and belly contrasing markedly with the dark ears and wings.

References

Herreid II, C.F. and Davis, R.B. (1960) Frequency and placement of white fur on free-tailed bats. *J. Mammal.*, 41, 117-119.

Holsworth, W.N. (1986) Homing ability of the little mastiff-bat *Mormopterus planiceps*. *Macroderma*, 2, 54-58.

McCoy, C.J. (1960) Albinism in *Tadarida*. *J. Mammal.*, 41, 119.

Setzer, H.W. (1950) Albinism in bats. *J. Mammal.*, 31, 350.

Swanson, S. (1980) An albino common sheath-tailed bat *Taphozous georgianus* from the Arnhem land escarpment. *N.T. Nat.*, 1, 6.

BOOK REVIEW

A GUIDE TO THE BATS OF SOUTH AUSTRALIA

by T.B. Reardon and S.C. Flavel (1987)

South Australian Museum, Adelaide.

It is with pleasure that I review this booklet, as I have keenly followed its progress over the three years or so since its inception. Both Terry and Stan vastly extended their local knowledge of bats after being flung out Australia-wide, with "Bats of Eastern Australia" in hand, conducting the Watts/Baverstock biochemical-taxogenetic field trips of the early 1980's. This forced training, using primitive tools, has not only shown them the intricacies of bat identification but, more importantly, what the essentials of a good field bible should be.

"Bats of South Australia" not only gives every clue to identifying the 20 species of bats in that state (including five of the nine *Eptesicus* now with us) but also treats the essential basic biological information concisely and with well explained biological precision. Other sections describe methods of catching, handling and measuring bats, all of which are well illustrated and, therefore, leave no room for misunderstandings.

The first third of the 80 or so pages is written in a light but serious vein which is occasionally broken by comments that indicate the authors' sense of humour. For example, in warning the reader about recognising late pregnancies by abdominal enlargement they state "but take care as one of us, in our early days, identified a well fed male Gould's wattled bat as being pregnant". Even with the "early days" proviso, I'm not sure if I would confess to that error! Each chapter in this first section is introduced with a humorous graphic, one of which is reproduced below (Figure 1). Through this section, the novice receives an excellent introduction to general bat biology.

Figure 1: Figure introducing the chapter "Getting Around in the Dark" in "A Guide to the Bats of South Australia".

The next section is an illustrated field key, colour plates and species accounts. The field key relies solely on external characters and, particularly for South Australia, is quite straightforward, with the exception of the dreaded *Eptesicus* group. Here the authors reluctantly advise that positive identification requires "skull or dental examination, or protein electrophoresis" particularly with females. The glans penis is another diagnostic character for this genus and scanning electron micrographs are provided to illustrate these features described in the text. The *Eptesicus* section of the key is, therefore, quite valuable.

The 27 colour plates are an excellent feature which will attract the layman to learn about our bat fauna. Photographs are a good backup for those who have found a bat by using the text, or as a first identification attempt (the same way that I identify birds!). To be picky I should point out that one of these photographs has been mis-positioned - the cluster of *Miniopterus schreibersii* on page 44 seem to be hanging on tightly in a westerly wind!

The remaining third of the booklet contains Species Profiles which are of a good standard. Half page maps show the distribution of each species in South Australia, with a much smaller supplemental map showing national distribution, the latter being a most useful feature. A handy addition would have been an introductory map of the State showing major habitats and some of the localities mentioned throughout the text.

Each species is well described and, where necessary, the best character to separate them from confusing species is outlined. Occasionally these characters are inadequately explained to the novice; for example, the radio-metacarpal pouches of some of the emballonurids. Nevertheless, the glossary covers most of the difficult terminology. Notes are also provided on the recognition of some species in flight or by the audible component of their echolocation calls.

I was pleased to see the informal description of the two distinct forms within *Mormopterus planiceps* which can be separated by their penis lengths and appearance of the fur. Formal taxonomic separation is obviously in the pipeline, but the authors have at least made their species coverage as comprehensive as possible. Although the authors have "reluctantly omitted" the Ghost bat (*Macroderma gigas*) since it has not been seen in South Australia for 50 years, it may have been wiser to have included a full description. Considering that part of this publication's market obviously includes the general public - a great source of information - some news of Ghost bat distribution may have been forthcoming as this book circulates in the future. There is, however, a small photograph that may help.

A few errors have crept in, unfortunately, these gremlins are always difficult to find during the review of countless drafts:

page 8 *Saccolaimus flaviventris* common name reads "Hill's sheath-tailed bat"

page 8 *Taphozous hilli* common name reads "Yellow-bellied sheath-tailed bat"

page 76 *Eptesicus darlingtoni* distribution should extend to North Queensland, not the NSW/Qld border.

page 73) *Eptesicus finlayeoni* and *E. baverstocki* nomenclatural authors
" 78) are Kitchener, Jones and Caputi 1987, not Kitchener 1987.

In closing, I feel that "A Guide to the Bats of South Australia" will be a useful booklet for bat researchers, particularly those new to this field and those old ones (like myself) that are rusty and inactive with microchiropteran taxonomy. This booklet is available from The Bookshop, South Australian Museum, North Terrace, Adelaide, S.A. 5000, for $10.95 plus $1.50 postage.

Congratulations to Terry and Stan.

[Greg Richards]

N O T I C E S

Bat Conservation

IUCN Red List of Threatened Animals

The International Union for the Conservation of Nature and Natural Resources (IUCN) is a Swiss-based organisation which monitors the conservation of species and ecosystems on a world-wide basis. The findings are published and regularly updated in (among others) RED LISTS of threatened animals and plants. The IUCN Red List of Threatened Animals was revised in early 1988, with input from various Specialist Groups of the Species Survival Commission.

The following bat species were recommended for inclusion in the IUCN Red List by members of the Chiroptera Specialist Group:

ORDER CHIROPTERA

Family Pteropodidae

Pteropus mariannus	Marianas flying-fox	V.	Marianas
Pteropus subniger		Ex.	Mauritius, Reunion
Pteropus niger	Mauritian flying-fox	V.	Mauritius, Carolines, Japan (Riukius)
Pteropus rodricensis	Rodrigues flying-fox	E.	Rodrigues
Pteropus seychellenis aldabrensis	Seychelles fruit bat	V.	Aldabra
Pteropus livingstonei	Comores fruit bat	E.	Comores
Pteropus samoensis	Samoan fruit bat	E.	Samoa
Pteropus pilosus	Palau fruit bat	Ex.	Palau
Pteropus insularis	Carolines fruit bat	I.	Carolines
Pteropus tokudae	Guam flying-fox	Ex.?	Guam
Pteropus phaeocephalus		I.	Carolines
Pteropus macrotis	Big-eared flying-fox	I.	S. New Guinea, Aru Islands
Pteropus molossinus		I.	E. Carolines
Pteropus tonganus	Insular flying-fox	I.	KarKar I., (N.E. New Guinea), Samoa, Cook Is.
Pteropus voeltzkowi	Pemba flying-fox	V.	Pemba
Haplonycteris fischeri		Ex.?	Phillipines

Dobsonia chapmani	Chapman's fruit bat	Ex.?	Negros Is, Phillipines
Dobsonia minor	Lesser Naked-backed fruit bat	Rare	Irian Jaya
Acerodon lucifer	Panay Giant fruit bat	Ex.?	Panay, Phillipines
Styloctenium wallacei	Stripe-faced fruit bat	Rare	Celebes
Megaerops kusnotoi	Javan Tailless fruit bat	Rare	Java
Thoopterus nigrescens	Short-nosed fruit bat	E.	Celebes

Family Emballonuridae

Emballonura semicaudata	Pacific Sheath-tailed bat		Marianas
Emballonura furax	Greater Sheath-tailed bat	Rare	New Guinea, Irian Jaya
Emballonura raffrayana	Raffray's Sheath-tailed bat	Rare	Seram, Irian Jaya
Coleura seychellensis	Seychelles Sheath-tailed bat	E.	Seychelles

Family Megadermatidae

Macroderma gigas	Australian false vampire	V.	Australia

Family Hipposideridae

Rhinonicteris aurantius	Orange Leaf-nosed bat	V.	Australia
Hipposideros papua	Geelvinck Bay Horseshoe bat	Rare	Misori Is.
Hipposideros ridleyi	Singapore Roundleaf Horseshoe bat	I.	Singapore Peninsula, Malaysia
Hipposideros pendleburyi		R.	Southern Thailand

Family Rhinolophidae

Rhinolophus hipposideros	Lesser Horseshoe bat	E.	Europe to Kashmir
Rhinolophus ferrum equinum	Greater Horseshoe bat	E.	N. Africa, Europe
Rhinolophus euryale	Mediterranean Horseshoe bat	R.	Mediterranean
Rhinolophus mehelyi	Meheyli's Horseshoe bat	R.	Mediterranean to Asia
Rhinolophus blasii	Blasius' Horseshoe bat	R.	Southern Europe to Africa

Family Phyllostomatidae

Leptonycteris nivalis	Long-nosed bat	E.	Central America
Leptonycteris sanborni		E.	Central America
Phyllonycteris major	Puerto Rican Flower bat	Ex.?	Puerto Rico
Phyllonycteris poeyi (obtusa)	Cuban Flower bat	V.	Cuba, Hispanolia
Phyllonycteris aphylla	Jamaican Flower bat	V.	Jamaica

Family Myzopodidae

Myzopoda aurita	Golden bat	V.	Madagascar

Family Mystacinidae

Mystacina tuberculata	New Zealand Short-tailed bat	E.	New Zealand
Mystacina robusta	New Zealand Short-tailed bat	E. (Ex.?)	New Zealand

Family Craseonycteridae

Craseonycteris thonglongyai	Kitti's Hog-nosed bat	R.	Thailand

Family Vespertilionidae

Myotis mystacinus	Whiskered bat	V.	Europe
Myotis dasycneme	Pond bat	E.	Europe
Myotis capaccini	Long-fingered bat	V.	Europe
Myotis bartelsi	Bartel's myotis	R.	Java
Myotis hermani	Herman's myotis	R.	Sumatra
Myotis grisescens	Gray bat	E.	USA
Myotis blythi	Blyth's myotis	E.	Europe
Myotis emarginatus	Notched eared myotis	E.	Europe
Myotis myotis	Mouse eared bat	V.	Europe
Myotis bechsteini	Bechstein's bat	V.	Europe
Myotis nattereri	Natterer's bat	V.	Europe

Myotis brandti	Brandt's bat	R.	Europe
Myotis sodalis	Indiana bat	V.	USA
Pipistrellus minahasae	Minahasa pipistrelle	R.	Celebes
Pipistrellus mordax	Fungus pipistrelle	R.	Java
Pipistrellus nathusii	Nathusius' pipistrelle	V.	Europe
Glischropus javanus	Javan thick-thumbed bat	R.	Java
Vespertilio murinus	Particoloured bat	R.	Europe
Nyctalus lasiopterus	Giant noctule	R.	Europe, Asia
Nyctalus leisleri	Leisler's bat	R.	Europe
Barbastella leucomelas	Eastern barbastelle	I.	Caucasus and Sinai to Japan
Barbastella barbastellus	Western barbastelle	R.	Europe
Plecotus austriacus	Grey Long-eared bat	R.	Europe
Plecotus townsendii	Townsend's Big-eared bat	I.	USA
Eptesicus nilssoni	Northern serotine	V.	Europe
Ia io	Great Evening bat	I.	N. India to Vietnam
Euderma maculatum	Spotted bat	V.	N. America
Lasiurus cinereus semotus	Hawaiian Hoary bat	I	USA
Kerivoula africana	Tanzanian Woolly bat	Ex.?	Tanzania
Kerivoula pellucida	Clear-winged bat	E.	Phillipines
Kerivoula jagorii	Forest bat	E.	Phillipines
Kerivoula hardwickei	Hardwick's Forest bat	E.	Phillipines
Phonisus papuensis	Papuan Trumpet-eared bat	V.	Australia
Murina suilla	Brown Tube-nosed bat	R.	N. Australia
Murina florium	Flores Tube-nosed bat	R.	N. Australia

Family Molossidae

Tadarida labiatus		R.	Java
Otomops formosus	Javan Mastiff bat	R.	Java

The status of the various species is indicated by symbols as follows:

EXTINCT (Ex)

Species not definitely located in the wild during the past 50 years (criterion as used by the Convention on International Trade in Endangered Species of Wild Fauna and Flora).

N.B. On a few occasions, the category **Ex?** has been assigned; this denotes that it is virtually certain that the taxon has recently become extinct.

ENDANGERED (E)

Taxa in danger of extinction and whose survival is unlikely if the causal factors continue operating.

Included are taxa whose numbers have been reduced to a critical level or whose habitats have been so drastically reduced that they are deemed to be in immediate danger of extinction. Also included are taxa that may be extinct but have definitely been seen in the wild in the past 50 years.

VULNERABLE (V)

Taxa believed likely to move into the 'Endangered' category in the near future if the causal factors continue operating.

Included are taxa of which most or all the populations are decreasing because of over-exploitation, extensive destruction of habitat or other environmental disturbance; taxa with populations that have been seriously depleted and whose ultimate security has not yet been assured; and taxa with populations that are still abundant but are under threat from severe adverse factors throughout their range.

N.B. In practice, 'Endangered' and 'Vulnerable' categories may include, temporarily, taxa whose populations are beginning to recover as a result of remedial action, but whose recovery is insufficient to justify their transfer to another category.

RARE (R)

Taxa with small world populations that are not at present 'Endangered' or 'Vulnerable', but are at risk.

These taxa are usually localised within restricted geographical areas or habitats or are thinly scattered over a more extensive range.

INDETERMINATE (I)

Taxa known to be endangered, vulnerable or rare, but where there is not enough information to say which of the three categories is appropriate.

Australian members of the Chiroptera Specialist Group are: Dr L.S. Hall, Department of Anatomy, University of Queensland, St. Lucia, Qld 4067; Dr J.E. Nelson, Department of Zoology, Monash University, Clayton, Vic 3168; Dr C.R. Tidemann, Department of Forestry, Australian National University, GPO Box 4, Canberra, ACT 2601; Dr D.P. Woodside, Taronga Park Zoo, PO Box 20, Mosman, NSW 2088.

The Chairman and Secretary respectively of the CSG are: Professor P.A. Racey, Department of Zoology, University of Aberdeen, Aberdeen AB9 2TN, UK and Mr A. Hutson, Fauna and Flora Preservation Society, 8-12 Camden High Street, London NW1 0JH, UK.

Trade in Endangered Species

The Convention on International Trade in Endangered Species of Wild Fauna and Flora (CITES) is an agreement entered into by member countries to prohibit international trade in species (or their products) which are in danger of extinction. Lists of species in various categories are maintained in Appendices to the Convention. Appendix I includes all species threatened with extinction which are or may be affected by trade. Appendix II includes species which are not necessarily threatened with extinction now, but are likely to become so unless trade is strictly regulated.

Following the meeting of the General Assembly of the IUCN in Costa Rica in February 1988, the following bat species were proposed and accepted for amendment to the Convention: *Pteropus mariannus* and *P. tokudae* for inclusion in Appendix I and *P. insularis, P. macrotis, P. molossinus, P. phaeocephalus, P. pilosus, P. samoensis* and *P. tonganus* for inclusion in Appendix II of CITES. This has been in response to a flourishing trade in flying-foxes for human consumption in the Pacific region. For further information readers are referred to: WILES, G.J. and PAYNE, N.H. (1986) The trade in fruit bats *Pteropus* spp. on Guam and other Pacific Islands. *Biol. Cons.* 38, 143-161, for further information - Editor.

Mount Etna

The General Assembly of IUCN at the Costa Rica meeting recognised that: Mount Etna contains caves which provide breeding habitat for the Little Bent-winged bat, *Miniopterus australis* and roosting sites of the vulnerable Ghost bat, *Macroderma gigas*; between 1976 and 1988 the Queensland Government and the Central Queensland Cement Company had successfully negotiated to establish effective legislative protection of 80 per cent of the Mount Etna caves, including the Bat Cleft complex; in 1976 the Fitzroy Caves National Park was established in an area adjacent to Mount Etna, conserving at least 100 caves and their dependent bats.

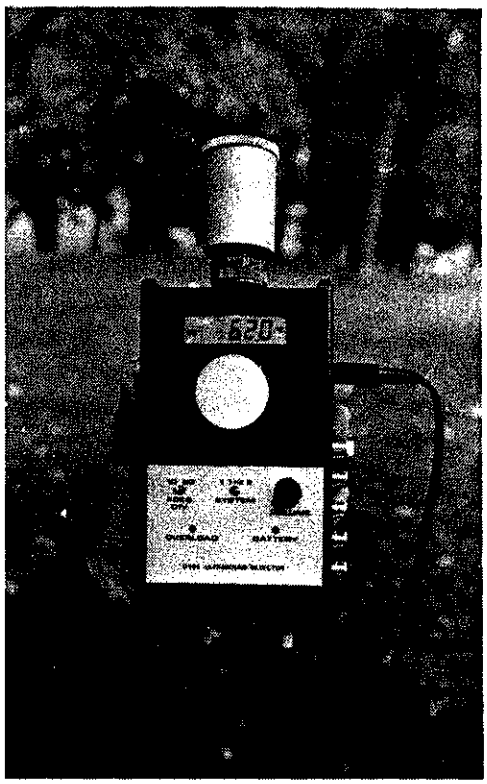
The General Assembly commended the Queensland Government's moves towards protecting a large section of Mount Etna's environs and encouraged its efforts further to protect them. The Assembly expressed concern that limestone mining could be a threat to a number of the caves including Ghost bat roosting sites and called on the Queensland

Government to extend total protection to Mount Etna, its caves and wildlife, and recommended that a long-term, state-wide management strategy for the Ghost bat should be developed.

Details of the more recent developments in this area are contained in the March 1988 issue of 'Ringtail', published by the Queensland National Parks and Wildlife Service in an article entitled 'Trustees to hold Mount Etna Caves'. In summary, the National Parks and Wildlife Service and the Mines Department will be joint trustees of a reserve to be declared over 80% of the area, covering 40 out of 46 cavernous areas on Mount Etna, including Bat Cleft. This area was previously included in a mining lease held by the Central Queensland Cement Company. The reserve will now allow for access of visitors from the south, instead of from the north across the Company's mining lease.

Details of the conservation significance of the caves on Mount Etna, particularly to Ghost bats are given by JOLLY, S. (1987) The mining of Mount Etna - an increasing threat to the Ghost bat, *Macroderma gigas*. *Macroderma* 3, 25-28.

D 940 AND D 960 ULTRASOUND DETECTORS



The D940 and D960 are compact, high performance instruments for serious investigation of ultrasound. A sensitive capacitance microphone with a low noise amplifier makes it possible to detect very weak signals.

Application areas include:

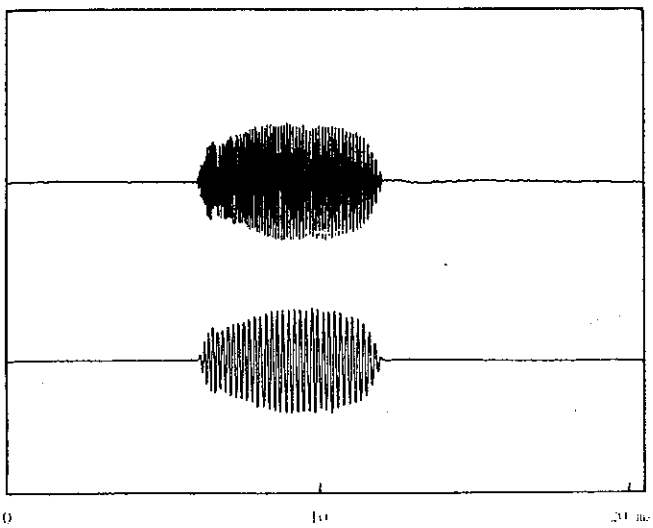
Bioacoustic ultrasound (bats, bush crickets etc.)
Mechanical ultrasound (motors, gears, bearings etc.)
Detection of leaks in pressurized systems
Detection of high voltage discharge

SPECIAL FEATURES:

★ Heterodyne and frequency division systems (D 940)
★ Heterodyne, frequency division and time expansion systems (D 960)
★ Tuned frequency displayed on LCD
★ HF and AF outputs to e.g. tape recorder
★ Overload and battery indicators
★ Small size
★ Low weight

The linear amplitude frequency division system

The frequency division system transforms the entire ultrasonic sound spectrum into the audible range without tuning.
It is possible to select a division factor of 10 or 20, meaning that an original frequency range of 0–200 kHz is transformed to the range 0–20 kHz or 0–10 kHz respectively.
In addition, the amplitude of the original signal is preserved, which is illustrated in the figure below. Since the main characteristics of the signal are retained, a laboratory analysis of a recording of the transformed signals may be very valuable. A standard cassette tape recorder is suitable for such recordings.



The heterodyne system

In this mode a selected frequency range may be transformed into the audible part of the spectrum. The mid frequency of the selected range is displayed on the LCD, and may be varied from 10 to 200 kHz. The display is equipped with a backlight for nocturnal use.
To the user, heterodyning means that the displayed frequency is subtracted from the incoming ultrasonic frequency. If the two frequencies are equal within appr. 5 kHz the resulting signal will be audible.
The heterodyne system is the most sensitive of the three systems, making it suitable to detect very weak ultrasonic signals.
It is also possible to determine the frequency of the signal. This is accomplished by tuning until the output frequency becomes as low as possible (ideally 0 Hz). The reading on the display then equals the frequency of the ultrasonic signal.

The time expansion system (D 960 only)

The principle of this conversion system is similar to that of a tape recorder on which a signal is recorded and then replayed at a reduced speed. However, in this case the memory is a digital memory, and the record-replay sequence is carried out automatically. The time expansion factor is 10, which makes it possible to hear very fine details of the ultrasound, not audible with other conversion systems. Furthermore, this is the only conversion system that retains all characteristics of the original signal, including harmonics. Using a standard cassette tape recorder to record the time expanded signals, enables you to analyze the signals in the laboratory. Apart from the limited length of each "recording" (0.75 s), this gives the same information as that obtained from an instrumentation recorder.
To use the time expansion system, the trigger level is set to a suitable value and the "ARM" push-button is pushed. As soon as the amplitude of the incoming ultrasound exceeds the preset level, the instrument stores the signal in the memory. After 0.75 s, the memory is full and the detector automatically switches to the replay mode. The recorded signal is read out repeatedly until the "ARM" button is pushed again. In order not to loose the beginning of the pulse, a 25% pre-trigger function is included.

Using the detectors

Two of the conversion systems may be monitored simultaneously using a set of stereo headphones. It is of course also possible to monitor only one of the systems. Via the AF outputs (one for each system), the output signals may be recorded on a tape recorder for later analysis. The direct microphone signal is also available at the HF output.
If the user wishes to record spoken comments on a connected tape recorder, this is possible without the use of an extra microphone. A switch on the detector is simply pressed, making the detector microphone signal connected to the heterodyne channel of the tape recorder (the heterodyne signal is temporarily replaced by the microphone signal).
The detectors are also equipped with both overload and battery indicators, as well as a switchable high-pass filter.

Technical specifications

Frequency range:	10-200 kHz freq. div. & het. syst.
	10-150 kHz time exp. system
Display accuracy:	1 kHz
AF outputs:	1 V rms max. (7.5 kohm)
HF output:	500 mV rms max. (1 kohm)
AF output headphones:	max. output power 2×10 mW
	load impedance ≧2×4 ohm
Time expansion system (D 960 only):	
Sampling frequency:	350 kHz
Memory size:	256K×8
Resolution:	8 bits
Record time:	750 ms
Time exp. factor:	10
Pre-trig.:	25%
Current consumption:	
D 940:	typ. 30 mA
D 960:	typ. 30 mA (time exp. syst. off)
D 960:	typ. 44 mA (time exp. syst. on)
Power requirements:	2×IEC 6LF22 (9V)
Size:	
D 940:	180×109×65 mm
D 960:	180×109×80 mm
Weight:	
D 940:	500 g incl. batteries
D 960:	730 g incl. batteries

CURRENT LITERATURE

Breed, W.G. and Leigh, C. 1985
Sperm head morphology of Australian molossid bats with special reference to the acrosomal structure.
Mammalia 49, 403-406.

Burton, R., Kennedy, M. and Fry, I. 1986
The threatened vertebrates.
In: Eds M. Kennedy and R. Burton, 'A Threatened Species Conservation Strategy for Australia - Policies for the Future'.
Ecofund Australia: Manly, New South Wales. Pp. 8-12.

Crichton, E.G. and Krutzsch, P.H. 1985
Comparative morphology of the corpus luteum of embryonic diapause in Chiroptera.
J. Anat. 143, 224. (Abstract)

Christensen, P., Annels, A., Liddelow, G. and Skinner, P. 1985
Vertebrate fauna in the southern forests of Western Australia. A survey.
Forests Dept. W. Aust. Bull. 94, 109 pp.

Churchill, S. 1987
Observations on breeding behaviour in cave-dwelling *Taphozous*.
Macroderma 3, 36-38.

Crossman, D.G. and Reiner, D.S. 1986
Mammals, birds, reptiles and amphibians of the Taroom Shire, central Queensland.
Qd. J. Agr. Anim. Sci. 43, 55-72.

Fry, I. and Kennedy, M. 1986
Correlating habitats with high priority threatened species.
In: Eds M. Kennedy and R. Burton, 'A Threatened Species Conservation Strategy for Australia - Policies for the Future'.
Ecofund Australia: Manly, New South Wales. Pp. 39-42.

Green, R.H., Rainbird, J.L. and McQuillan, P.B. 1986
Food of the southern boobook *Ninox novaeseelandiae*.
Tas. Nat. No. 86, 1-3.

Hand, S.J. 1985
New Miocene megadermatids (Chiroptera: Megadermatidae) from Australia with comments on megadermatid phylogeny.
Aust. Mammal. 8, 5-43.

Hill, J.E. 1985
Records of bats (Chiroptera) from New Guinea, with the description of a new *Hipposideros* (Hipposideridae).
Mammalia 49, 525-535.