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The African Armyworm Handbook

Humanitarian Aid and Civil Protection

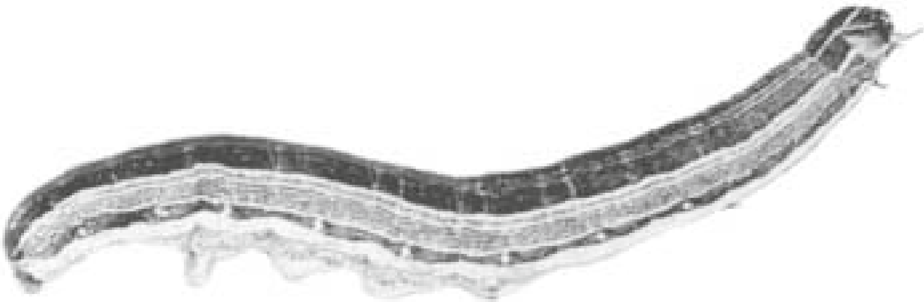
DFID Department for International Development

The African Armyworm Handbook

The Status, Biology, Ecology, Epidemiology and Management of *Spodoptera exempta* (Lepidoptera: Noctuidae)

Second Edition

D. J. W. Rose[1], C. F. Dewhurst[2] and W. W. Page[2]



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The Natural Resources Institute (NRI) of the University of Greenwich is an internationally recognized centre of expertise in research and consultancy in the environment and natural resources sector. The Institute carries out research and development and training to promote efficient management and use of renewable natural resources in support of sustainable livelihoods.

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PREFACE

This is the first revision of *The African Armyworm Handbook* (originally published in 1997) which includes new and updated information and addresses the need to encourage interest from a wider audience as a result of the widespread armyworm outbreaks during the 1998/99 season.

This handbook is intended as a source of information for agricultural entomologists, crop protection personnel and scientists. It is not designed as a ‘user manual’, but as a reference text, from which training and advisory literature may be produced. The work cited in this manual comes from many scientific papers, reports, personal communications and experiences, some as yet unpublished. Further information on the subject contained in each section may be found in the key references cited. The comprehensive bibliography, which also includes some useful, more general references, will provide source material for those interested in further research. It is hoped that this compilation will thus serve many purposes.

The handbook is based on the experience and research carried out mainly in eastern Africa by the Regional Armyworm Programme of the Desert Locust Control Organization for Eastern Africa (DLCO-EA); the Natural Resources Institute (NRI) of the University of Greenwich; the International Centre for Insect Physiology and Ecology (ICIPE), Nairobi; the Kenya Agricultural Research Institute (KARI) and its predecessor, the East African Agriculture and Forestry Research Organization (EAAFRO); Pest Control Services, Arusha, Tanzania (PCS); the plant protection services of the then member countries of DLCO-EA (Djibouti, Ethiopia, Kenya, Somalia, Sudan, Tanzania and Uganda), and those of other African countries, notably Burundi, Malawi, South Africa and Zimbabwe, as well as the Yemen in the Arabian Peninsula.

Every effort has been made to produce as complete a bibliography as possible, in line with the purpose of the handbook to provide a comprehensive reference text on the African armyworm.

DEDICATION

This handbook is dedicated to everyone who has been involved in helping to unravel the complex ecology and biology of the African armyworm and develop methodologies for the management of this pest in Africa.

In particular we would like to pay tribute to the enthusiasm, tenacity, foresight and originality of the pioneers in this field, especially Eric S. Brown and Derek J. W. Rose in eastern and southern Africa, and J. C. Faure and C. C. Hattingh in South Africa.

The challenge for all of us was *Spodoptera exempta*.

Charles Dewhurst, Bill Page

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Information on the African armyworm has been collected over many years by field staff of a great many countries and many armyworm related projects.

Although personal communications and unpublished data provided by scientists are not individually acknowledged in the text, the value of such observations is very worthwhile and much appreciated. We hope that this acknowledgement will be accepted as recognition of the important contributions made by all that have shared in the study of the African armyworm.

The important work carried out by moth trap operators, control personnel, staff of the Ministries of Agriculture, Crop Protection Services and the National Armyworm Co-ordinating Offices in Burundi, Djibouti, Eritrea, Ethiopia, Kenya, Malawi, Somalia, South Africa, Rwanda, Sudan, Tanzania, Uganda, Yemen, Zambia and Zimbabwe is especially appreciated and acknowledged. Their data have provided the basis for understanding the patterns of events as they were gradually revealed year by year, and for their subsequent interpretation by specialists world-wide.

The specialists who participated with the DLCO-EA in these armyworm studies are all gratefully acknowledged. They include scientists from a wide range of disciplines who have provided their skills to investigate particular aspects of armyworm biology, ecology and management. Written contributions and personal communications we have received have been incorporated into the text and are gratefully acknowledged.

We wish particularly to acknowledge the long-standing and constant support that the Regional Armyworm Programme received from the following organizations, institutions and governments:

The Director and past Directors-General, the past Director of Scientific Research, the Director of Operations and staff of the Desert Locust Control Organization for Eastern Africa (DLCO-EA);

Past and present colleagues, both scientists and librarians, at the Natural Resources Institute (NRI) and its predecessors of the Department for International Development (DFID) of the UK Government;

Technical Co-operation projects and many short-term visits in Kenya and Tanzania supported by DFID of the UK Government;

The Delegation of the Commission of the European Union in Nairobi, Kenya;

Pest Control Services (PCS), Arusha, Tanzania and the Crop Protection Branch, National Agricultural Research Laboratories (NARL), Nairobi, Kenya for their particular roles in the development of integrated national armyworm control programmes and strategic control procedures;

The Director, National Agricultural Research Centre (NARC) of the Kenya Agricultural Research Institute (KARI) at Muguga, and its predecessor, the East African Agriculture and Forestry Research Organisation (EAAFRO), Nairobi, Kenya;

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The Directors of the Kenya Meteorological Department, Tanzania Meteorological Department, Ethiopia National Meteorological Services Agency and the Department of Meteorological Services, Zimbabwe;

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Major contributors to sections within this handbook were:

Mrs E. M. Ambridge, Mr J. F. Cooper, Mr C. F. Dewhurst, Mr H. M. Dobson, Dr A. G. Gatehouse, Mr D. Grzywacz, Miss M. J. Haggis, Professor D. R. Hall, Dr M. R. K. Lambert, Dr N. R. Maslen, Mr L. J. McVeigh, Mr W. W. Page, Mr D. E. Pedgley, Dr D. R. Reynolds, Dr D. J. W. Rose, Mr P. J. Scott, Dr M. R. Tucker,

The photographs were taken by W. W. Page, the late E. S. Brown, C. F. Dewhurst, J. Riley and the late N. Tabu. Many of the illustrations were redrawn by Sasha Lauer and G. J. du Heaume.

The original draft scripts were collated and typed by W. W. Page and C. F. Dewhurst. The script was read and corrected by Regional Armyworm Programme staff and special thanks are due to P. O. Odiyo and T. Megenasa. Editing of the first edition of the script was undertaken by the compilers and Miss P. J. Wortley, and final editing by Miss M. J. Haggis and C. F. Dewhurst.

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Revision and updating of the second edition was undertaken by W. W. Page and C. F. Dewhurst. We thank Mr J. Stickings (ASSC manager) for his support of this work. The typesetting and production were undertaken at the University of Greenwich.

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CONTENTS

1

SUMMARY

The African armyworm, *Spodoptera exempta* (Lepidoptera: Noctuidae) is a pest of pastures and cereal crops in Africa south of the Sahara, parts of Arabia, Asia, Australia and the Pacific, including Hawaii. This noctuid moth is a migrant and, in plague years, many thousands of square kilometres in eastern, central and southern Africa may be infested with its larvae at densities up to and occasionally exceeding 1000/m^2, often in patchy outbreaks which are seldom contiguous. Due to the armyworm's sudden appearance, crop protection services become overwhelmed with requests for help during the short time available for control operations. As a consequence, the African armyworm has gained notoriety as a pest species second only to locusts. Its occurrence must be notified to the agricultural authorities in those countries where government assistance is provided for its control. Economic losses caused nationally or to individual farmers may be considerable, even though available figures are imprecise. Action thresholds have been tentatively estimated to prevent a 15% crop loss.

The rate of feeding by larvae increases rapidly during the 2–3-week development period, until they suddenly disappear by burrowing into the ground to pupate. Larvae feed on a wide range of grasses, related cereals and sedges. Dicotyledons are rarely eaten. Important cereal crops attacked include maize, wheat, sorghum, millet, teff and rice. Both the variety and the condition of host plants affect their suitability as food for armyworm. Some varieties of maize are more resistant to attack than others.

The larvae of other noctuid species are often mistaken for armyworm, and this handbook includes keys, figures and plates to illustrate and distinguish eggs, larvae, male and female moths. Pupae are very difficult to identify, but when they are found in large numbers, *S. exempta* may be reasonably inferred. Wherever there is doubt, expert advice should be sought to ensure the correct identity of the stage in question.

The distribution of outbreaks varies both temporally and spatially, and follows the onset of the wet seasons when dry grasslands produce new growth and cereal crops are planted. Major outbreaks of armyworm are commonly preceded by extended drought. The African armyworm is well adapted to exploit the food available in the seasonal grasslands in which it lives, and it has sophisticated ways of reacting to the environment through physiological and genetic changes. These changes are expressed through the physiology and behaviour of larvae and moths.

The crowding of larvae results in changes in both their colour and behaviour. Larvae in outbreaks in the dark *gregaria* phase are conspicuous, active and voracious feeders, while the low-density *solitaria* larvae appear superficially to be a different species, especially to the untrained eye. They are cryptically coloured, sluggish, hide at the base of grasses, and are found at low densities. It is in this form that populations survive between outbreak seasons.

In East Africa, areas favourable for larval survival during the dry 'off-season' are most extensive in coastal regions, but armyworm populations can also persist in the highlands, where low temperatures prolong development.

Although colour and other external phase changes have not been detected in moths, physiological and genetic differences have been identified which affect their flight behaviour. The flight duration of female moths, and their fecundity, is greater in populations from gregarious phase larvae than from solitary phase larvae. Once the moths have ceased migrating, they mate, the females attracting males by producing a sex pheromone. Females usually lay eggs in batches of up to 600 in a mass. They lay one egg batch every night until approximately 1000 eggs have been laid.

The number of nights that moths are capable of migratory flight largely corresponds to the female pre-oviposition period, which may last from 2 to 13 or more nights. Migrating moths are carried downwind at heights up to several hundred metres above the ground. They do not remain in cohesive swarms like locusts, but become widely dispersed unless they are reconcentrated by convergent winds, as commonly happens in association with storm outflows and topography. Further outbreaks may then develop in areas where the moths become reconcentrated. These attributes ensure dispersive migration by moths leaving outbreak sites and the rapid colonization of new habitats.

The population dynamics, onset and spread of armyworm outbreaks are strongly influenced by weather conditions. In eastern Africa, the first outbreaks of the season appear to be caused by moths derived mainly from coastal source areas, where out-of-season rainfall enables low density populations to survive the dry ‘off-season’; the moths are carried inland on the dominant easterly winds. These ‘primary’ outbreaks occur in association with the early rainstorms of the ‘short rains’ season in central or northern Tanzania or south-eastern Kenya. They are often found on the east side of high ground inland from the coast, where annual rainfall is erratic and usually low.

Outbreaks usually spread westwards and northwards from central or northern Tanzania towards Burundi, Rwanda and Kenya, and from Tanzania and Kenya to Uganda, southern Sudan, Ethiopia, Eritrea, Somalia and the Yemen. The patterns of spread are determined by the directions of the dominant winds and locations of rainstorms during the times of moth flights. Outbreak areas can be predicted from a knowledge of the wind fields and the distribution of rainstorms and moth sources.

There appears to be a simultaneous southwards spread of outbreaks from southern Tanzania, Malawi and Mozambique into Zambia, Zimbabwe, Botswana, Swaziland and South Africa, again determined by the winds. Little is currently published about these movements, and virtually nothing about the activity of *S. exempta* in western Africa, where serious outbreaks sometimes occur. The three regions of eastern, central-southern and western Africa are suggested as suitable for centralized armyworm management, to correspond with the major zones of armyworm movements.

Strategies for the management of armyworm outbreaks are based on rapid reporting of outbreaks and monitoring subsequent moth populations and their movements. In eastern Africa, armyworm management is co-ordinated by the Desert Locust Control Organization for Eastern Africa (DLCO-EA). Further south the International Red Locust Control Organisation for Central and Southern Africa (IRLCO-CSA) assumes this role.

The numbers of moths caught nightly in networks of light and pheromone traps in collaborating countries are recorded on standard record sheets and pre-paid printed cards. Also recorded are the dates and places of outbreaks, daily wind direction and rainfall. National forecasts of expected locations of outbreaks are issued weekly. DLCO-EA co-ordinates the exchange of information between all its member countries in eastern Africa, thus providing an overview of reported and expected population developments and movements of moths within and between countries. Regional co-operation is essential for effective monitoring, survey, forecasting and control of migrant pests.

Crop protection may be direct, controlling armyworm that are damaging crops, or indirect, by limiting the spread of outbreaks, i.e. strategic control. Strategic control is designed to destroy as many of the primary outbreaks as possible, and any other outbreaks defined for control decision purposes as ‘critical’. Critical outbreaks are those which, if left uncontrolled, would be major sources of moths likely to invade new areas downwind (especially land under agricultural production), resulting in the spread of further outbreaks throughout the region. Primary and critical outbreaks may be recognized, with experience, by the time and location of their occurrence in relation to weather patterns.

To minimize the downwind spread of emergent moths, the largest, densest and oldest outbreaks must be eliminated first, whether they are on crops or grassland. Measures for monitoring, surveying, forecasting and controlling should be intensified at the times and in the areas when and where the first outbreaks typically occur. Once the wet season has arrived and outbreaks have started, moths emerge comparatively synchronously from each outbreak site and outbreaks then become the major sources of moths which spread downwind to cause subsequent outbreaks. Evidence for this movement has been obtained by analysis of historical records as well as intensive field studies and provides the basis for forecasting outbreaks.

Forecasting accuracy has been greatly improved by availability of daily wind streamline charts and nightly satellite images of storm-cloud cover for the region. Rapid communication and knowledge of areas of responsibility are essential to ensure the early control of outbreaks. Farmers and agricultural field staff must be warned quickly through radio broadcasts, the national press and any other means of the current situation and the probability and expected locations of imminent armyworm attacks.

Insecticides recommended for the control of armyworms have been chosen for their efficacy, cost-effectiveness, safety to operators and the environment. For control on cereals or pastures, an ultra-low-volume (ULV) formulation of 2.5% cypermethrin applied by drift spraying at the rate of 0.3–0.4 l/ha can be used under some circumstances, although further trials are needed as application techniques continue to improve. Drift spraying using hand-held, vehicle- or aircraft-mounted applicators provides the speed and versatility needed for the treatment of small or large outbreaks on crops and pastures. The development of biological pesticides is ongoing and field testing of conventional and novel techniques and insecticides will be needed as new products become available.

Regular training programmes and exchange of information at workshops are essential for all aspects of armyworm management. Training videos, pamphlets, slides and an illustrated booklet are available at DLCO-EA and the Natural Resources Institute (NRI), from where advice may be provided to national units for the production of training materials applicable to local conditions.

The names and addresses of relevant organizations, sources of equipment and a comprehensive list of scientific papers and reports are listed at the end of this handbook. Key references for each topic are listed at the end of each section in the text.

2

STATUS

2.1 Scientific and common names

The taxonomic position of the African armyworm is as follows:

Class:	Insecta
Order:	Lepidoptera
Family:	Noctuidae
Genus:	*Spodoptera* (Guenée)
Species:	*exempta* (Walker)

This species was originally described as *Agrotis exempta* (Walker 1856). It was known as *Laphygma exempta* (Walker) Hampson 1909 until 1958, when it was renamed by Zimmerman and is now known as *Spodoptera exempta* (Walker) Zimmerman 1958.

A male and female moth are shown enlarged in Plate 1.

Common names

(i)	English:	
	Africa:	African armyworm, mystery worm, hail worm, rain worm
	Hawaii:	nutgrass armyworm,
	Australia:	day-feeding armyworm, variegated armyworm, leaf-eating grassworm
(ii)	French:	chenille légionnaire, chenille processionaire
(iii)	German:	heerwurm
(iv)	Dutch:	legerworm

(v) Other local names:

		Language	Local names
North-eastern Africa			
	Eritrea	Tigrinia	barnosay
	Ethiopia	Amharic	temch
		Oromo	geiry
	Somalia	Somali	diirta afrikaanka
	Sudan	Arabic	el-Dudah, el-Zahfa, el-Afrigia
	Yemen		gidami
Eastern Africa			
	Kenya	Swahili	viwavi jeshi
		Maa	ng'urrto
		Kikamba	keenyu
		Kikuyu	ngonga
		Luo	kungu
	Tanzania	Swahili	viwavi jeshi
		Maa	ng'urrto
		Local tribal languages:	karikari, mbirizi, ngwegu, n'gungu, nseto, nyonge, vigoda
	Uganda	Luo	omor, n'kungula
Central and southern Africa			
	Malawi	Chichewa	nchembere, zandonda (literally 'old ladies follow each other')
		Yoa	chipakusu, kapuchi
	Mozambique		nyanja
	South Africa	Afrikaans	kommandowurm
	Zimbabwe	Shona	mhundururu
		Sindebele	imhogoyi

(The authors would be pleased to receive any additional local names for armyworm.)

Spodoptera exempta moths may be distinguished from other *Spodoptera* species occurring in Africa by using the key given in Appendix 1 and Figures 1a (antennae), 1b (male genitalia), 1c (bursa copulatrix of the female), 1d (spermatophores), and Plate 2 (males and females of the other *Spodoptera* species). See also Section 3.1.1 below.

An important aid to identification of the females, the bursa copulatrix, can be easily exposed by pulling apart the last few segments of the abdomen with a pin. The characteristic features of the bursa copulatrix, and of the spermatophore (when removed from the bursa of a female which has mated), can be seen using a dissecting microscope and are shown in Figures 1c and 1d.

The black hair-scales at the tip of the abdomen of the female moth, and the grey, racket-shaped scales on the outer part of the genital valves of the male moth, are useful external characters which will aid the identification of specimens damaged in light trap catches. Both features are characteristic of *S. exempta*.

Key reference: Brown and Dewhurst (1975)

(a) **Antennae**

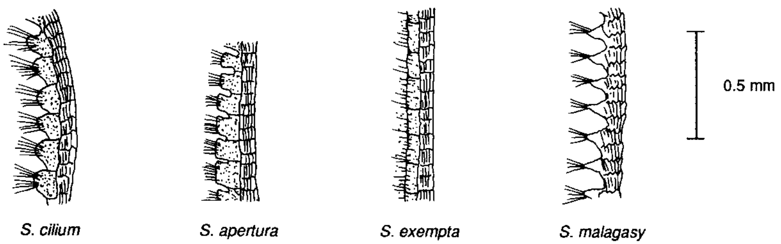
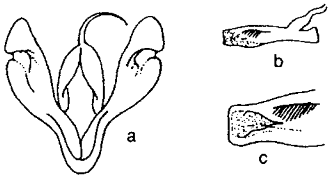
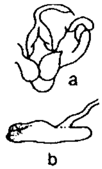


Figure 1. Characters for the identification of moths of *Spodoptera* species (after Brown and Dewhurst, 1975).

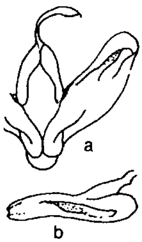
(b) **Male genitalia**



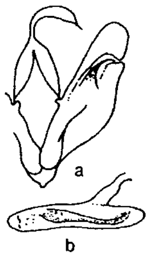
S. exempta, (a) aedeagophore with valves and uncus, (b) aedeagus and (c) cornuti further enlarged



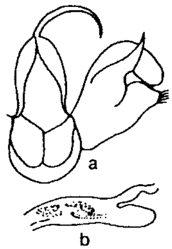
S. cilium, (a) aedeagophore with valves and uncus and (b) aedeagus



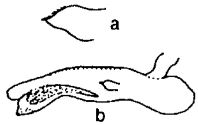
S. triturata, (a) aedeagophore with valves and uncus and (b) aedeagus



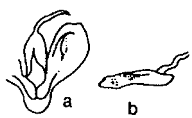
S. mauritia, (a) aedeagophore with valves and uncus and (b) aedeagus



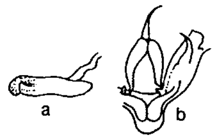
S. littoralis, (a) aedeagophore with valves and uncus and (b) aedeagus



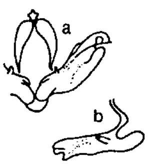
S. litura, (a) cornuti further enlarged and (b) aedeagus



S. exigua, (a) aedeagophore with valves and uncus and (b) aedeagus

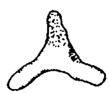


S. apertura, (a) aedeagus and (b) aedeagophore with valves and uncus



S. malagasy, (a) aedeagophore with valves and uncus and (b) aedeagus

juxta of *S. littoralis*

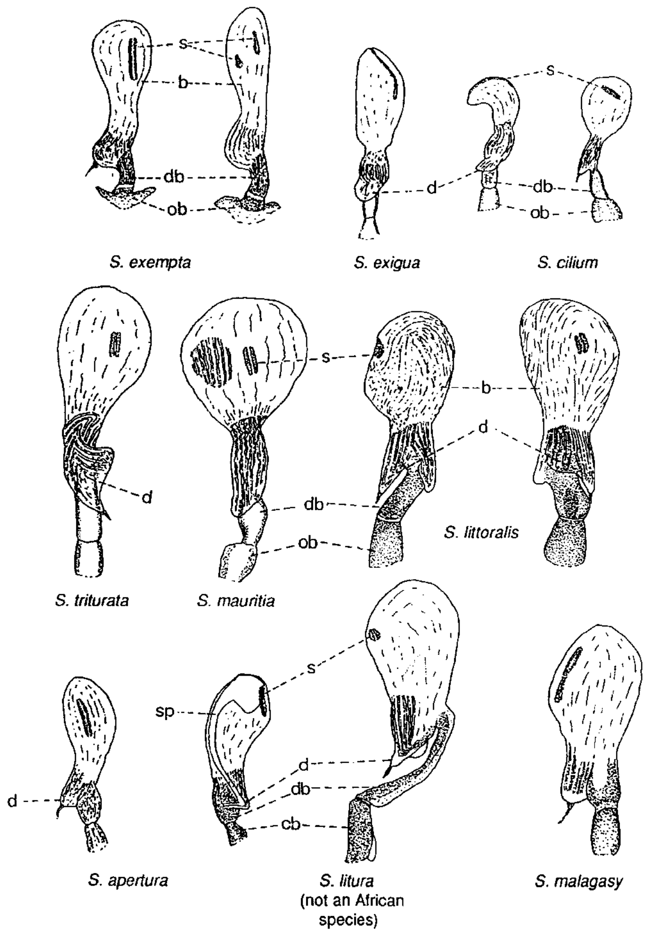


juxta of *S. litura*

(juxta greatly enlarged)

Figure 1 *cont.* Characters for the identification of moths of *Spodoptera* species (after Brown and Dewhurst, 1975).

(c) **Bursa copulatrix of the female**



b *bursa copulatrix*
d *diverticulum*
db *ductus bursae*
ob *ostium bursae*
s *signum*
sp *spermatophore*

Figure 1 *cont.* Characters for the identification of moths of *Spodoptera* species (after Brown and Dewhurst, 1975).

(d) **Spermatophores**

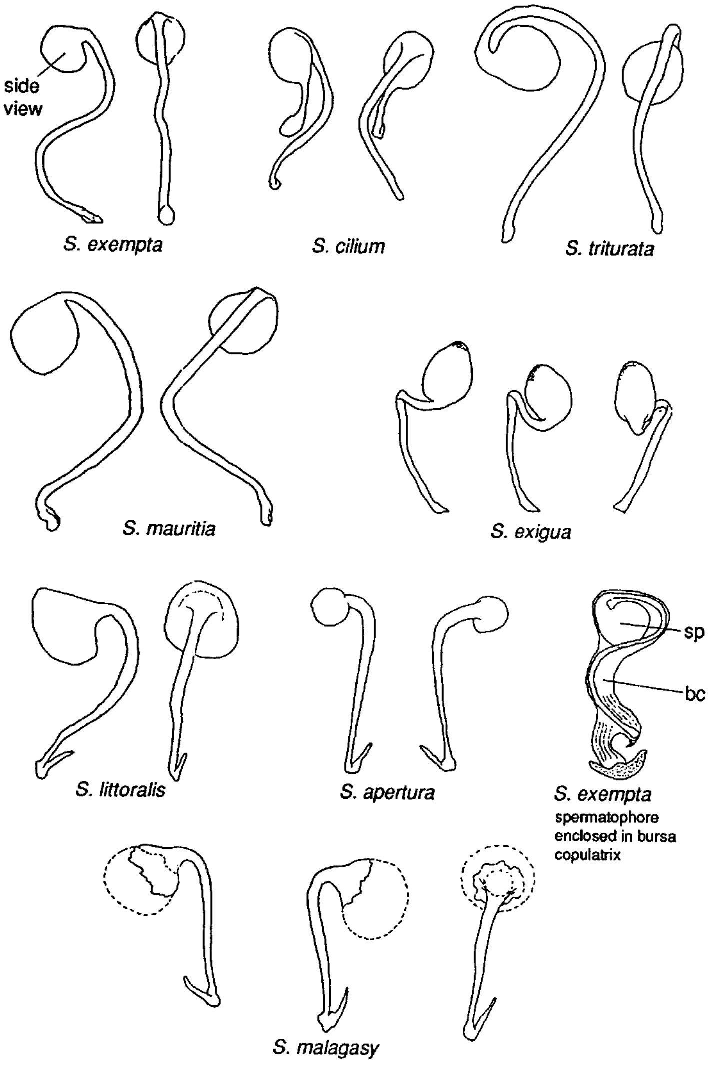


Figure 1 *cont.* Characters for the identification of moths of *Spodoptera* species (after Brown and Dewhurst, 1975).

2.2 Related *Spodoptera* species

2.2.1 *Spodoptera* species found in Africa (Plate 2)

Species	Common names	Remarks
S. exigua (Hübner)	lesser armyworm, pigweed caterpillar, beet armyworm, small mottled willow	causes outbreaks, is polyphagous and may be confused with the African armyworm as the larvae can also be dark in colour
S. littoralis (Boisduval) (previously known as *Prodenia litura* F.)	cotton leafworm (Egypt), tomato caterpillar, maize worm (Zimbabwe)	a major pest of cotton, greenhouse vegetables and other crops (NB. The closely related species *Spodoptera litura* does not occur in Africa.)
S. mauritia (Boisduval)	lawn armyworm, rice swarming armyworm, Far Eastern armyworm, paddy armyworm	uncommon, and not a pest in Africa, recorded from coastal areas of East Africa, it is a serious pest of rice in the Far East
S. cilium Guenée (previously known as *S. capicola*)	lawn caterpillar, dark mottled willow	an important pest of lawns in southern Africa and the Near East
S. triturata (Walker)	larger lawn caterpillar*	larvae are found at the base of *Cynodon* and other grasses, and may be confused with the solitary phase *S. exempta*, of no recorded economic importance
S. apertura (Walker)	dark armyworm*	uncommon and of no economic importance

* New names, assigned in this text.

S. malagasy Viette	Malagasy armyworm*	only recorded from Madagascar, of no recorded economic importance

2.2.2 Important *Spodoptera* species not found in Africa

Species	Common names	Remarks
S. litura (Fabricius)	Indian cotton leafworm	a serious pest of cotton and other crops in India, the Far East, Australia and the Pacific region
S. frugiperda (J.E. Smith)	fall armyworm	a pest similar to African armyworm, found in North America and the Caribbean

Key reference: Brown and Dewhurst (1975)

(Note should be made of a forthcoming taxonomic review of the genus *Spodoptera* by M. Pogue.)

2.3 Geographical distribution

The world-wide distribution of *S. exempta* is illustrated in Figure 2. Outside Africa it has been recorded from the south-western Arabian Peninsula, Australia, and various countries of South East Asia and the Pacific region.

The African armyworm is widespread on the continent of Africa south of the Sahara. It is most prevalent on the eastern side of the continent (Figure 3), with outbreaks having been recorded most frequently in Ethiopia, Kenya, Tanzania, South Africa and Zimbabwe. This pattern of occurrence is associated with the inland topography. Outbreaks occur much less frequently at altitudes higher than 2200 m above sea level.

The numbers of years in which outbreaks of *S. exempta* have been recorded in each country in Africa during each month from 1930 to

* New names, assigned in this text.

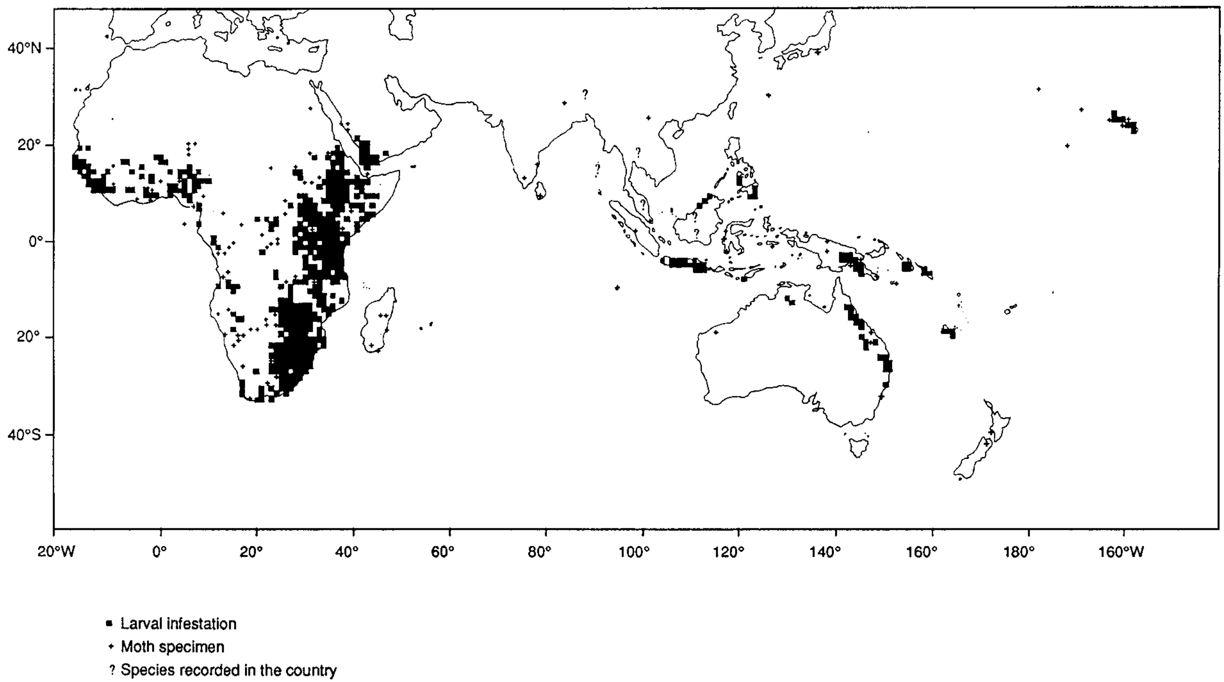


Figure 2. World-wide distribution of *Spodoptera exempta* (Walker) (after Haggis, 1984).

Figure 3. Number of seasons in which infestations of *Spodoptera exempta* were recorded in each territory in Africa and south-western Arabia, 1930–88 (adapted and revised from Haggis, 1984). (A season is defined as the calendar year for countries north of the equator and as being from October to the following September in countries from Kenya southwards.)

1988 and, where possible to 1999, are shown in Table 1. Outbreaks have been recorded from some part of Africa almost every year since 1919. The earliest known records are from Ethiopia in 1520, Hawaii in 1873, South Africa in 1878, and Sierra Leone in 1888. Prior to 1900, adult moths had been collected from Australia, Ethiopia, Gabon, Hawaii, Kenya, Madagascar, Mozambique, Papua New Guinea, Somalia, Sudan, Uganda and Yemen. Although this distribution reflects the vagaries of collecting and recording during these early years, the records do show that armyworm has occurred in Africa for many years and that it is certainly indigenous. Recent evidence suggests that armyworm is occurring more regularly and from countries in central and west Africa that have had few reported outbreaks in the past.

There is a marked seasonal distribution in the occurrence of outbreaks, which is closely associated with seasonal rainfall. The general sequence of occurrence of outbreaks is in a northward direction from central Tanzania and follows the movement of the Inter-Tropical Convergence Zone (ITCZ), which is the meeting of the north-easterly and south-easterly tropical wind systems (see Section 4.1.2). In other countries the seasonal distribution of outbreaks is also related to the rains, with successive generations spreading southwards from southern Tanzania, Malawi and northern Mozambique. Work at IRLCO-CSA continues to update knowledge of the pest in southern Africa.

Maps illustrating the distribution of outbreaks in Africa month by month are given in Figure 4. As an example of the chances of outbreaks occurring in a country on a year to year basis, the annual probability of outbreaks occurring in South Africa where armyworm threatens 122.9 million people in the SADC (Southern African Development Community) region (67% of the total population whose livelihoods depend on agriculture) is illustrated in Figure 5.

Key references: Annecke and Moran (1982)
Botha (2000)
Commonwealth Institute of Entomology (1972)
Haggis (1984)

Table 1. Number of years in which outbreaks of *Spodoptera exempta* were recorded each month in each territory in Africa and south-western Arabia from 1930 to 1988 and, where possible, to 1999

	J	F	M	A	M	J	J	A	S	O	N	D
WEST AFRICA												
Mauritania	-	-	-	-	-	1	1	-	1	-	-	-
Senegal	-	-	-	-	-	2	4	2	2	-	-	-
The Gambia	-	-	-	-	-	3	4	4	1	-	-	-
Guinea-Bissau	-	-	-	-	-	1	3	-	-	-	-	-
Mali	-	-	-	-	-	1	2	-	-	1	1	-
Burkina Faso	-	-	-	-	-	2	3	-	-	-	-	-
Niger	-	-	-	-	-	1	1	1	-	-	-	-
Chad	-	-	-	-	-	-	-	-	-	-	-	-
Guinea	-	-	-	-	-	2	4	5	1	2	2	-
Sierra Leone	-	-	1	1	5	5	5	2	2	-	-	-
Liberia	-	-	-	-	-	-	-	1	1	-	-	-
Côte d'Ivoire	-	-	-	-	-	-	-	1	1	-	-	-
Ghana	-	-	1	1	1	6	7	5	3	3	-	-
Togo	-	-	-	-	-	-	-	-	-	-	-	-
Benin	-	-	-	-	-	-	-	-	-	-	-	-
Nigeria	-	-	-	2	7	1	0	5	2	3	1	-
Cameroon	-	-	-	-	2	-	-	-	-	-	-	-
Central African Republic	-	-	-	-	-	-	-	-	-	-	-	-
ARABIA, NORTH-EAST AFRICA												
Saudi Arabia	-	-	-	-	-	-	-	1	-	-	-	-
Yemen	-	-	-	-	8	21	20	11	-	-	-	-
Djibouti	-	-	-	-	-	-	-	-	-	-	-	-
Somalia	-	-	1	6	8		2	1	-	1	5	1
Ethiopia*	1	-	4	16	23	26	21	16	13	6	2	-
Sudan	-	1	2	3	3	3	3	-	3	3	-	-
EQUATORIAL AFRICA												
Equatorial Guinea	-	-	-	-	-	-	-	-	-	-	-	-
Gabon	-	-	-	1	1	-	-	-	-	-	-	-
Congo	-	1	-	-	-	-	-	-	-	2	1	-
Congo (Democratic Republic)	-	1	-	1	2	1	-	-	-	-	-	-
Rwanda*	-	1	2	1	1	1	1	-	-	-	-	-
Burundi*	1	2	3	2	3	1	1	-	-	-	-	-
Uganda*	3	11	16	12	11	4	2	-	-	-	-	-
Kenya*	18	24	29	30	27	24	8	1	-	5	5	12
Tanzania*	39	33	32	28	18	2	-	-	1	2	12	27
CENTRAL AFRICA												
Mozambique*	15	14	8	3	3	-	-	-	-	-	1	3
Malawi*	21	10	8	10	9	1	-	-	1	6	10	17
Zimbabwe*	31	17	15	10	2	1	-	-	-	3	9	23
Zambia*	10	3	7	3	1	-	-	-	-	2	4	10
Angola	-	1	2	1	2	-	-	-	-	-	-	-
SOUTHERN AFRICA												
Namibia	1	2	1	1	-	-	-	-	-	-	-	-
Botswana*	1	5	4	3	1	-	-	-	-	-	-	-
Swaziland	1	4	3	-	-	-	-	-	-	-	-	2
Lesotho	1	1	-	-	-	-	-	-	-	-	-	-
South Africa	20	35	37	11	2	1	-	-	-	-	-	4

* Data up to 1999. Revised from Haggis (1984).

(a) **January**

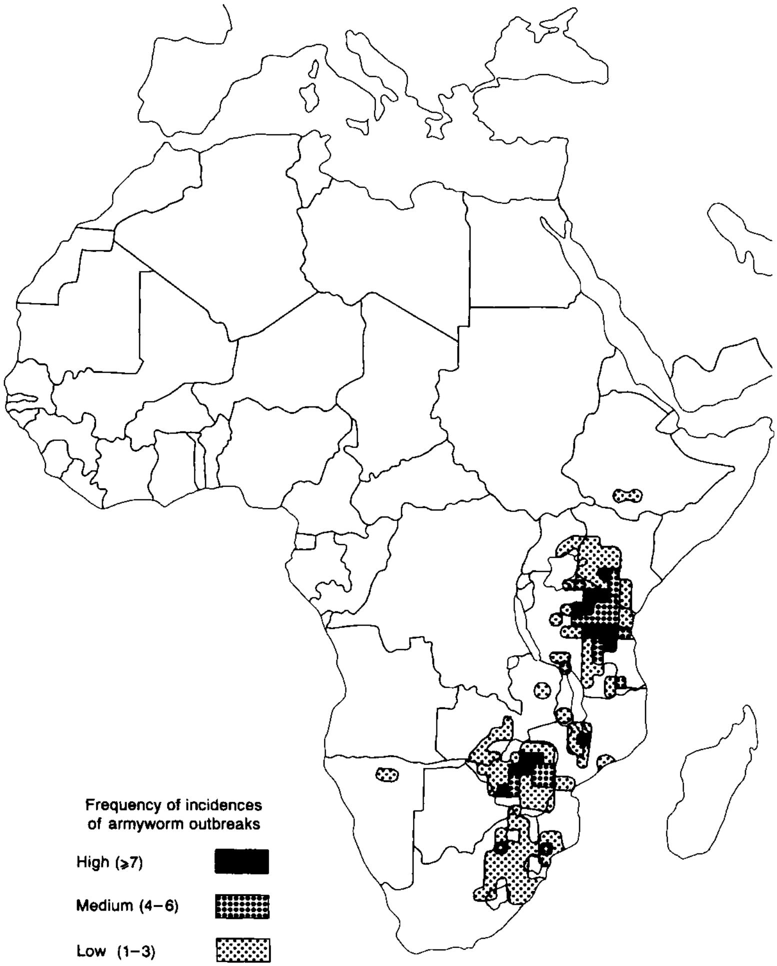


Figure 4. Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(b) **February**

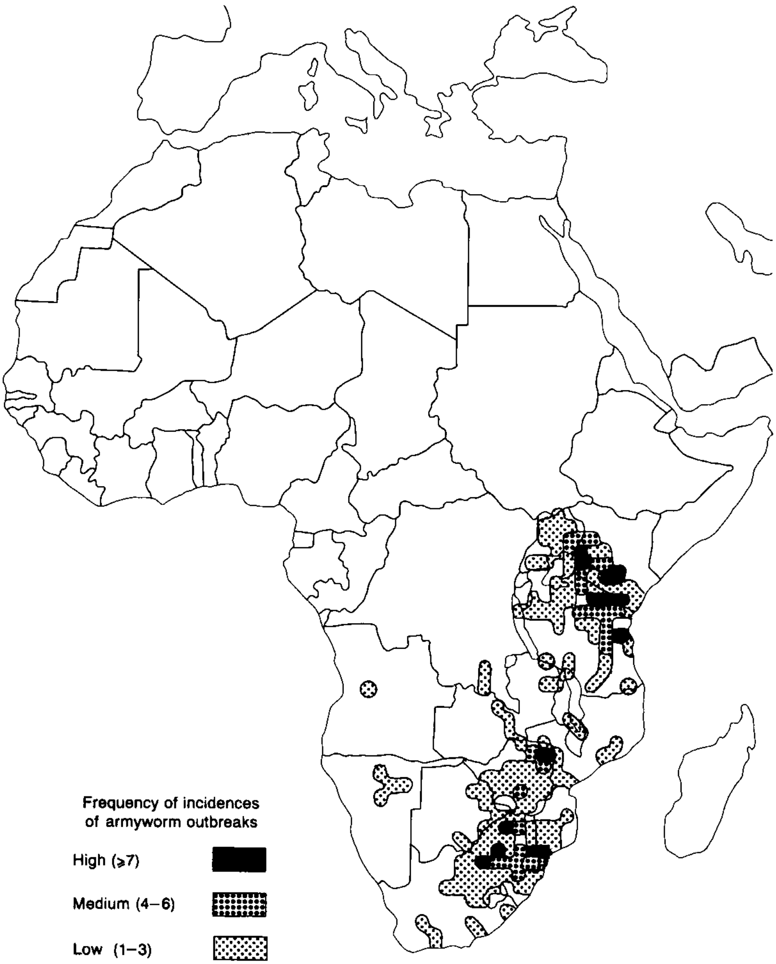


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(c) **March**

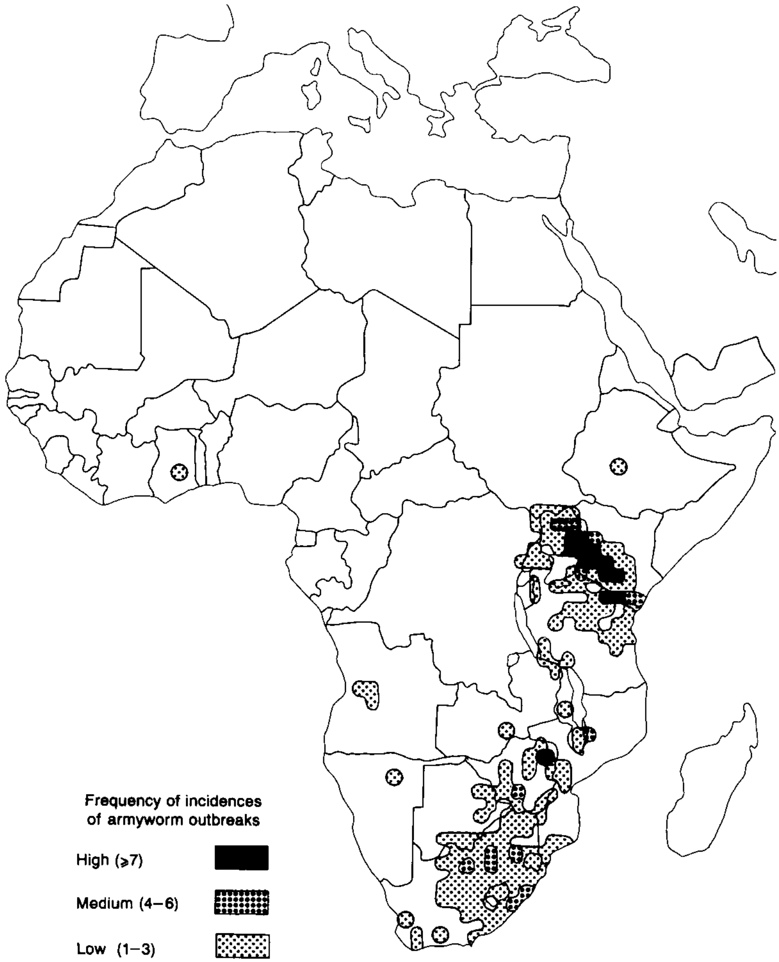


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(d) **April**

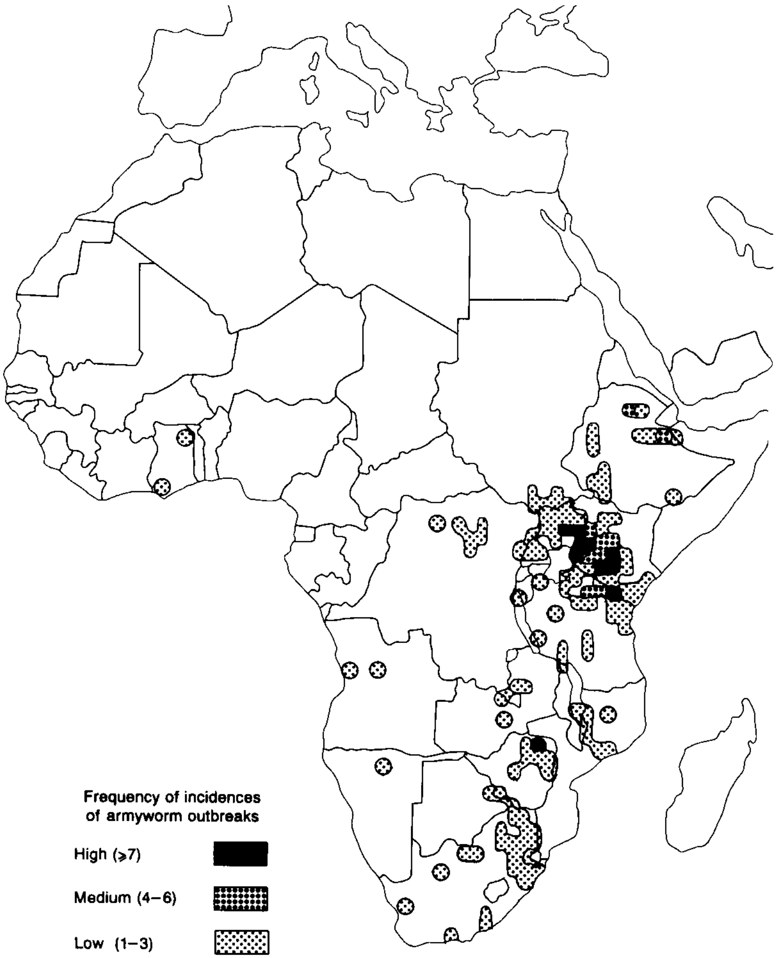


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(e) **May**

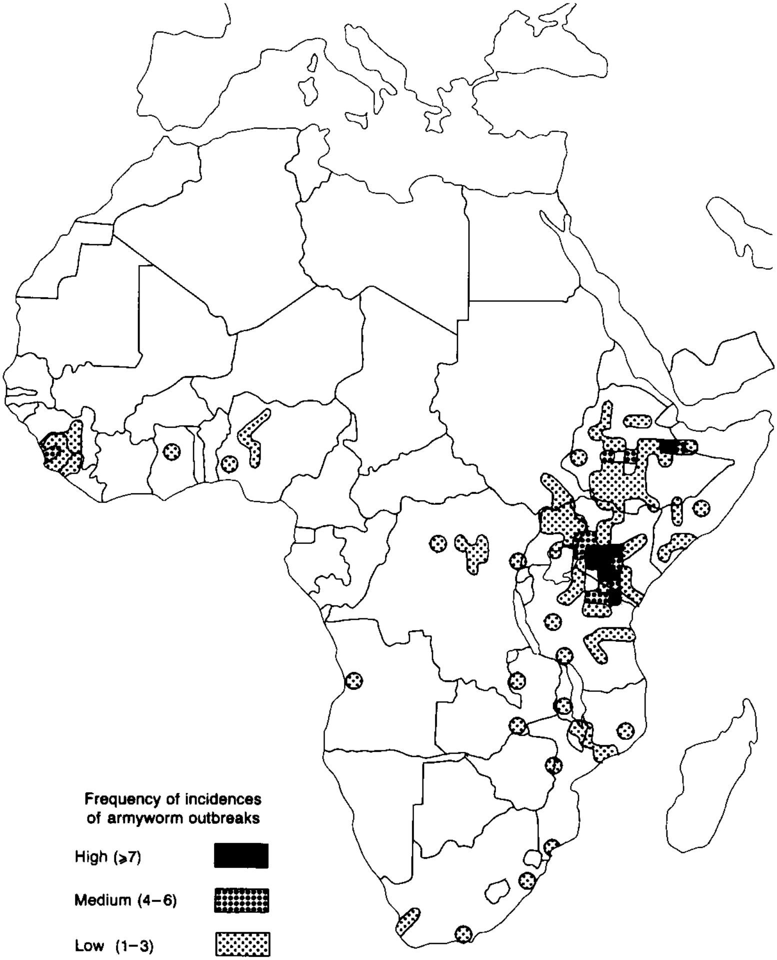


Figure 4 ***cont.*** Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(f) **June**

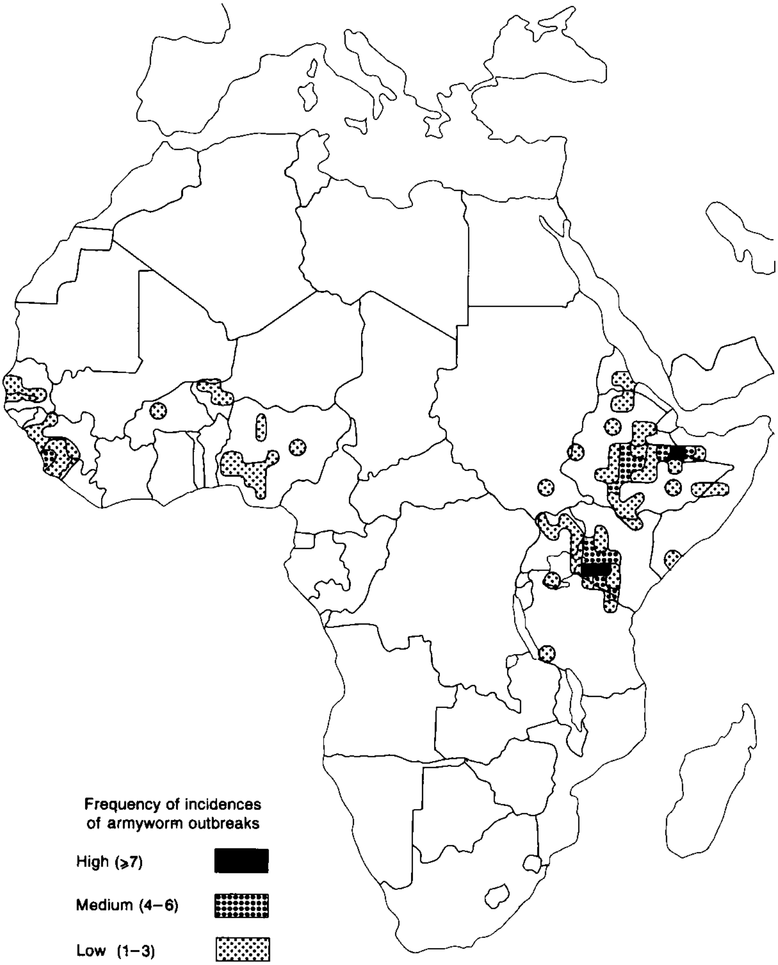


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(g) **July**

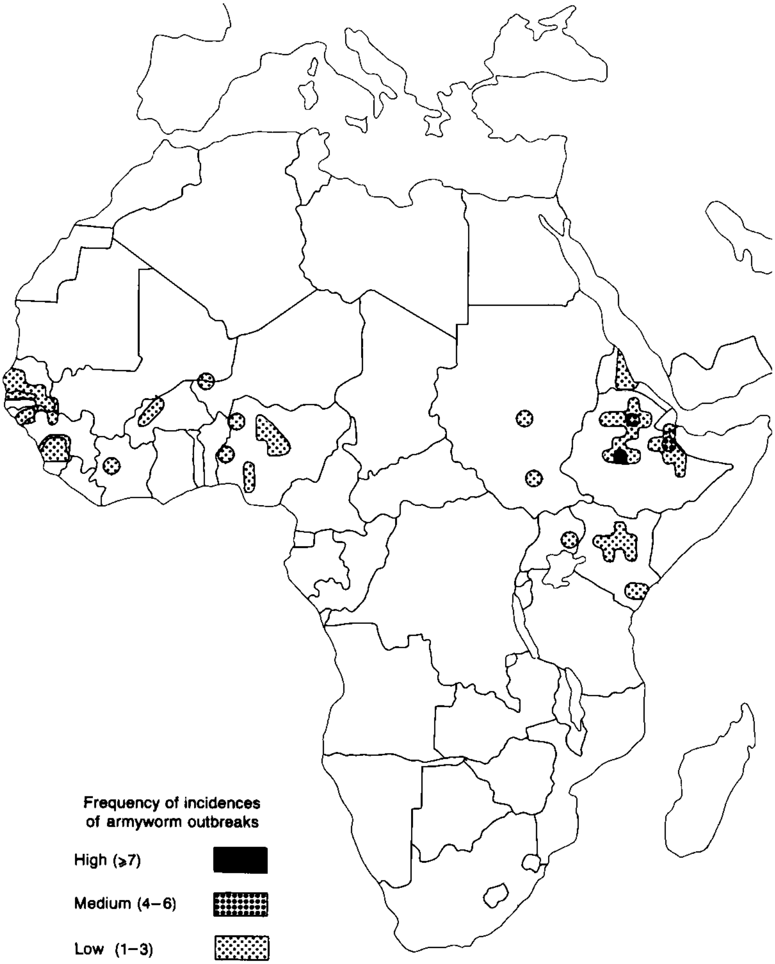


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(h) **August**

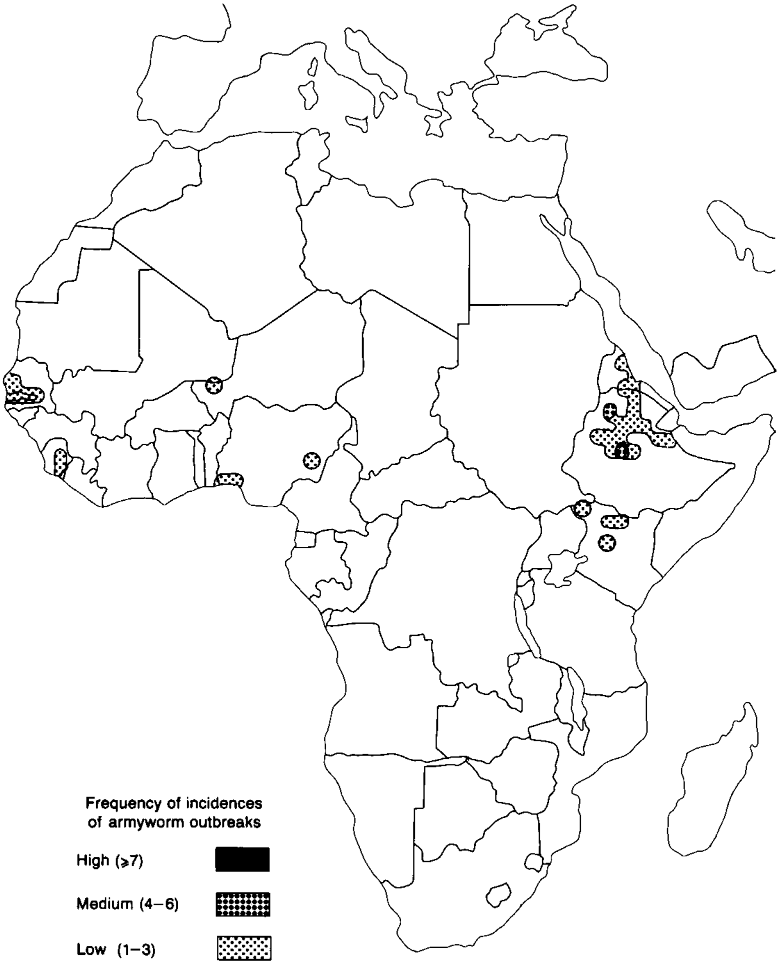


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(i) **September**

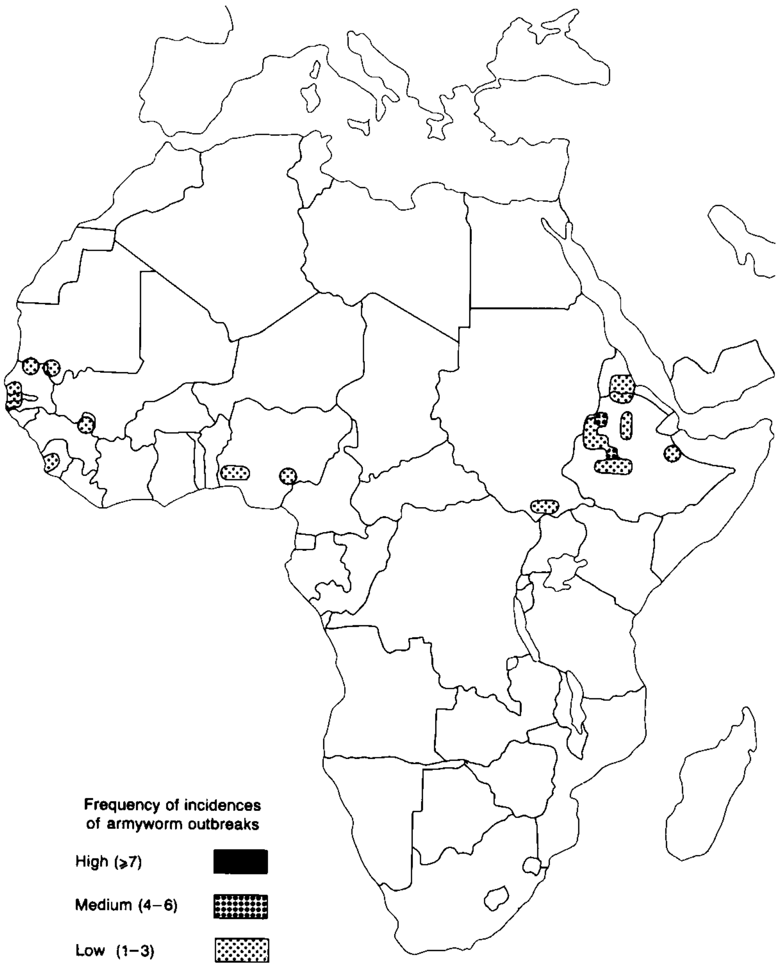


Figure 4 ***cont.*** Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(j) **October**

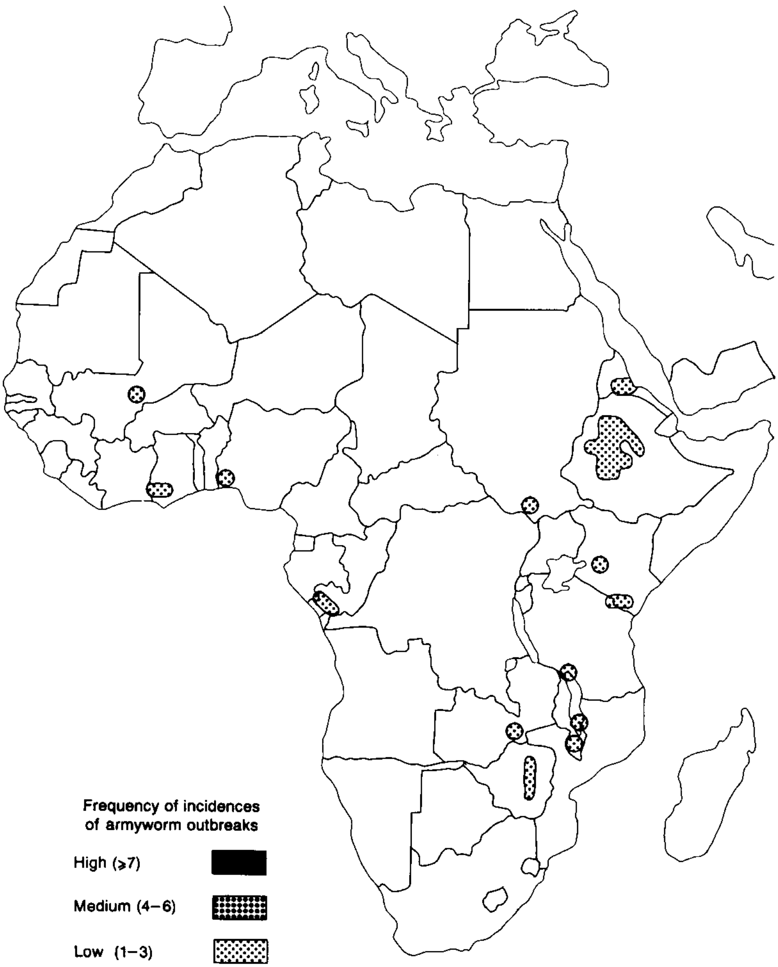


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(k) **November**

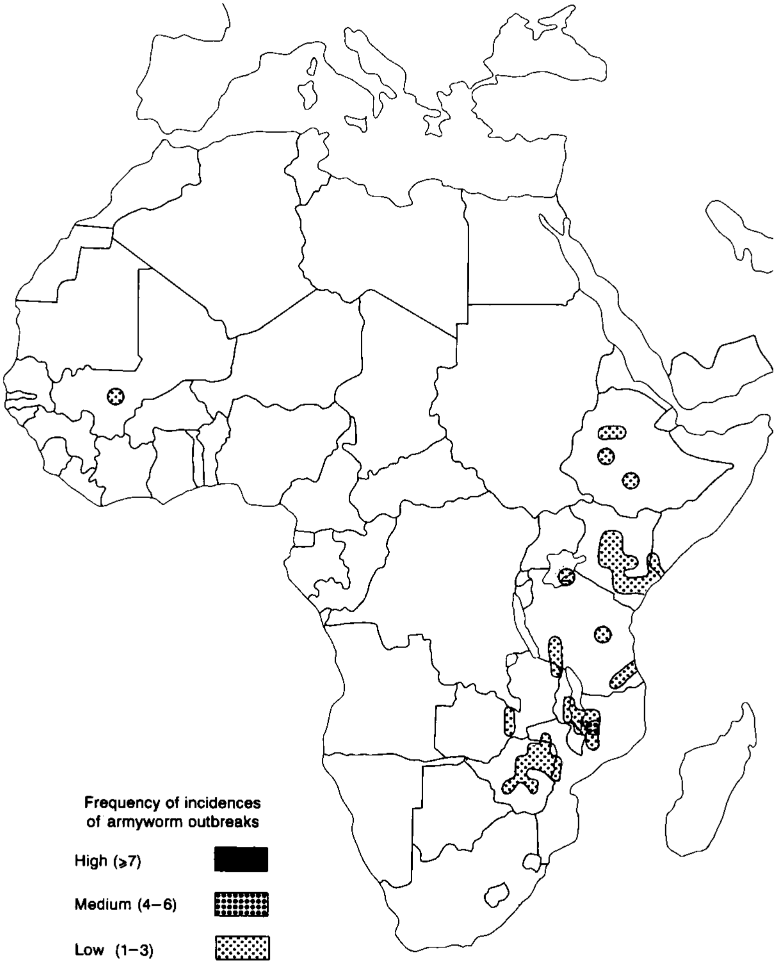


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

(l) **December**

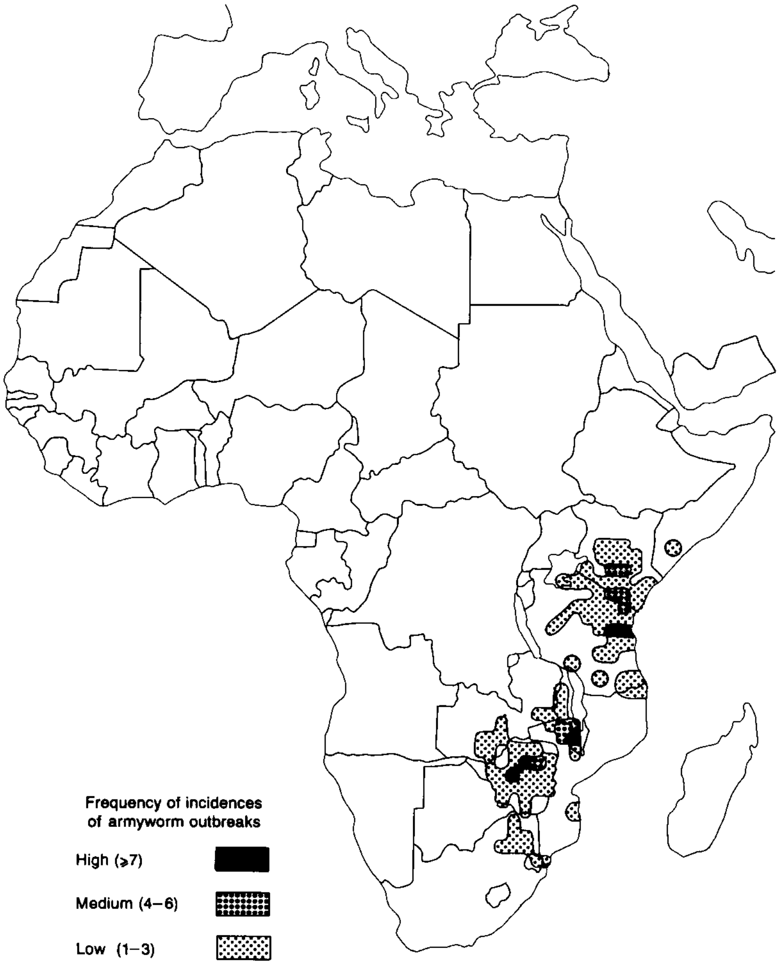


Figure 4 *cont.* Frequency of incidence of armyworm outbreaks in Africa, in each 1° square of latitude and longitude, 1940–82 (after Haggis, 1984).

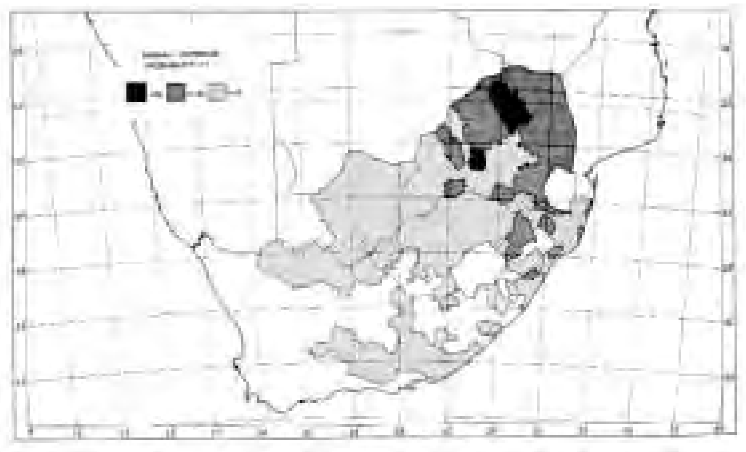


Figure 5. The annual probability of outbreaks occurring in South Africa (after Annecke and Moran, 1982).

2.4 Outbreaks

Definition. An armyworm outbreak is defined as an occurrence of larvae in very large numbers with the majority being in the black (gregarious) form or phase (see Section 3.1.3). 'Outbreak' is a term which aptly describes their sudden appearance, which results from an explosive increase in numbers of *S. exempta* larvae over a relatively short period of time (see Sections 3.1.2 and 4.2.1).

The density of larvae varies considerably both within and between outbreaks. Armyworm may appear as plagues, with larvae in densities from less than one up to, typically, tens to hundreds per square metre and occasionally exceeding 1000/m^2. Many infestations may occur simultaneously but they are rarely continuous, which makes it difficult to delimit their boundaries and estimate the total size of the infested area (see Section 5.2.5). Outbreaks can occur over large areas within which concentrations with widely varying densities are found in a clumped distribution among more scattered larvae.

It is the sudden and widespread appearance of large numbers of larvae, resulting in extensive damage to host plants in just a few days, which makes the African armyworm such a notorious pest. Frequently, farmers and pest control services are overwhelmed by reports of infested areas requiring control. The time available to achieve control is limited because the armyworm is in the larval stage for only 2 to a maximum of 3 weeks. Usually, they are not seen until they are conspicuous and well grown (i.e. they have reached about the fourth (IV) or fifth (V) instar – see Section 3.1.3), leaving little time (typically 1 week) available for control operations to be planned and executed.

The severity and location of outbreaks varies considerably from year to year, and is influenced by the weather. Particularly serious outbreaks developed in East Africa in 1961/62, 1970/71 and 1984/85 after drought, with other severe armyworm seasons occurring in 1964/65, 1965/66, 1973/74, 1976/77, 1978/79, 1979/80, 1980/81, 1981/82, 1983/84, 1985/86, 1987/88, 1991/92, 1992/93, 1993/94 and 1998/99. Thus there are no clearly identifiable cycles of infestation, as are exhibited by some pest species. With the widespread outbreaks affecting eastern and central Africa during the 1998/99 armyworm season, evidence is growing to suggest that outbreaks are being reported more regularly and in some countries are now an almost annual occurrence.

Key references: Brown *et al*. (1969)
Haggis (1984, 1986a)
Odiyo (1981, 1984)

2.5 Economic losses

Losses in agricultural production due to the African armyworm affect the economies of both farmers and nations. Armyworm attack and thus cause the loss of cereal crops and sugarcane, pastures and rangeland. They are, therefore, also of importance to livestock production, especially for pasturalists.

2.5.1 Damage to crops

Damage to cereal crops can be caused in three different ways:

(i) direct attack on young plants less than 30 cm high by young larvae which either hatch from eggs laid on the crop, or are carried by the wind on to the crops, drifting on silken threads produced by the larvae soon after hatching;

(ii) by older larvae crawling into the crop from nearby wild grasses or grass weeds growing among the crops and on which they had been feeding;

(iii) by large-scale invasion by nearly full-grown larvae from adjacent infested grasslands which they had completely eaten; when this occurs even older crops can be totally destroyed.

Resource-poor smallholder farmers are particularly vulnerable to severe economic loss because of their insufficient resources to cope with armyworm invasions. Often they do not have the equipment, pesticide or funds to enable them to control infestations in time. Smallholder farmers seldom have spare seed for replanting if their crops are destroyed. As neighbouring farms, often widely separated, may be infested simultaneously, there is little opportunity for sharing transport, labour or equipment.

In some countries, armyworm control is the responsibility of the government but, due to economic and logistical constraints, agricultural (crop protection) extension services may be able to provide only limited assistance and farmers, particularly the larger ones, are expected to purchase and apply their own insecticide to control the armyworm larvae.

If drought conditions follow an outbreak, plants are unlikely to recover from defoliation and even replanting may not be successful. Farmers may not be able to replant for various reasons such as availability of seed, cost of buying replacement seed, or insufficient rain.

In **maize**, yield reduction caused by defoliation is almost directly proportional to the percentage of leaf area available to the larvae at the time of attack. Reported yield losses range from 9% at early whorl (4–6 leaves), to 100% at pre-tassel stage (12 leaves). Under some circumstances, however, young maize will recover from total defoliation. The ability of young maize to recover from armyworm damage depends on the position of the growing point (apical meristem) at the time of attack and on the amount of root development when the larvae ceased feeding. Damage is serious if the apical meristem is damaged but, as it remains at the base of the plant until near 'tasselling'*, it is likely to be below ground during the armyworm outbreak period and thus escape damage.

Farmers will sometimes replant maize when armyworm larvae have eaten the first sowing of plants down to ground level, even though the optimum planting date will have passed. Yield losses of 6% have been estimated for each day's delay after the optimum planting date in high rainfall areas in Kenya. Areas with less rainfall may suffer higher losses. Yield losses in maize of up to 92% have been measured in trials in Malawi and Kenya (see Table 2).

In **sorghum**, **millet**, **rice** and **teff**, armyworm damage may stimulate 'tillering'† which could, in favourable conditions, increase yield. If subsequent rainfall is favourable for crop growth and development, yield losses may be relatively small provided that the attack occurs before the critical grain initiation stage has been reached.

Crops attacked at heights of 0–30 cm were: maize, 65.9%; sorghum, 49.5%; while in finger millet, 99% (Iles *et al*., unpublished NRI report).

The **action threshold for crops** is the density of armyworm larvae at which control measures should be undertaken to prevent economic damage. Tentative nominal action thresholds have been determined for maize. It is estimated that to avoid yield losses of more than 15%, action thresholds for early whorl (4–6 leaf) maize should be taken as 200 second (II), 80 third (III) or 20 fourth (IV) instar larvae per 100 plants (see Section 3.1.3 for description of larval stages).

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* Tasselling – the production of male inflorescences.

† Tillering – the production of further shoots from the base of the main stem.

Table 2. Examples of recorded crop losses due to armyworm in different maize plots in Kenya; yields from controlled and uncontrolled village plots A–H at Enkoiperiae (maize at 3–4 leaf stage) and plots I–K at Keturo (maize at 4–6 leaf stage) in Kenya

Plots	**Mean larvae per plant in controlled sub-plots**	**Mean larvae per plant in uncontrolled sub-plots**	**Mean yield (kg/ha) controlled sub-plots**	**Mean yield (kg/ha) uncontrolled sub-plots**	**Difference**	**Loss (%)**
A	6.40	4.5	2222	1562	660	29.70
B	19.80	22.4	1947	1115	832	42.73
C	7.55	3.4	2127	1998	129	6.06
D	3.60	3.4	2636	1780	856	32.47
E	4.15	3.9	1945	1721	224	11.50
F	5.10	4.9	3380	2517	863	25.53
G	5.65	4.5	2376	2122	254	10.69
H	0.55	2.4	3682	2541	1141	30.99
I			3802	315	3487	91.72
J			4099	934	3165	77.21
K			3655	3246	409	11.19

Enkoiperiae: mean loss = 23.7; Keturo: mean loss = 60.0
Adapted from Gathuru *et al*. (1991).

The economic injury level is the fourth instar, as this is the larval stage at which serious damage begins to occur.

More information on the micro- and macro-economic effects of armyworm infestations of crops, pastures and rangeland is needed.

2.5.2 Damage affecting livestock production

Damage to pasture and rangeland can be both severe and extensive. The resultant change in sward composition can persist for many years if armyworm damage to grasses gives dicotyledons (broad-leafed plants) a growth advantage, as is likely in lower rainfall areas. This effect is reinforced by drought and overgrazing by cattle, sheep and goats. On the other hand, in areas of good rainfall (both in amount and distribution), the direct effects of armyworm on grazing land are not as long-lived, and regeneration depends on the grazing pressure and rainfall. Good rainfall after armyworm attack is an important factor in pasture recovery. In Kenya, vegetation changes have remained for many years before good grass cover returned after management of the broad-leafed weeds which grew up in place of the eaten grasses.

Survey data from Tanzania indicate that the effects of armyworm damage may last for more than 8 weeks but, in the majority of areas of good rainfall, they seldom last for more than 5 weeks.

Action threshold for grassland. As a general rule, control measures for the protection of grassland are not recommended unless numbers of larvae exceed $10/m^2$ (see Section 5.2.5 for methods of counting larvae).

When herds are grazed on pastures that have recently been heavily infested by armyworm, cattle deaths may sometimes occur. Such losses have been reported by herdsmen in southern Ethiopia (Borana), Somalia, Tanzania and Kenya (Maasai), as well as from parts of southern Africa. On one occasion in Somalia, 100 cattle were reported to have died after they had grazed an area where there had been an armyworm outbreak. During the 1989 armyworm season, in three districts of northern Tanzania, 25 cattle died as a result of feeding on pasture where armyworm had been, while farmers in Mbulu district reported a 25–50% reduction in milk production during this time.

Work on *Cynodon* spp. has shown that high levels of cyanide are produced by grass infested by armyworm. Cattle may develop symptoms characteristic of cyanide poisoning after eating the grass. Cases of cattle deaths have also occasionally been attributed to the ingestion of large numbers of larvae or to the mycotoxins produced by fungi growing on armyworm faeces during the 3–40-day period after deposition, when there have been heavy armyworm infestations on pastures. These conclusions are, however, speculative and more information is required.

Key references: Bogdan (1963)
Brown and Mohamed (1972)
Brown and Odiyo (1968)
Bryson (1982)
Cheke and Tucker (1995)
Gathuru *et al*. (1991)
Georgiadis and McNaughton (1988)
Iles *et al*. (1988)
Nyirenda (1985b)

3

BIOLOGY

3.1 Description of armyworm stages

The life cycle is summarized in Figure 6. Moth behaviour is described in Section 3.2.

3.1.1 Imago (moth)

Male and female moths of *S. exempta* may be distinguished by the pattern and colour of their forewings (see Figure 7, Plate 1 and Appendix 1) and the black hair-scales on the tip of the female abdomen and racket-shaped scales on the male genitalia.

Both sexes have white hindwings with dark veins running through them.

One way of sexing damaged armyworm moths, from which the wing and body scales have been lost, is to examine the wing coupling mechanism. The coupling mechanism consists of fine hooks and bristles inter-linked on the under-side of the wings, near where the wings join the thorax. The males have a single stout bristle (*frenulum*) on the front (leading) edge of each hindwing, whereas the females have two or more bristles on the front (leading) edge of each hindwing. These bristles lock into a series of tiny hooks (*retinaculum*) on the leading edge of the under-side of the forewings, and hold the wings together during flight. The wing coupling mechanism may be examined by turning the moths upside down and looking carefully with the naked eye or with a low-powered hand lens, while gently pulling the fore- and hindwings apart.

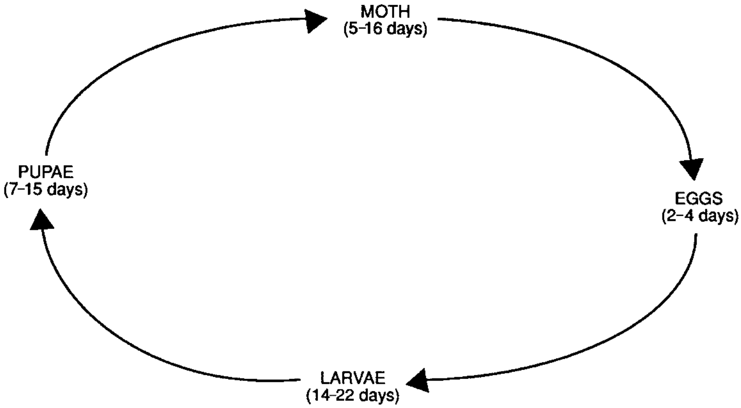


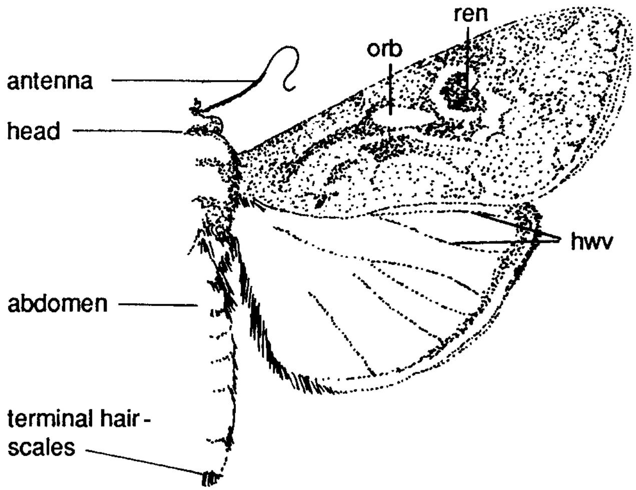
Figure 6. The life cycle of *Spodoptera exempta,* showing duration of each stage under typical outbreak conditions.

Correct identification of the species and sex of moths when wing markings are indistinct requires examination of the genitalia by dissection (see Figures 1b and 1c and Appendix 1).

3.1.2 Ovum (egg)

The individual ova (eggs) are conical, with a slightly rounded apex. The surface is densely sculptured (Plates 3a and 3b). Each egg is about 0.5 mm in diameter, and is pale yellow in colour when newly laid. As the eggs develop, they darken until just before they hatch, when the black head capsules of the larvae can be seen through the eggshells.

A female moth is capable of laying more than 1000 eggs during a period of up to 6 nights (see Section 3.2.5). The eggs are laid in batches of 10–600 on separate nights. They are deposited as a mass, typically in one layer, occasionally in two. The egg mass is protected from dehydration, parasitoids and predators by a covering of black hair-scales from the tip of the female abdomen. As successive egg batches are laid, the quantity of available hair-scales decreases, and the protective covering over the eggs becomes thinner (see also Section 4.2.1).



MALE

Forewings
Grey brown with lighter patches
Inner spot (orb) elongated and pale in colour
Outer spot (ren) kidney or arrow-shaped

Hindwings
Whitish colour with dark veins (hwv)
The front wings cover these when the moth is resting

Abdomen
Tip of the body has slate grey hair-scales

Antennae
Simple and smooth, without spines

Overall appearance
A medium-sized moth. With wings spread, measuring about 25-35 mm (1-1.5 inches)

FEMALE

Forewings
Uniformly brown-black
Inner spot (orb) distinctly elongated
Outer spot (ren) often not clearly seen

Hindwings
Whitish colour with dark veins (hwv)
The front wings cover these when the moth is resting

Abdomen
Tip of the body has black hair-scales

Antennae
Simple and smooth, without spines

Overall appearance
A medium-sized moth. With wings spread, measuring about 25-35 mm (1-1.5 inches)

Figure 7. Distinguishing characteristics of wing patterns of male and female *Spodoptera exempta* moths (after Brown and Dewhurst, 1975).

The covering of black hair-scales distinguishes the egg masses of this species from all other species of *Spodoptera*.

3.1.3 Larva (caterpillar): identification and phase

External characters. *S. exempta* larvae occur in two colour forms. These are the black, crowded or gregarious form or phase (*gregaria*) and the green, non-crowded or solitary form or phase (*solitaria*). Most casual observers do not recognize the solitary form as being the same species. A variety of other colour forms, sometimes known collectively as *transiens*, also occur. This colour variation is sometimes described as **phase**, although this definition is not strictly correct since phase, as applied to grasshoppers and locusts, involves changes in morphology as well as in behaviour and colour. For armyworm, although **form** is a more appropriate term, both definitions are used.

Gregaria phase larvae of *S. exempta* are shown in Plates 4a and 4b. The main distinguishing features are the velvety black dorsal (upper) surface with pale lateral lines (along the sides), a green or pale yellow ventral surface (under-side), and the absence of any hairs on the body. The head is shiny black. A stripe running lengthways along the top of the body is always paler than the black area on either side of it. The three white parallel lines on the dorsal surface of the prothoracic (first body) segment, the white spot at the rear of each abdominal spiracle, and the shape of the mandibles (Figure 8) are distinguishing characters in older *S. exempta* larvae.

These features are in contrast to the larvae of the following species which are often confused with armyworm (see also Section 2.2).

Helicoverpa (= *Heliothis*) species, which are covered in short hairs and typically have a speckled, not black, head capsule. The larvae are usually green or shades of brown (not illustrated).

S. exigua is often very similar to *S. exempta*, but is paler dorsally than laterally (Plate 5). *S. exigua* also occurs in *gregaria* and *solitaria* forms. A simple key for separating these two common species of armyworm larvae is given in Table 3.

Sawflies (Symphyta) are typically black or green, hairless with more than five (usually seven) pairs of prolegs on the abdominal segments (Plate 6), as compared to the *Spodoptera* larvae which lack prolegs on the first two abdominal segments.

A typical *solitaria* phase larva of *S. exempta* is shown in Plate 7. This bears little resemblance to the African armyworm larvae usually seen

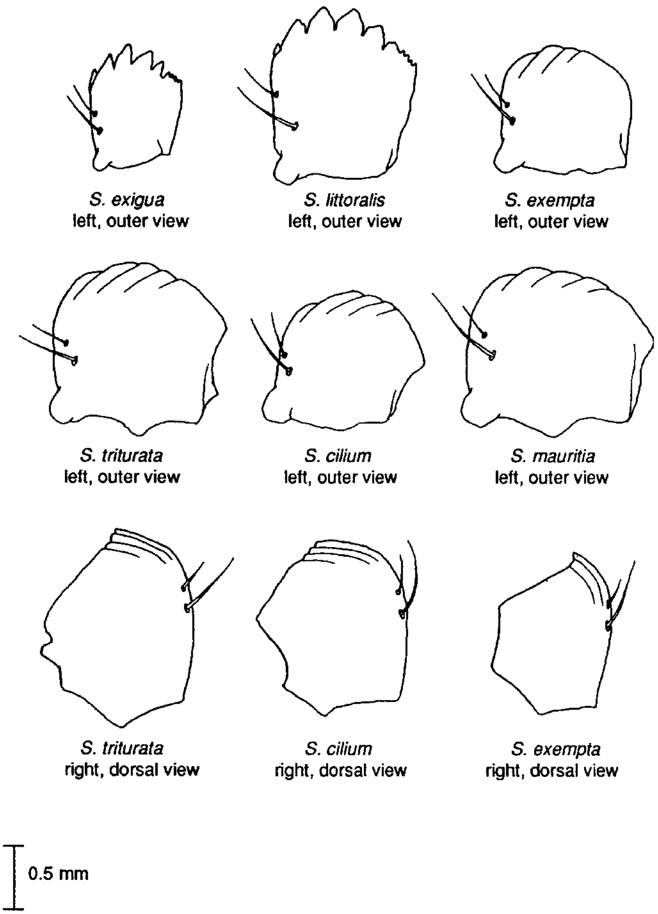


Figure 8. The mandibles of late instar larvae of *Spodoptera* spp. (after Brown and Dewhurst, 1975).

Table 3. Characters used for the recognition of *gregaria* and *solitaria* larvae of the two commonly confused species of the genus *Spodoptera* of agricultural importance in Africa

1	Larvae with hairs or spines	…. Not *Spodoptera*
–	Larvae hairless	...2
2	Larvae with black, or dark background coloration on the dorsal surface	...3
–	Larvae with green, greenish-brown or occasionally pinkish colour when seen from above	...4
3	Mid-dorsal line dark with lower area lighter; mandibles deeply serrated	...*S. exigua (gregaria)*
–	Mid-dorsal line pale, yellowish in colour, lower area on dorsal surface dark; three distinct parallel white lines on prothoracic (1st abdominal) segment; mandibles with a more or less smooth cutting edge	...*S. exempta (gregaria)*
4	Lateral edges of abdominal segments with darkened, somewhat triangular marks, becoming smaller near the head and larger distally	...*S. exempta (solitaria)*
–	Lateral edges of abdominal segments with triangular markings of uniform size	...*S. exigua (solitaria)*

Adapted from Blair (1968) and Brown and Dewhurst (1975).

by farmers in outbreaks. The solitary phase larvae, especially in the early instars, are difficult to distinguish from the larvae of other species found in grasslands as they are cryptically coloured, being shades of green, brown or pink. The head is pale or speckled, never black.

During the daytime these solitary larvae hide at the bases of the food plants, and feed at night. When they are found, they are normally curled up, fat in appearance and sluggish in their behaviour. *Solitaria* larvae may be found at quite high densities (10/m^2), provided they are not in contact with one another (e.g. if they are in thick vegetation). Usually they are sparsely distributed and difficult to find. In Kenya, 6 h of searching time have been required to find one larva, and in South Africa it took up to 8 h to find a single larva. The larvae may be more easily found at night by the light of a torch.

Variations in physiology. Change in phase is induced principally by crowding, but is partly controlled genetically. When larvae from *gregaria* populations are reared through several generations in continuous isolation, the *solitaria* characteristics of successive generations intensify. This implies a genetic component in phase determination.

Biochemical differences have been reported between *solitaria* and *gregaria* larvae. Gregarious larvae have been shown to contain:

- higher fat content
- greater concentrations of lactic acid and hydrogen ions in the haemolymph
- lower concentrations of juvenile hormone
- lower concentration of uric acid in the haemolymph
- lower protein and glycoprotein content.

Laboratory studies suggest that a hormone produced by the sub-oesophageal ganglion has an effect on colour change.

Behaviour. The gregarious phase larvae of *S. exempta*, characteristic of crowded populations, are adapted physiologically and behaviourally for accelerated larval development. In the field, gregarious larvae increase their body temperature by exposing themselves to the sun and by avoiding shade. High body temperatures are achieved by absorption of solar radiation, which is aided by the black pigmentation of the larvae, and result in elevated metabolic rates and rapid development. Feeding rates are increased and there may be fewer larval instars.

Older gregarious larvae may ‘march’ in large numbers. It is this characteristic which has given rise to the name armyworm (Plate 8). Marching appears to be related to:

- the search for food when the host plants have been depleted
- mutual stimulation due to crowding
- the search for pupation sites.

3.1.4 Larvae: development

Young larvae. On hatching from the eggs during the early hours of the morning, the translucent larvae (which have black head capsules) feed on their eggshells for about 1 h before spinning silken threads on which they are dispersed by the wind (Plate 9). Windborne larvae can drift for many metres. For example, on one occasion in Zimbabwe, young larvae were found on all the 1-day-old maize plants sprouting in a ploughed 50-ha field. As there were no weeds on which eggs could have been laid, the larvae must have been carried on the wind in large numbers to infest the field.

Newly hatched larvae are only capable of feeding on the young leaves of their host plant. They do so by rasping the epidermis on the underside of the leaf with their sharp (serrate-edged) mandibles. The effect of feeding by the young larvae is skeletonization of the leaf of the food plant, making a pattern on the leaves descriptively termed ‘windowing’ (Plate 10).

Growth. As larvae grow, they pass through a series of moults. The stage between each moult is known as an **instar**. Each moult is preceded by a reduction in activity and a cessation of feeding. The number of instars is variable, with food quality and phase being the major influences. Typically, there are five or six instars, rarely seven. It is during the last two to three instars that the larvae have voracious appetites and do most damage to crops and grassland (Figure 9). The convention is that instars are referred to using Roman numerals (I–V, VI or VII).

The larvae remain green during the first three instars whether they are at high or low densities. At the moult from III to IV instar, the crowded and solitary phase larvae diverge by changing phase, colour and character. At this stage, according to whether they are crowded together or not, *solitaria* larvae stay green, whereas *gregaria* larvae change colour to black.

The width of the head capsule remains unchanged for the duration of each instar but increases at each moult. In *S. exempta*, within instars the variability in head capsule width is so great between cohorts* that it is often difficult to assign any one larva to a particular instar. For this reason, the **mean head capsule** width of large samples (not less than 50 larvae), collected at random from an outbreak (see Section 5.2.5) is used to estimate the age of a population. Table 4 shows examples of mean head capsule widths for all instars, whether for V, VI or VII instar populations.

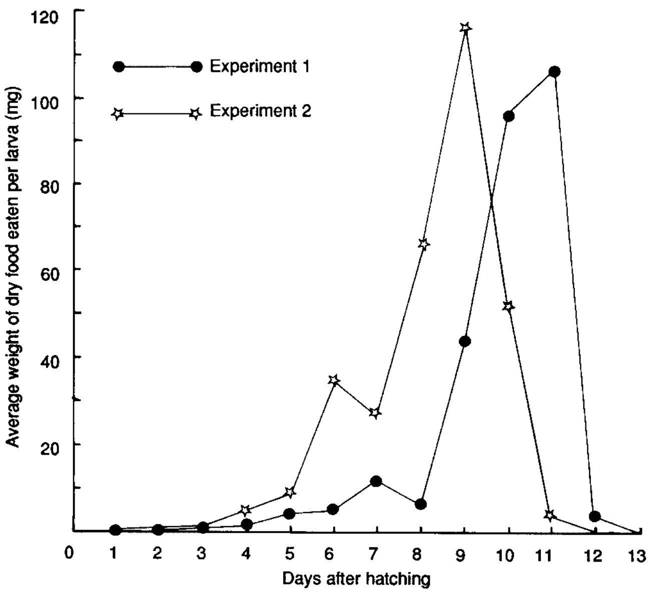


Figure 9. Increase in food consumption by *Spodoptera exempta* larvae during their development (after Brown and Odiyo, 1968).

* Cohort – larvae from a sample collected at the same time and place.

As the larvae moult from third (III) to fourth (IV) instar, the mandibles also change form and function from a rasping edge to a cutting and grinding edge (Plate 11), enabling the larvae to feed from the leaf edge. This form of damage to the host plant is more easily seen than the windowing.

It is possible to determine the sex of both *solitaria* and *gregaria* larvae older than the third instar by the presence, in the female, of small pits on the ventral surface of the 8th and 9th abdominal segments.

Feeding. When young larvae start feeding, they are positively phototactic and negatively geotactic, i.e. they are attracted to light and climb upwards. This brings the larvae to the upper parts of the plant, which are often the youngest parts richest in nitrogen. Young larvae feed on the lower epidermis (under-side) of the leaves and gradually become green (Plate 10).

S. exempta larvae will feed on a wide range of Gramineae, Cyperaceae and certain non-Gramineae (see Appendix 2 and Section 3.3). The age, species and quality of food plants affect the duration of larval

Table 4. Examples of mean head capsule widths of all larval instars in V, VI and VII instar populations of *Spodoptera exempta*

	V instars		VI instars		VII instars	
Instar	**Mean (mm)**	**SE**	**Mean (mm)**	**SE**	**Mean (mm)**	**SE**
I	0.34	0.008	0.35	0.008	0.35	0.008
II	0.55	0.003	0.48	0.003	0.53	0.078
III	0.93	0.009	0.82	0.010	0.74	0.018
IV	1.52	0.006	1.21	0.012	0.95	0.004
V	2.55	0.011	1.92	0.015	1.37	0.015
VI			2.56	0.016	1.86	0.019
VII					2.51	0.023

After Yarro (1985); larvae reared on different food plants.

development. The first flush of plant growth at the start of the rains contains high levels of nitrogen, giving a rich source of protein for the young larvae.

Older larvae feed mainly on the edges of the middle leaves of the plant, where the leaves lie horizontally and photosynthetic activity is consequently greatest. As the larvae grow older, their food consumption increases rapidly until it reaches about 0.7 g/day for full-grown (last instar) larvae feeding on maize (Figure 9). This represents more than a 550-fold increase in the weight of the food eaten from the time of hatching until the end of the last instar, and amounts to about 2.0 g of fresh maize leaf per larva during the peak feeding period of the last instar.

Larvae feeding in large numbers on the leaves of trees, bushes or other broad-leafed plants are unlikely to be *S. exempta*. There have, however, been occasional reports, a few confirmed, of armyworm larvae feeding on non-graminaceous host plants (see Section 3.3 and Appendix 2).

In general, the more nutritious the food plant, the shorter the armyworm life cycle, the fewer the number of instars and the higher the survival rate.

Mortality. There can be very high mortality of young larvae at hatching, due to the larvae failing to locate a suitable food plant. Young larvae can only live for about 1 day without food and older grasses and cereal crops may be too coarse for them to feed on. Small larvae have been observed trapped and drowned by rain drops and others washed off food plants by rain. Larvae at any stage may be drowned during periods of prolonged heavy rain.

All stages of the life cycle are vulnerable to parasitoids and predators, and the larvae are particularly liable to infection by pathogens. If larvae are parasitized by certain Hymenoptera such as *Campoletis pedunculata* (Ichneumonidae) and *Euplectrus laphygmae* (Eulophidae), this reduces and eventually inhibits feeding, thus apparently extending the larval instar duration by several days. This is often noticeable in instars III and IV (see Section 3.4). Natural enemies recorded from *Spodoptera exempta* are listed in Appendix 3.

Generation time. There are typically 6–8 outbreak generations annually in eastern Africa and 4–5 in southern Africa, with an ‘off-season’ of 3–5 months when outbreaks are not reported. In areas favourable for low-density populations to persist throughout the year, there is the potential for up to 13 generations to occur.

During the **outbreak season**, November to June in many parts of eastern, central and southern Africa, the average generation time is about 1 month. With the high temperatures found at low altitudes this may be reduced to 2 weeks.

At other times of year (in the **off-season**), under cool conditions such as are found in the southern winter or in the highlands, the generation time may be extended for as long as 10 weeks. There has been one report of a greatly extended duration (diapause) in the pupal stage, but this has not been confirmed by subsequent investigations.

The development of gregarious phase larvae is well synchronized in an outbreak so that most larvae pupate over a relatively short time (3–7 days). The development of solitary phase larvae is staggered and slower so that pupation is spread over a longer period and subsequent generations may overlap.

The duration of the different stages in the life cycle is affected particularly by temperature, but also by larval density, sex, effects of parasitism, and type and quality of food plant.

Temperature. Minimum temperature threshold requirements for the development of the various life stages are given in Table 5. The effect of temperature on the development of all stages has been studied in the laboratory, as well as in the field. At constant temperatures below 15 °C the hatch of ova is poor and larvae which hatch fail to survive.

When rearing larvae under constant temperature conditions, solitary phase larvae take longer than gregarious phase larvae to reach the pupal stage.

Because of the longer development times at high altitude due to the lower temperatures, three different life tables were developed, each based on detailed field observations of infestations in representative areas frequently subject to outbreaks (Table 6).

The range and typical duration of the different stages of development on the life cycle in outbreaks are given in Table 7.

Density. Outbreaks can occur over large areas, within which aggregations are found in a 'clumped' distribution among more scattered larvae. Many infestations may occur simultaneously but they are rarely continuous, which makes it difficult to delimit their boundaries and estimate the size of the infested area. The density of larvae varies considerably both within and between outbreaks, with larvae occurring in densities from less than one up to and sometimes exceeding 1000/m^2, typically tens to hundreds. Estimation of the density of larvae within any outbreak is, therefore, at best an approximation and should be undertaken using quadrats (see Section 5.2.5).

Table 5. Minimum temperature requirements for the development of different stages of *Spodoptera exempta* from experiments at constant temperature

Stages	Min. temperature (° C)	Notes
Ovum	12	No hatching (Hattingh found some hatching at 10 °C)
Larva	14	No survival to pupation
Pupa	13	No emergence
Imago	20	Very little oviposition at this temperature and below
Ova – imago	14	No survival

Data of Hattingh (1941) and Ma in ICIPE (1974).

Table 6. Life tables for *Spodoptera exempta* used in forecasting to estimate, from larval samples, the dates of oviposition (Ov.) and emergence (Em.) of that population by counting the number of days backwards or forwards

Stage	Nairobi area, Kenya* 36 days		Harare area, Zimbabwe† 30 days		Kenya Coast‡ 23 days	
	Ov.	Em.	Ov.	Em.	Ov.	Em.
Ovum	3	33	3	27	2	21
I	8	28	6	24	5	18
II	10	26	8	22	6	16
III	12	24	10	20	8	15
IV	14	22	12	18	9	13
V	19	17	15	15	12	11
VI	25	11	20	10	16	7
Pupa	31	5	26	4	19	4
Moth	36	0	30	0	23	0

* Unpublished field data.

† Rose (1975a).

‡ Brown and Odiyo (1968).

Table 7. Duration (days) of the different stages of development in the life cycle of *Spodoptera exempta*

Stage	Range	Average*
Ovum	2– 5	3
Larva	11–24	21
Pre-pupa and pupa	7–12	10
Pre-oviposition period (PRP)	2–13	3
Total	22–54	37

*Average under typical outbreak conditions.

Pupation. The full-grown larvae seek soft damp soil, the base of plants or sandy banks in which to burrow and pupate (Plate 12). This is a critical period for survival. If the soil is too dry and hard many larvae will perish. If there is rain at this time, farmers will often report that all larvae have been killed, whereas in reality they have burrowed underground to pupate (Plate 13).

Having ceased feeding and burrowed underground, the larva constructs a flimsy, silk-lined chamber, 2–3 cm below the surface. It rotates itself into a head-upwards position and contracts in length (Plate 14). This stage is known as the pre-pupa and lasts 1–2 days (this period is usually included in the calculation for pupal duration). The cuticle of the pre-pupa then splits lengthways from just behind the head and is cast off from the tail end, leaving a soft pale green pupa which gradually hardens and darkens (see Section 3.1.5. and Plate 14).

Key references: Brown and Odiyo (1968)
Faure (1943a)
Gatehouse (1986)
Hattingh (1941)
Janssen (1993a)
Ma, in ICIPE (1974)
Matthee (1946)
Page (1988)
Page and Dewhurst (1987, 1992)
Persson (1981)
Simmonds and Blaney (1986)
Yarro (1982)

3.1.5 Pupa (chrysalis)

The pupa is pale green and soft when newly formed; its colour gradually deepens to a deep red brown once it hardens.

The pupae of all *Spodoptera* species are similar and may be distinguished from one another only with difficulty. If, however, by digging, large numbers of pupae are found over a wide area, they are likely to have resulted from an earlier outbreak of African armyworm.

Pupae may be sexed easily, using a low-powered hand lens and looking at the lower surface of the pointed tail end of the pupa. Male and female pupae are illustrated in Figure 10.

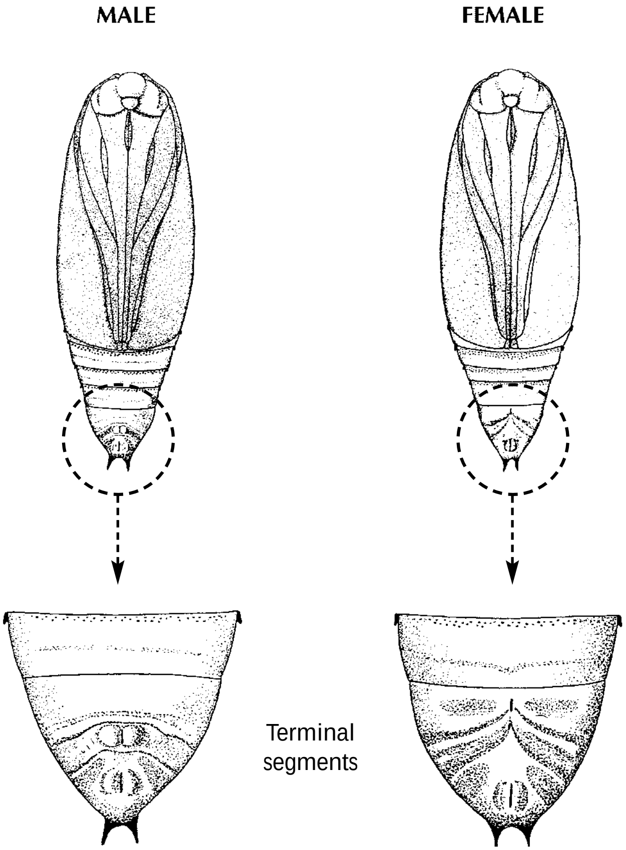


Figure 10. Features of the ventral surface of pupae for distinguishing males and females.

When the moth is ready to emerge from the pupa, usually after 7–12 days, the cuticle splits down the back of the head and the emerging moth pushes its way up through the soil to the surface, where a small drop of moisture has been observed, produced by the emerging moth. The purpose and composition of this droplet is unknown, but it may be to moisten and soften the soil and thus ease the moth's exit. Once out of the ground, the moth climbs the nearest grass stem or other available vertical surface to expand its wings (see Section 3.2.1).

3.2 Moth behaviour

3.2.1 Emergence

Moths emerge from pupae in the ground during the early part of the night. The time of peak emergence may vary from night to night between the hours of 20.00 and 22.00 (Figure 11). Moth emergence from a single outbreak may extend over a period of about 12 days (Figure 12).

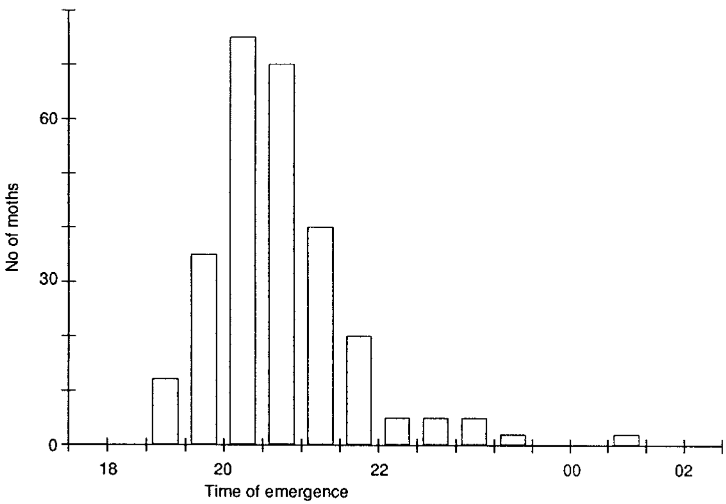


Figure 11. Times of *Spodoptera exempta* moth emergence at night.

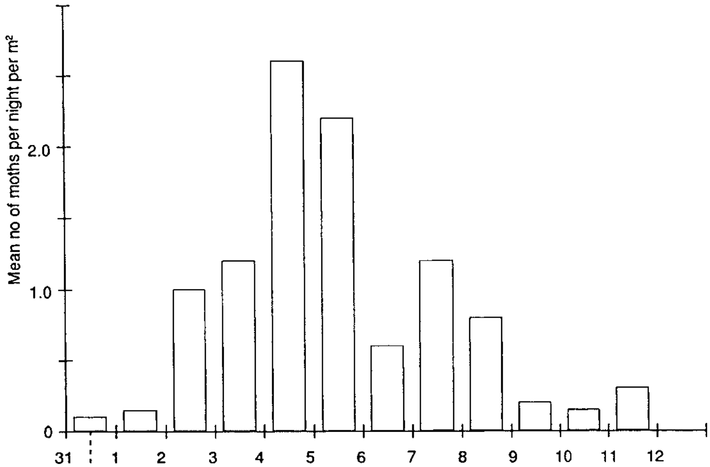


Figure 12. Period of emergence and number of moths emerging from an outbreak in Kenya, March–April 1984.

Immediately after emergence, before expanding their wings, the moths, (imaginatively likened to 'tadpoles'), climb up grass stems or other nearby vegetation, where they remain motionless while haemolymph (insect blood) is pumped into the wing veins. Once the wings have expanded they are held at right angles to the body of the insect in a position more characteristic of butterflies (both stages are shown in Plate 15). The wings are then flicked down on either side of the body into the typical 'moth' position. When their wings have dried, the moths fly away. The whole process takes about 1.5–2 h. The times taken for each stage vary with temperature and are illustrated in Figure 13. It is not known what effect humidity may have on the process.

The majority of newly emerged moths fly into nearby trees. Flying up from the grasses, the moths fly downwind past a tree and then turn back to settle in the lee of the nearest tree (Plate 16). Moths that do not settle in trees fly up into the night sky and are carried away downwind. The moths in the trees are in an active state (antennae raised) and they may remain there throughout the night before flying off at dawn to find shelters in which to hide during the day (day shelters). Some moths fly from the trees at any time during the night in small groups (known as ‘plumes’) and migrate away downwind from the site of emergence.

If rain falls during the night, moths that are resting in trees rapidly descend to the ground, where they shelter among the vegetation until the rain stops, before they fly back again into the trees.

The phenomenon of flight into trees, where they become concentrated, follows a similar behavioural pattern every night during the period of moth emergence. Numbers reach a peak in the trees at about midnight. Only trees in the immediate vicinity of an outbreak and those greater than 1.5 m in height are used by the moths. Many thousands of moths may be found in each small tree in an outbreak area.

The moths remaining in the trees at dawn leave on short flights which end with the moths hiding during the day in grass clumps, under

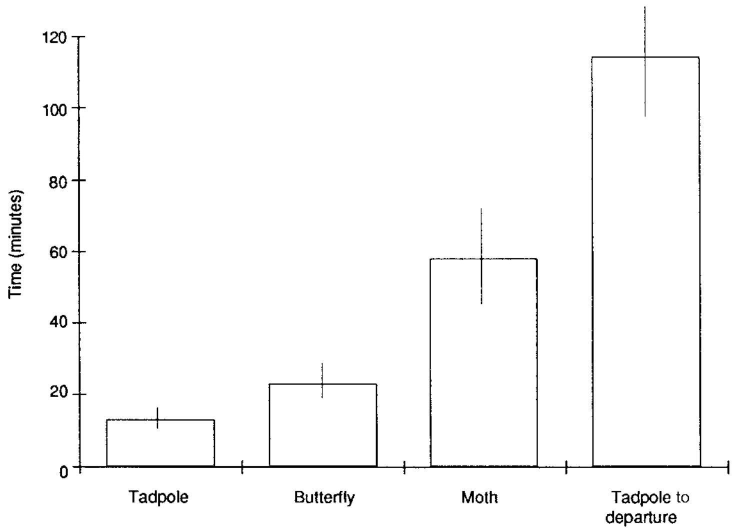


Figure 13. Histogram showing duration of each stage of emergence of *Spodoptera exempta* moths; standard deviations shown as bars.

cowpats (Plate 17), in crevices, under stones (Plate 18) or under the bark of trees. They emerge from these day shelters at dusk, some to depart downwind on migration flights, while others fly briefly into trees before leaving the area during the night.

Three peaks of flight activity have been observed in the field, at dusk, in the middle of the night and at dawn. Figures 14 and 15 summarize the emergence pattern, the numbers of moths moving into trees and the three flight periods at 'dusk', 'midnight' (= middle of night) and 'dawn' (see also Section 3.2.2).

The 'dawn' flight results only in local displacement, ending with the moths seeking day shelters. Moths do not normally fly during the day, although large numbers have been seen flying around and feeding on *Acacia nubica* (Benth.) flowers during the day at Gelai near Lake Natron, Tanzania.

Key references: Riley *et al*. (1981, 1983)
Rose and Dewhurst (1979)

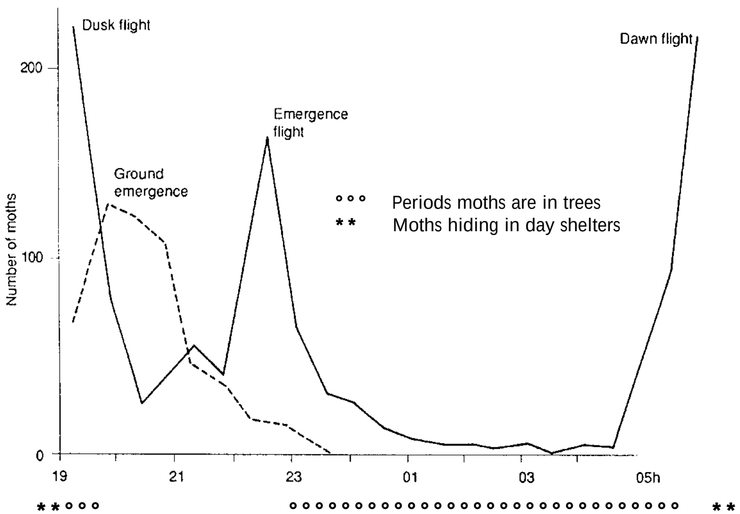


Figure 14. Emergence and flight periods of *Spodoptera exempta* moths during one night (field observations).

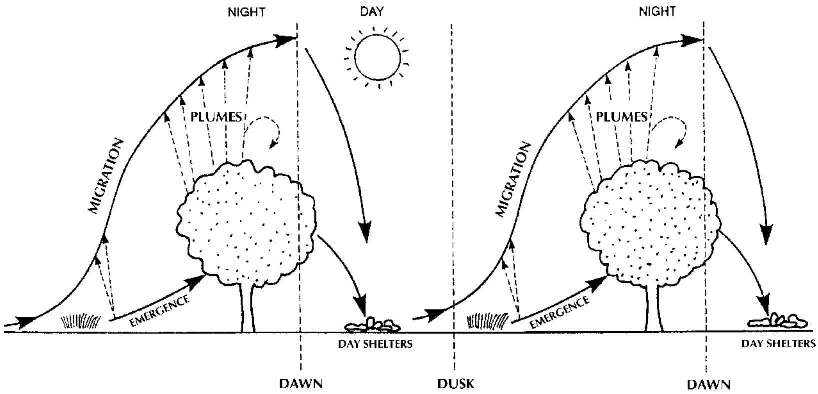


Figure 15. Schematic summary of moth behaviour at an emergence site.

3.2.2 Flight

The main migration flights start at dusk or in the first part of the night, the dusk flight being generally the more important. On migration flights, moths ascend rapidly to an average maximum height of 420 m above ground level in open savannah country, although in the Kenya Rift Valley, near Nairobi, the average maximum height was 870 m.

Moths leaving an emergence site move more or less downwind because their airspeed (about 5 km/h as measured in the laboratory and 3.0±0.5 km/h as observed in the field) is generally less than the wind speed. Movement continues to be downwind, even on nights when the surface wind decreases to near calm, because the wind is still blowing at the height at which the moths are usually flying.

During the main outbreak season, the prevailing easterly winds over East Africa are strong during the first half of the night. The winds at several 100 m altitude may even increase during the night. As dawn approaches, the easterly winds often become weak and variable. At this time moth orientation may become upwind or crosswind and displacement distances are consequently reduced.

Radar observations have shown that the mean height of moth flight often corresponds to the level of the strongest mean wind speed in the prevailing easterly winds. As a result, those moths that fly for several hours can move 100 km or more in one night or up to several hundred kilometres in a sequence of only a few nights.

Marked *S. exempta* moths have been recovered at intervals up to 147 km downwind from an emergence site. However, studies of the spread of outbreaks indicate that the moths may be displaced over much greater distances than this. Such studies have indicated that movements of 200 km are quite common and that 700 km is not exceptional. *S. exempta* moths have been recorded arriving on board a ship in the Gulf of Aden, 87 km from the nearest land and 122 km from the African coast. In New Zealand, a few *S. exempta* moths have been recovered 3200 km from their nearest known source. Flights over sea are probably exceptional in that the moths must have continued to fly during the daytime.

Radar observations have shown that *S. exempta* moths do not fly in cohesive swarms like desert locusts, but become dispersed as they are carried downwind. They have also shown that migrating moths descend to the ground if moderate or heavy rain falls on them when they are flying, as do moths resting in trees (see Section 3.2.1).

Moths become widely dispersed downwind in both space and time because of the extended emergence period (about 12 days), the subsequent emigration period, the different departure flight times from the trees, different flight duration for different individuals and their continuing dispersal in the air. They, therefore, have to be reconcentrated by wind convergence (see Section 4.1.1) for outbreaks to be produced in the next generation.

All moths emerging from outbreaks appear to leave the immediate vicinity of the emergence site by downwind migratory flight. *S. exempta* must, therefore, be considered as an obligate migrant. Light trap data show that migrating female moths are sexually immature.

Reinfestation by successive generations rarely occurs at the same site. However, when this does occur it appears to be associated with inhibition of migratory flight by rainfall soon after local moth emergence, or by reinvasion with more moths coming in from a distance. Such subsequent outbreaks may suffer high mortality due to natural enemies and pathogens (see Section 3.1.4 and Appendix 3).

Some moths reared in the laboratory have survived for as long as 36 days, but typically in the field they survive for 7–16 days, with females living longer than males.

Key references: Aidley (1974)
Brown (1970a)
Riley *et al*. (1981, 1983)
Rose *et al*. (1987)

3.2.3 Pre-reproductive development

Newly emerged female *S. exempta* moths have small oocytes (developing ova), some of which grow during the first 20 h to half the size of the final ova. This stage is reached by the time of the dusk flight of the evening following emergence, and is termed the 'potentially arrested development' stage.

In some moths, the oocytes continue to develop until oviposition. Other moths, however, may halt oocyte development and may continue in this state of arrested development for up to 13 days or more after moth emergence, before completing the development of their oocytes and laying eggs.

Oocyte development is both genetically and environmentally controlled. Genetic control of oocyte development is reflected in variations in the pre-reproductive period between populations. Separate lines of long and short pre-reproductive period moths have been produced in the laboratory by selective breeding.

Water must be available for the female moths to drink, for the complete development of their oocytes. *S. exempta* moths do not need food before oviposition, but if they do feed on plant nectar or honeydew, fecundity is increased and more eggs are laid. The fecundity of moths derived from *gregaria* larvae is greater than that from *solitaria* larvae.

S. exempta moths have been seen to feed on nectar and drink dew before, between and after migration flights. Moths are readily attracted to molasses bait sprayed on to trees.

The flight potential of armyworm moths is also genetically determined. The duration of flight on any one night varies from moth to moth. By selective breeding, the proportions of 'long-flight' and 'short-flight' moths in separate lines can be increased.

Genetically determined flight performance is moderated by larval density. Moths reared from crowded larvae show enhanced flight performance compared with their siblings reared solitary. At emergence, moths from gregarious larvae have a higher fat (glyceride) energy reserve and lower water content than moths from solitary larvae. Armyworm moths, therefore, have varying flight potentials, as a consequence of different pre-reproductive periods. The result is greater dispersal in both space and time by moths from gregarious populations.

Male moths reach reproductive maturity earlier than females and there is little variation between populations. It has been concluded that males may continue migration after reaching maturity, mating with females at different locations on different nights.

Key references: Fox (1978)
Gatehouse (1989)
Gunn *et al*. (1988)
Laird (1962)
Page (1985, 1988)
Parker and Gatehouse (1985a, 1985b)
Rose *et al.* (1985)
Woodrow *et al*. (1987)

3.2.4 Copulation (mating)

At the end of the migration flight *S. exempta* moths again settle in trees, where mating takes place either on the same night or during the following night. The behaviour in trees at this time differs from that after emergence (see Section 3.2.1) in that the moths move into the trees earlier in the night and tend to remain there until the dawn flight. A sex pheromone (mating atractant) is produced by the sexually receptive female moth ‘calling’ to attract a male.

S. exempta females can become sexually receptive from the second night after emergence and may continue to be so for up to at least five nights. They will only release the pheromone at specific times of night when they are ready to mate. Once mated, females produce less pheromone for any subsequent mating.

Only sexually receptive males are attracted by the pheromone. Once they have detected the pheromone *S. exempta* males fly upwind, following the odour plume to its source by flying a zigzag flight path to intercept the plume repeatedly, until they reach the female (see also Sections 5.2.1 and 5.2.2).

During copulation pairs of moths are joined by the tips of their abdomens. A spermatophore (a capsule enclosing the sperm – see Figure 1d), which is characteristic for the species, is produced by the male and introduced via the aedeagus (penis) into the bursa copulatrix of the female to fertilize the eggs. After copulating, the aedeagus is withdrawn.

Copulating pairs of *S. exempta* moths have been found between 21.00 h and 05.00 h with a peak between midnight and 02.00 h (Figure 16). As many as ten copulating pairs have been observed in one tree after midnight.

A single mating will provide enough sperm to fertilize all the eggs the female will lay, but many females mate more than once. The successful mating frequency can be found by dissecting the female and removing the tough, translucent, creamy-white spermatophores from the bursa

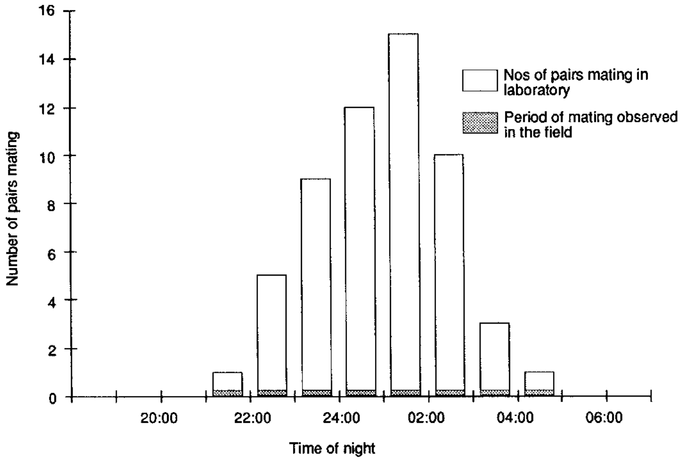


Figure 16. Observed times of mating by *Spodoptera exempta* moths in the laboratory and field.

copulatrix. Females have been found to mate up to seven times, but usually one or two spermatophores are found.

In Lepidoptera, the sex pheromones produced by the female moths differ from the pheromones produced by the males. Male pheromones have only a short-range effect, are usually produced when the male is in close proximity to a female and apparently act as aphrodisiacs that are important in the courtship prior to copulation. A male pheromone has not been identified in the African armyworm, *S. exempta*.

Key references: Brown and Dewhurst (1975)
Dewhurst (1984)
Khasimuddin (1978a)
Rose *et al.* (1987)

3.2.5 Oviposition (egg laying)

Moth behaviour during oviposition was studied in an area in which immigrant *S. exempta* moths had settled and mating pairs had been seen in trees. Female moths were found throughout most of the night sitting in a head-up position on grasses. During the early part of the night some females were observed to have turned to a head-down position on the grass stems or other substrate. Oviposition followed, with the female *S. exempta* slowly moving her abdomen from side to side as she moved down the substrate, covering the eggs with the black hair-scales from the tip of her abdomen (Plate 19). Typically, in the laboratory, oviposition begins between 20.00 h and 21.00 h and a complete egg batch is laid in about 30 min. There is a second oviposition period by some females just before dawn (Figure 17).

A female is capable of laying eggs for up to six nights, with peak numbers of eggs being laid on the second night. Individuals may lay up to eight batches of eggs, although more than six is unusual. After the second night, the number of eggs laid in a batch decreases as the moth gets older; egg masses can contain between 10 and 600 eggs. A single female can lay between 400 and 1300 eggs during her life (see also Section 4.2.1).

There is a strong correlation between the weight of the female pupa and number of eggs laid by the female moth (about 660 ova per 100 mg weight of pharate* adult). Moths from *gregaria* populations lay more eggs than those from *solitaria* populations except when the *solitaria* moths have access to nectar, when comparable numbers of eggs are laid.

Oviposition sites are not necessarily on host plants. Ova may also be laid on other available substrates, for example, on tall plants, leaves and twigs of bushes or trees, dry grass stems, or on buildings. The higher the mass of eggs is above the ground, the further the newly hatched larvae are likely to be dispersed by the wind on the silken threads produced by the larvae (see Section 3.1.4).

Key references: Brown (1970a)
Brown and Swaine (1965b)
Gunn and Gatehouse (1985, 1987)
Hattingh (1941)
Janssen (1993a)

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* Pharate – fully formed adult still within the pupa.

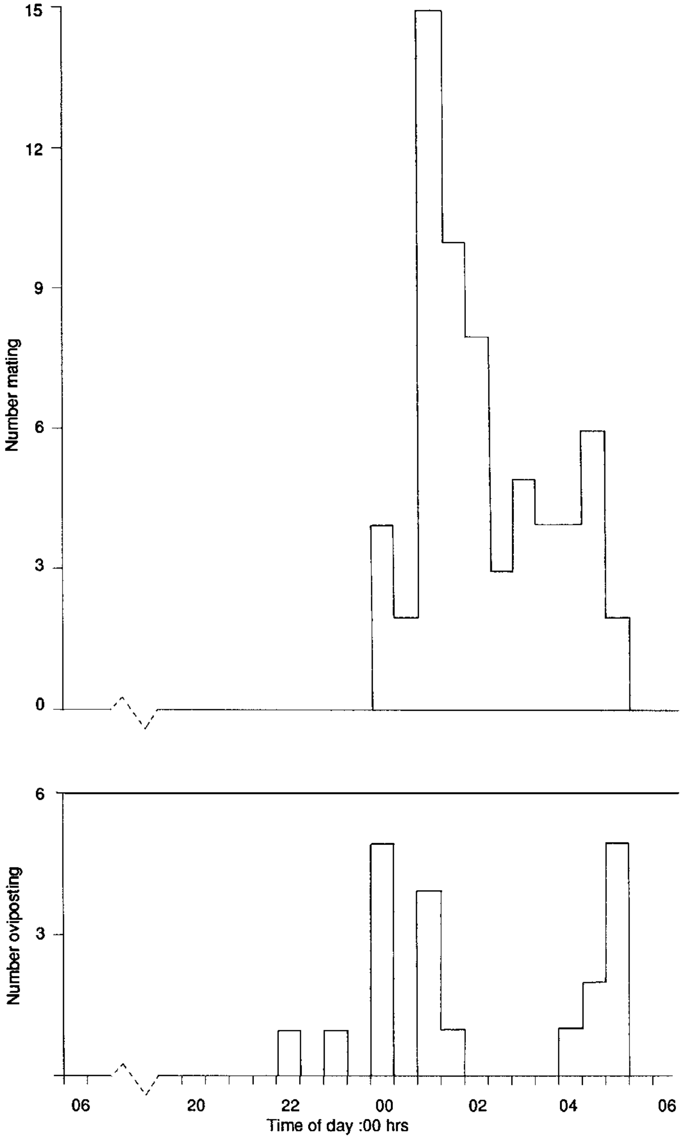


Figure 17. Observed times of mating and oviposition by *Spodoptera exempta* moths in the laboratory.

3.3 Host plants

Armyworm larvae normally feed on plants of the families Gramineae and Cyperaceae. Among the host grasses most commonly eaten are perennials, such as *Cynodon dactylon* and *Pennisetum* spp., as well as many annual grasses including *Eleusine indica* and *Urochloa* spp.

S. exempta larvae have once been found causing considerable damage to coconut seedlings (*Cocos nucifera*) and, on another occasion, to young tea plants (*Camellia sinensis*). During large-scale outbreaks, non-host plants including tobacco (*Nicotiana tabacum*) and cotton (*Gossypium* spp.) may also be eaten.

The major crops of economic importance which are damaged are:

Barley	(*Hordeum vulgare*)
Bulrush millet	(*Pennisetum americanum*)
Finger millet	(*Eleusine coracana*)
Maize	(*Zea mays*)
Oats	(*Avena sativa*)
Rice	(*Oryza sativa*)
Sorghum	(*Sorghum vulgare*)
Sugarcane	(*Saccharum officinarum*)
Teff	(*Eragrostis tef*)
Wheat	(*Triticum vulgare*)
Pasture grasses, especially	*Cynodon* and *Pennisetum* spp.

S. exempta larvae show marked food preferences within the Gramineae. Varieties of cereal crops vary greatly in their susceptibility to attack.

Appendix 2 gives a list of grasses (Gramineae), sedges (Cyperaceae) and other plants occasionally recorded as being eaten by *S. exempta* larvae. It should be noted that 'sweet' grasses preferred by livestock are also favoured by armyworm larvae.

Key references: Brown (1962)
Brown and Dewhurst (1975)
Brown and Odiyo (1968, 1969b)
Page and Dewhurst (1987, 1992)

3.4 Natural enemies

Armyworms of *S. exempta* are attacked by a great variety of natural enemies, including parasitoids, pathogens (bacteria, protozoa, viruses and fungi) and predators (see Appendix 3). Some of the natural enemies sometimes play an important role in reducing populations, although, with a migrant species such as armyworm, their influence is difficult to quantify. Because *S. exempta* has a short life cycle and is a migrant there is seldom an opportunity for parasitoids to build up to numbers sufficient to kill a significant proportion of a population before it has completed its life cycle and moved out of the area. If there are subsequent outbreaks in the same area, however, parasitism will increase and may well affect in excess of 70% of the later generations of larvae.

3.4.1 Parasitoids

Parasitoids are represented by Diptera (flies) (Bombyliidae, Tachinidae), and Hymenoptera (wasps) (Braconidae, Chalcididae, Eulophidae, Ichneumonidae, Pteromalidae and Scelionidae). Of the 19 genera of Diptera and 23 genera of Hymenoptera recorded so far, only four Diptera and four Hymenoptera are regularly found in appreciable numbers. A sceleonid wasp, *Telonomus* sp., parasitizes armyworm eggs in Zimbabwe, and may also do so in southern Tanzania, from where an unconfirmed report was received.

Parasitic nematodes, from the genera *Mermis* sp. and *Gordius* sp. are sometimes found in moths (Appendix 3).

3.4.2 Pathogens

Virus. *S. exempta* is attacked by a lethal, highly infectious insect virus, the *S. exempta* nucleopolyhedrovirus (previously nuclear polyhedrosis virus) or SpexNPV. This is the most important natural cause of mortality in armyworm outbreaks and while the disease in *S. exempta* outbreaks has been reported since the 1930s, it was only in 1963 that this was firmly identified as an NPV. This virus is specific to *S. exempta* and is shown in Plate 20a under a light microscope.

During the first outbreaks of the season this NPV is rare but it becomes increasingly common as the season progresses and mortality in later outbreaks may reach 98% causing the collapse of many outbreaks. The SpexNPV is infectious to the larvae only and the normal route of infection is when the infective particles (called occlusion bodies or polyhedra) are ingested while feeding. The virus subsequently infects the cells of the midgut, proliferates in these then spreads throughout the insect to the tissues of the fat body, haemocoel, trachea and epidermis. The larvae normally die 5–7 days after infection with the body a mere sack of disintegrating tissue and progeny virus. In such larvae the NPV may proliferate to reach 200 million viral particles per larva. A dying larva will often climb to the top of a plant (see Plate 20b) where it ruptures and leaks infective particles on to the plant that other larvae may ingest to complete the cycle of infection.

SpexNPV can also exist as a ‘latent’ or asymptomatic form in which it can persist in insects (larvae, pupae and adults) and pass from generation to generation with no overt sign of disease. This ‘latent’ virus can be activated into a lethal infection in the larva by stresses such as overcrowding, heat, cold or poor diet and may be an important mechanism for the SpexNPV’s persistence in low-density populations of *S. exempta* between armyworm seasons.

The ecology and management potential of SpexNPV is now under investigation as a potential control agent for *S. exempta*. Field trials have shown that artificially propagated SpexNPV can be sprayed on to outbreaks to initiate disease outbreaks and trigger population collapses. As a highly specific pathogen of *S. exempta,* it has the advantage over chemical insecticides in that it is self-replicating and has been found to be completely safe to humans, animals, and even other insects.

Another virus, a cytoplasmic polyhedrovirus, can also infect *S. exempta* though its ecological importance is much less understood. The cytoplasmic virus (CPV), kills both pre-pupae and pupae.

Fungi. Fungi are of little importance as a natural mortality factor as they require particular conditions of high humidity and temperature for propagation. The fungus commonly found attacking armyworm is *Nomuraea rileyi* (Plate 21).

Bacteria and protozoa. Bacteria and protozoa are, in general, of only minor importance. The use of strains of the bacterium *Bacillus thuringiensis* formulated as a biocide may provide a control agent in the future.

3.4.3 Predators

Predators include both vertebrates (mainly birds) and invertebrates (e.g. ants, beetles, thrips and spiders). Ants and thrips destroy eggs and ants kill many young larvae. Storks and crows may decimate smaller outbreaks, but have little effect on larger ones (see Appendix 3).

Solitaria S. exempta larvae may not be seen by birds because they occur at low density, and their cryptic colour and secretive habits makes them difficult to find.

Key references: Bai *et al*. (1992)
Brown and Swaine (1965a)
Merrett (1986b)
Odindo (19881a)
Persson (1981)
Swaine (1966)

4

EPIDEMIOLOGY: THE ONSET AND SPREAD OF ARMYWORM OUTBREAKS

4.1 Effects of weather

4.1.1 Effects of wind on moth migration

The direction and speed of winds at the time of migratory flight play a major role in determining the distances travelled by *S. exempta* moths. The flight speed and behaviour of the moths themselves (see Section 3.2.2) may also, to some extent, influence where they settle to mate and lay eggs.

Because moths **disperse** downwind from their emergence sites (see Section 3.2.2), outbreaks occur only if the moths are subsequently **reconcentrated** by **wind convergence**, such as is associated with rainstorms or topography. When wind convergence persists locally, moths flying into the vicinity are held there, mate, lay eggs and outbreaks may subsequently develop. Moths that are not concentrated by wind convergence will remain dispersed and subsequently produce scattered low-density populations in suitable vegetation in areas of rainfall.

The areas of strongest wind convergence occur at the gust fronts caused by the outflow from storms (Figure 18a) and outbreaks often develop after moths have been concentrated at the edge of rainstorms. The importance of gust fronts caused by winds from storm outflows in the concentration of moths has been confirmed by radar observations and by many studies using meteorological and biogeographical techniques. Outbreaks are found predominantly on the edge of the storm towards which the prevailing wind is blowing (the windward

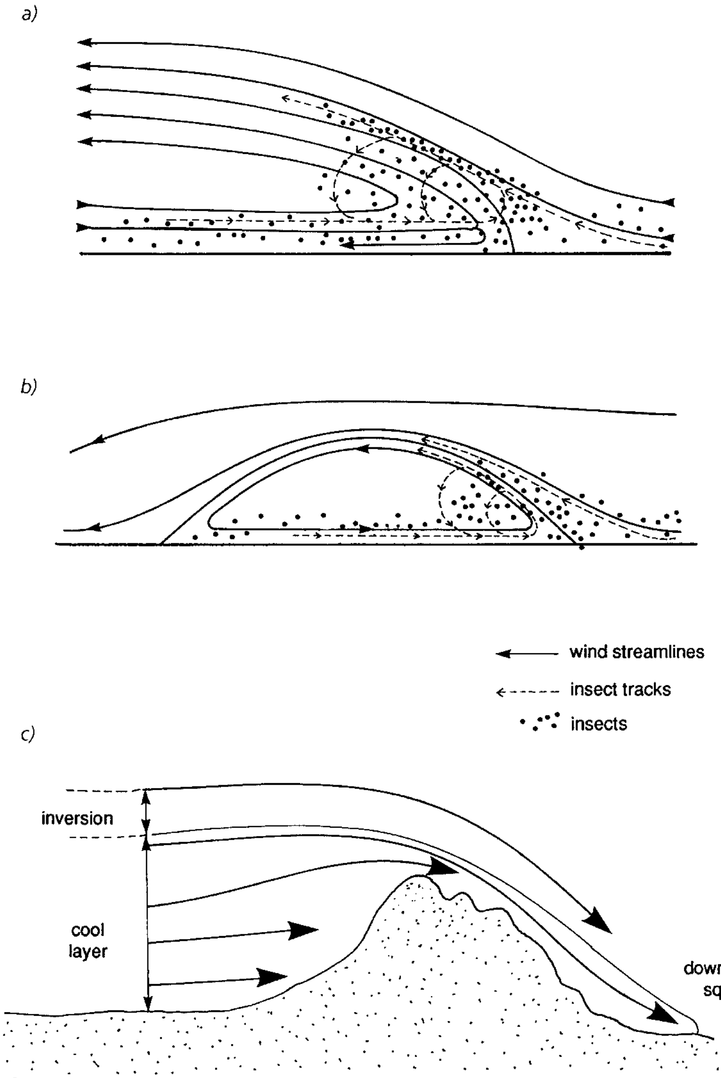


Figure 18. Schematic representation of how insects are concentrated at a gust front (after Pedgley, 1982).
(a) Gust front at a storm outflow
(b) Rotor in the lee of hill
(c) Katabatic (downslope drainage) winds

side), where a gust front is created between the prevailing wind and the storm outflow (Figure 19 and Plate 22). Intense convective rainstorms typically last for 1–2 h, so flying moths caught up in them might be expected to be concentrated 10–20-fold. If these moths landed before being re-dispersed, the increase in area (as distinct from volume) density would be 100–400-fold.

Downdraughts, and the resulting gust fronts from storms, can be particularly strong in the dry air at the start of the wet season. Storms at that time are more likely to occur over high ground than over the nearby plains (as in the example shown in Figure 19). Outbreaks at the start of the wet season in East Africa, therefore, tend to occur on the eastern side of high ground, as moths flying in the dominant easterly winds are carried into the storm-induced lines of wind convergence and are concentrated at the gust fronts. Many meteorological studies using wind trajectories (backtracking) have indicated that the main sources of moths causing initial outbreaks are between these outbreak sites and the coast.

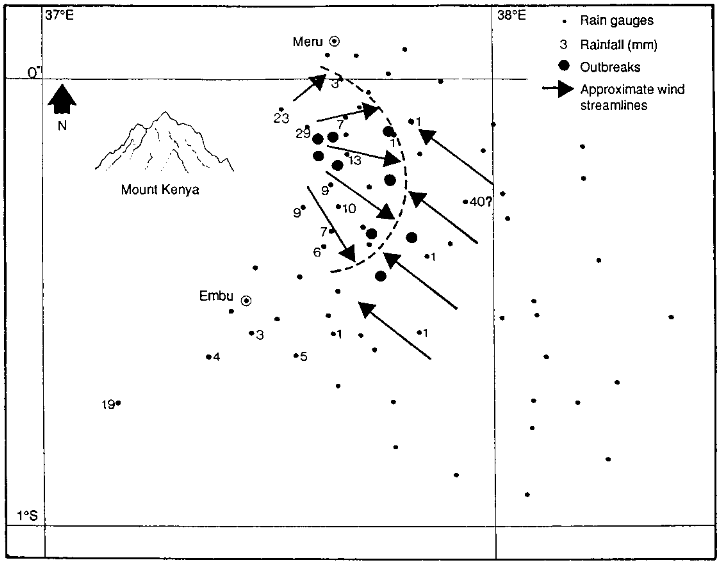


Figure 19. Locations of outbreaks of *Spodoptera exempta* larvae around the windward side of a storm (after Pedgley *et al*., 1982).

The importance of rainstorms for concentrating flying moths may well be enhanced by the moths' behaviour, as they have been observed to descend to the ground when it rains.

Lines of wind convergence also form in large-scale weather systems, but the rate of convergence is not as great as at a storm outflow gust front. When wind convergence at this scale persists in an area, sometimes for several days, moths flying in the locality are held there and outbreaks may develop. Figure 20 shows an example of a persistent, large-scale system of wind convergence within which moths were trapped, giving rise to outbreaks in the next generation.

At the coast, winds blow onshore during the day and die out, or are even reversed, during the night. The leading edge of the onshore wind called the sea breeze can be a well-defined convergence line similar to that of a gust front and is known as the sea breeze front. A similar feature forms around the shore of Lake Victoria. In conditions of prolonged sunshine the sea breeze or lake breeze will form regularly in the same area every day and moths flying in the first part of the night (see Sections 3.2.1 and 3.2.2) could be held in the coastal zone.

High ground acts as a barrier to the airflow and winds blow over and around hills. As a result, a complex pattern of eddies such as rotors form within the large-scale wind system (Figures 18b and 21a). In a region where the wind is predominantly from one direction, wind convergence induced by hills can be sufficiently strong and persistent to lead to moth concentration and, subsequently, armyworm outbreaks.

Also in highland areas, night-time down-slope drainage winds called katabatic winds (Figure 21), regularly occur in certain valleys and may bring moths down to the foothills. Where these valleys open towards the prevailing (broad-scale) wind, a squall line similar to a gust front is formed at the leading edge of the katabatic wind (Figures 18c and 21b).

Moth flight is inhibited in temperatures of less than about 13 °C, such as the moths would experience if taken aloft by the updraughts that necessarily accompany convergent winds. As night flying insects tend

(a)

(b)

(c)

(d)

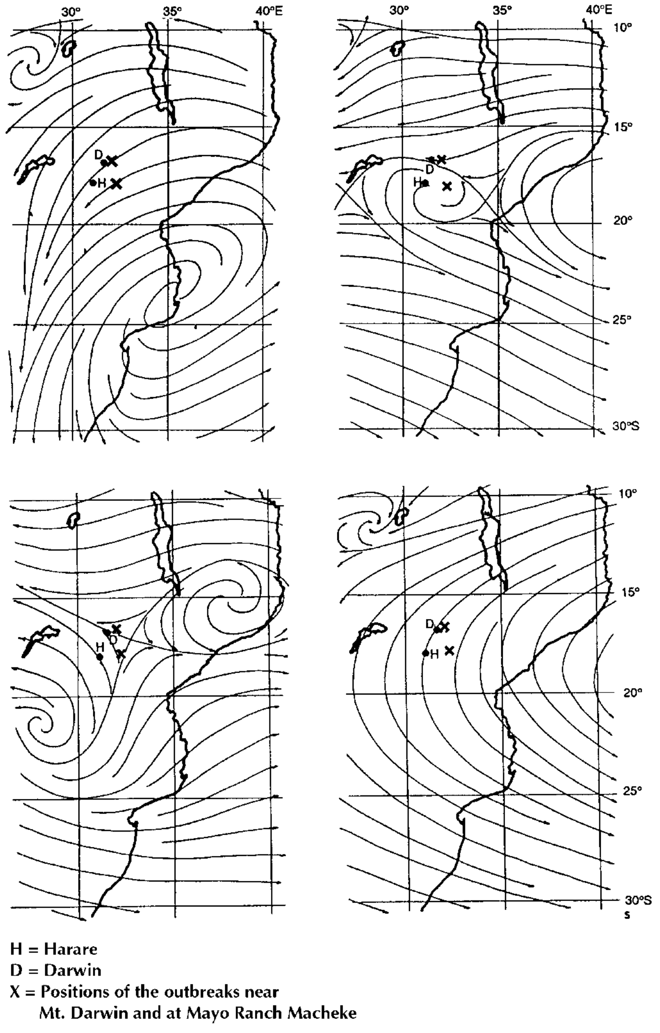


Figure 20. Synoptic charts showing position of outbreaks of *Spodoptera exempta* larvae in relation to low level wind streamlines, confluence and convergence in Zimbabwe (after Rose and Law, 1976).
(a) 18 October 1955
(b) 19 October 1955
(c) 20 October 1955
(d) 21 October 1955

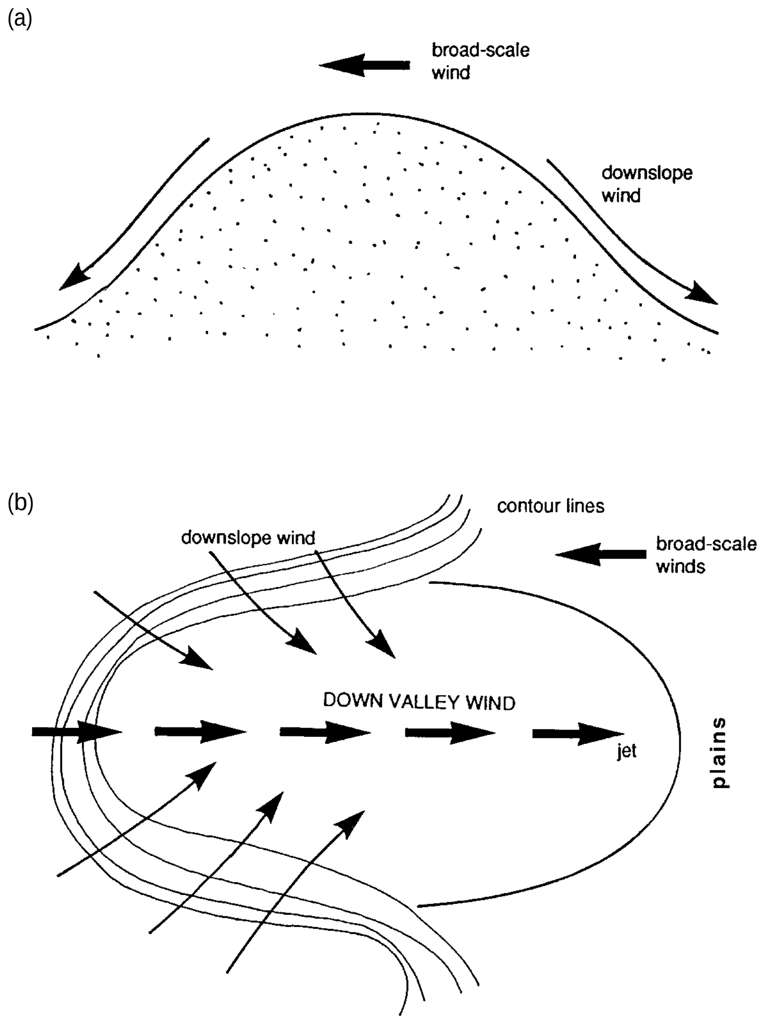


Figure 21. Winds associated with topography (after Pedgley, 1982).
(a) Cross-section of windflow over a hill
(b) Plan view of drainage winds down a valley

to fly in the warmest air, usually at the bottom of the temperature inversion* (Figure 18c) and they may actively avoid being carried upwards, when this inversion is broken by convection, by descending. During the months of the outbreak season, temperatures at ground level are rarely low enough to inhibit moth flight, even in the Kenya highlands.

4.1.2 Seasonal rainfall and outbreaks

After the dry season, outbreaks can occur only where there has been sufficient rain to allow the growth of grasses to provide food for the developing larvae. It is the first rains of the short wet season in some countries, particularly in Tanzania and Kenya, which are of the greatest importance in the initiation and spread of armyworm outbreaks within the region. These rains provide suitable conditions for the development of the first outbreaks, from which successive outbreaks spread.

The wet seasons, and the storms associated with them which are most likely to cause moth concentration in each country in eastern Africa, are related to the position of the ITCZ (e.g. see Plate 23). The ITCZ is the meeting place of the Northern Hemisphere north-easterly trade winds and the Southern Hemisphere south-easterly trade winds. The latter, during the Northern Hemisphere summer, are deflected to become the south-westerly monsoon.

The ITCZ moves seasonally northwards and southwards following the annual movement of the sun, with a lag of about 2 months. During the Northern Hemisphere summer, from July to September, the ITCZ is usually over northern Sudan and the south-western Arabian Peninsula. It moves southwards through Eritrea, Ethiopia and Somalia during October, across Kenya during November, reaching Tanzania by December and as far south as northern Mozambique and Zimbabwe in January. During February and March it starts moving north through Tanzania and crosses Kenya in April and May. During May and June the ITCZ crosses southern Sudan, Ethiopia, Eritrea and Somalia,

*Temperature inversion – the increase of temperature with altitude which is typical of calm conditions at the surface during the times of *S. exempta* moth flight and contrasts with normal conditions when temperature decreases with altitude.

reaching northern Sudan and the south-western Arabian peninsula again by July. In some years, the progress of seasonal changes of wind and rain may differ slightly from the average.

The southward movement of the ITCZ is associated with the ‘short rains’ in southern Ethiopia, Somalia, Kenya and northern Tanzania, and the northward movement is associated with the ‘long rains’ in those countries. Zones at the extremes of the traverse of the ITCZ generally have only a single wet season.

The usual pattern of winds and rain may be interrupted for spells of several days by other meso-scale weather systems. Incursions of the African Rift Convergence Zone (ARCZ) (also known as the Congo Air Boundary, CAB) frequently bring westerly winds from the Congo across eastern Africa. Sometimes tropical cyclones in the Indian Ocean move near the African coast or into the Mozambique Channel. Storms and wind convergence are associated with both of these weather systems and may have a major effect on the distribution of armyworm moths.

Key references: Betts (1976)
Pedgley *et al*. (1982, 1989)
Riley *et al*. (1981)
Rose and Law (1976)
Tucker (1983, 1984a, 1993)
Tucker *et al*. (1982, 1983, 1984b)

4.2 Population dynamics

4.2.1 Armyworm population increase and survival

Major factors affecting the rate of increase (R) of the population in any particular place are:

- the size of the initial moth population
- the rate of reproduction
- survival
- the strong migratory capability of the armyworm moth.

S. exempta is an '*r*-strategy' insect, having a short generation time and many offspring. Studies have shown that the finite rate of increase '*R*' under laboratory conditions is 142 on maize and 125 on *Cynodon* sp., whereas, on poor quality grasses, the increase may be as low as 15. A net reproductive rate (R_0) of 100 could easily be achieved for infestations feeding on new young grasses early in the season. At this rate, assuming 80% mortality (see Section 3.1.4) and each female moth laying 1000 eggs (see Section 3.1.2), there could be a 10 000-fold increase over two generations. Given the 1:1 sex ratio typical of this species in the field we see:

in generation 1, from **1** female:

$$\frac{1000 \text{ eggs} \times 20}{100} \text{ (\% survival)} = 200 \text{ moths}$$

(i.e. **100** females + **100** males)

which in generation 2, from those 100 females gives:

$$\frac{100 \times 1000 \text{ eggs} \times 20}{100} \text{ (\% survival)} = 20\,000 \text{ moths}$$

(**10 000** females + **10 000** males)

The strategy of rapid development, high reproductive capacity and migration is the adaptation that enables *S. exempta* to survive in ephemeral and often marginal grasslands where seasonal drought conditions prevail.

During the dry season, grasses dry out in the majority of areas in Africa and become unsuitable for larval survival. Low-density populations of *S. exempta,* however, may remain in the localized areas of green grassland where rain falls for much of the year. In these places numbers of larvae remain low and their cryptic coloration makes it difficult for parasitoids and predators to find them. It is still likely that mortality is high at this time as grasses mature and become less palatable. Nevertheless, these low-density populations provide a reservoir of moths available to initiate outbreaks through population build-up when the rains return. They are referred to as **source area** populations. Low-density populations, as indicated by moth trap catches in eastern and central Africa, seem to persist in places such as Malawi, possibly western Uganda and south-western Ethiopia, as well as in the coastal areas and the highlands of Kenya and Tanzania, and possibly the shoreline of Lake Victoria. Populations may also persist throughout the year in other countries that normally have a dry season, if food plants remain palatable. Studies at IRLCO-CSA may provide new information on the seasonal population distribution of *S. exempta* in central and southern Africa.

Studies in the coastal region of Kenya during 3 consecutive years demonstrated that *S. exempta* moth populations persist there throughout the year. These moth populations reached peak numbers 2 months after the beginning of each of the two annual wet seasons, indicating that breeding is localized on the coast. This contrasts with the normal outbreak situation when peaks in moth numbers occur with the onset of the rains as the moths are concentrated inland. The study also demonstrated the mobile nature of the populations that utilize the locally available rains within the coastal source area of Kenya.

Many meteorological studies using wind trajectories (backtracking) have indicated that the sources of moths causing the initial outbreaks are between the outbreak sites and the coast. Even so, source areas in the highlands cannot be ruled out, as katabatic winds (night-time down-slope drainage winds) may bring moths down to the foothills (see Section 4.1.1) into the areas where the first outbreaks are most often recorded (see Section 4.2.2).

During the dry season (July–October), the highlands of eastern Africa are, in general, cold and, therefore, the development period for *S. exempta* is prolonged. This precludes any major build-up of populations in these highland areas. As temperatures rise everywhere at the end of the cool season, armyworm development rates increase, leading to more synchronous moth emergence throughout the region at the onset of the warmer weather. This phenomenon is known also for other grassland insects in Africa. *S. exempta* is well adapted, therefore, to survive in the solitary phase at low population densities, and survival is achieved by dispersive migratory flight within each generation (see Section 3.2.2). Concentration of flying moths by wind convergence induced by weather systems and/or topography (see

Section 4.1.1) during insect migration results in high-density outbreak populations during the seasonal rains. Due to the mobile, ephemeral nature and scattered distribution of rainstorms, the concentration of moths by wind in any particular place is largely fortuitous and the number of moths being concentrated is related to the size and proximity of the moth source upwind.

Key references: Gatehouse (1986, 1987b)
Haggis (1996)
Merrett (1986a)
Nyirenda (1985a)
Persson (1981)
Rose (1975a, 1979a)
Yarro (1982)

4.2.2 Outbreak initiation and spread

When the rains arrive, the moths produced from widely scattered low-density populations in the source areas are concentrated by the winds and outbreaks develop. These are called **primary** outbreaks. They are usually small in area, widely scattered with very variable larval densities, and as a consequence often remain undetected.

The synchronized and often massive emergence of moths from outbreaks results in the subsequent spread of outbreaks, as the moths emigrate downwind.

The successful development of primary outbreaks is enhanced by:

- preceding drought
- suitable storms for the concentration of moths
- the flush of young grasses
- rainless and sunny periods after moth concentration, to minimize stress and reduce disease such as virus during the larval stages.

The first flush of new grasses after rainstorms following drought have been shown to contain higher levels of nitrogen than the grasses growing later in the wet season. When larval host plants contained higher levels of nitrogen, the fecundity of the next generation moths was increased. It has, however, not yet been demonstrated that the survival of larvae is enhanced and their development more rapid under these conditions.

In eastern Africa, areas in which primary outbreaks most often occur are shown in Figure 22. A more detailed map showing the actual locations of first reported outbreaks in Kenya between 1961 and 1992 is given in Figure 23. Some of these reports may not necessarily have been primary outbreaks (which may not have been detected), but were the first reported outbreaks. Typically they occur in areas of low and erratic rainfall. The most important primary outbreaks are those occurring inland from the coast, east of the first areas of high ground (Plates 24 and 25).

It is in these areas of Kenya and Tanzania (Figure 22), countries with two wet seasons a year, that the first outbreaks usually occur, and it is these first outbreaks which initiate the onset and spread of subsequent **secondary** outbreaks throughout eastern Africa and as far north as the Yemen.

Primary outbreaks derived from low-density populations may also occasionally develop later in the season, especially in western Kenya and north-western Tanzania. Such outbreaks are difficult to separate from contemporary secondary outbreaks where generations have overlapped, except by retrospective analysis.

A primary outbreak is an outbreak derived from low-density populations.

A secondary outbreak is an outbreak derived from earlier outbreaks.

Secondary outbreaks can be recognized by their location near where rainstorms occur downwind from previous outbreaks at the time when the moths are known to be emerging and emigrating. Dates of moth emergence may be estimated from larval head capsule width measurement (see Section 3.1.4, Figure 33 and Table 4), from samples of larvae collected in outbreaks (see Section 5.2.5), by reference to life tables (Table 6) and by extrapolation from records of moth catches from trap networks.

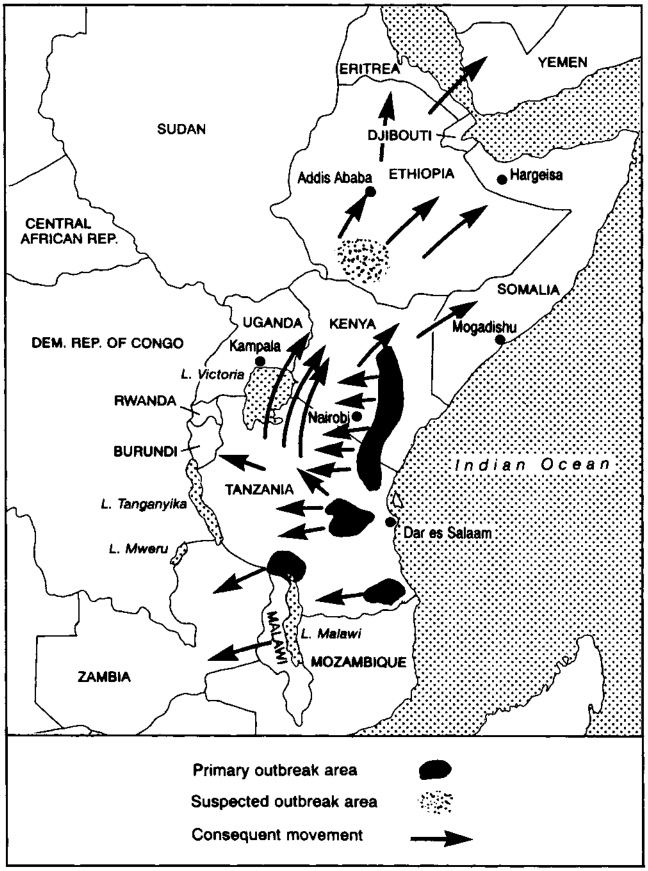


Figure 22. Known primary outbreak areas of *Spodoptera exempta* in eastern Africa and typical movements from these areas during a major outbreak season.

For primary outbreaks to initiate secondary outbreaks and an upsurge to develop, the weather needs to be suitable for larvae to survive, to complete their development and to burrow successfully into the soil for pupation to take place (see Section 3.1.4). Moth emergence must also be successful. Rainstorms are also necessary downwind of the emergence site for those moths to become reconcentrated by the

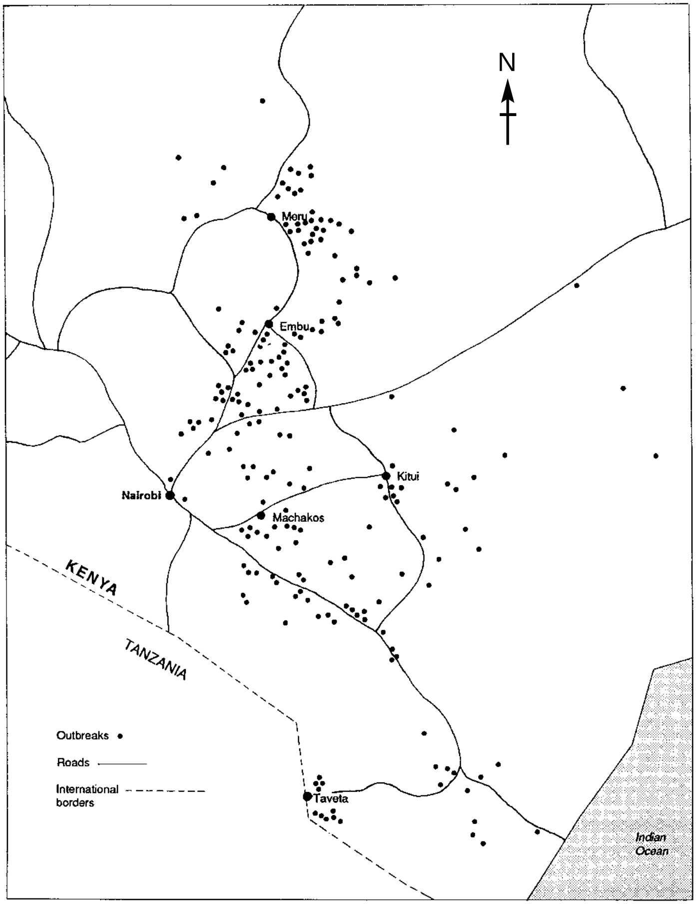


Figure 23. Locations of first recorded *Spodoptera exempta* outbreaks of the season in Kenya between 1961 and 1990.

associated wind convergence (see Sections 3.2.2 and 4.1.1). A major upsurge and spread of outbreaks may occur when there are sporadic rainstorms with long periods of sunshine during the outbreak period. Such weather often occurs during the short rains season in November–December in East Africa.

Conversely, prolonged cool, cloudy and wet weather will suppress any subsequent upsurge and spread of outbreaks. For this reason, primary outbreaks do not usually begin in the wetter parts of the region in the west, and the success of outbreaks in the western parts of eastern Africa is reduced, except under unusually dry conditions.

Field studies have shown that the females from moth populations which develop in places of low sporadic rainfall have the potential to migrate further than those from wetter areas, and thus may increase the spread of armyworm outbreaks.

Case studies of the distribution of rainfall in 5 years in Kenya suggested that the severity and timing of the first reported outbreaks of the season are commensurate with the size and number of available habitats where populations could carry-over through the dry season.

The spread of armyworm outbreaks is largely predictable from knowledge of the seasonally dominant winds of the area and from meteorological observations on the times and places where rainstorms occur during periods of mass moth flight.

Key references: Gatehouse (1986)
Haggis (1996)
Janssen (1993c, 1994)
Pedgley *et al*. (1989)
Tucker (1983, 1984b)
Tucker *et al*. (1982, 1983)
Wilson and Gatehouse (1993)

4.2.3 Seasonal migrations and variations

The typical pattern of incidence of armyworm outbreaks is well documented for Tanzania, Kenya, Uganda, Ethiopia, Eritrea, Somalia and Yemen (see Sections 2.3 and 4.1.2 and also Figures 4 and 24). For example, the progressive appearance of outbreaks further west in East Africa from November to March is because the winds that carry the moths are predominantly easterly. Moths from Tanzania may be taken as far west as Burundi and Rwanda before the start of the more northward movements (see Figure 24a).

Although outbreaks usually begin in Tanzania during late November or December, they sometimes begin earlier, in Kenya or Somalia (Figures 24c and 24d). Moths from these outbreaks may invade Tanzania prior to the westward and northward spread.

(a)

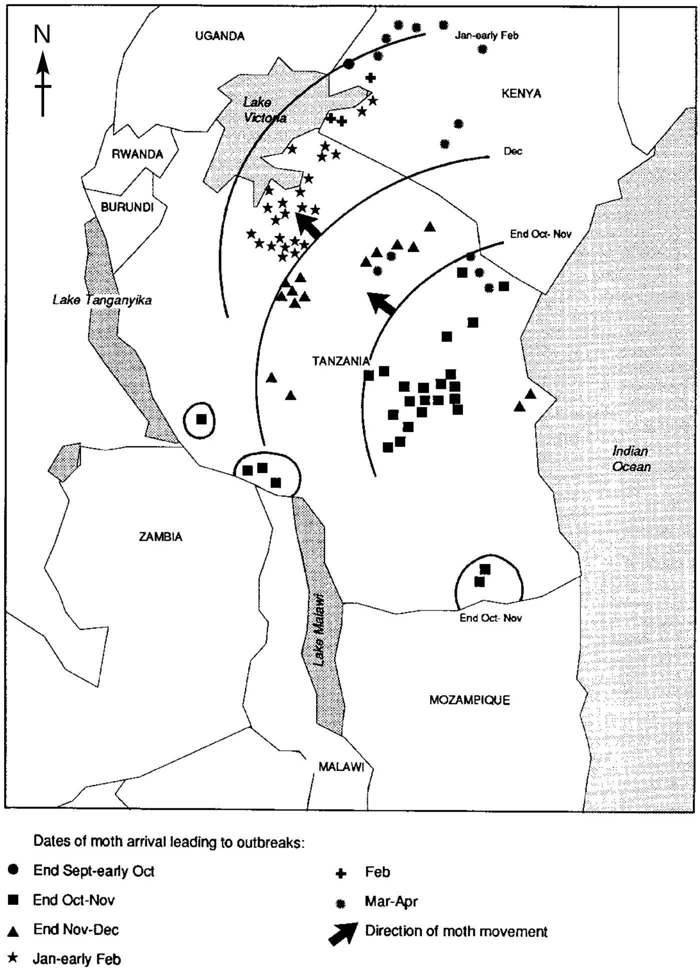


Figure 24. Generalized movements showing how *Spodoptera exempta* outbreaks spread in eastern Africa, from **source areas** in:
(a) Tanzania (December 1989).

(b)

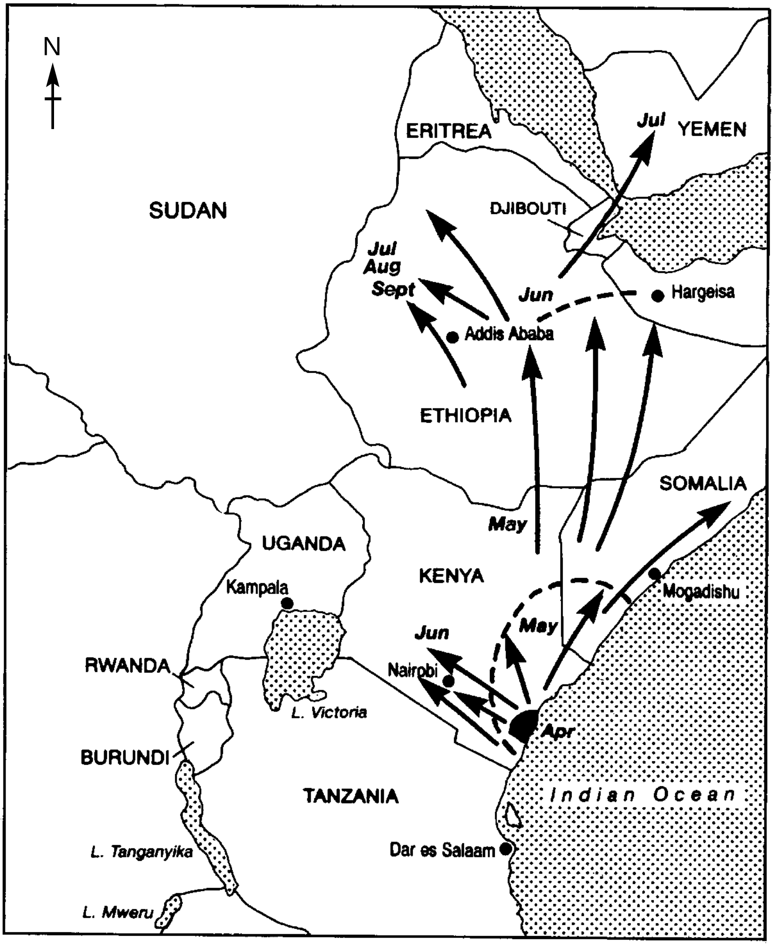


Figure 24 *cont*. Generalized movements showing how *Spodoptera exempta* outbreaks spread in eastern Africa, from **source areas** in:
(b) Kenya (April 1984).

(c)

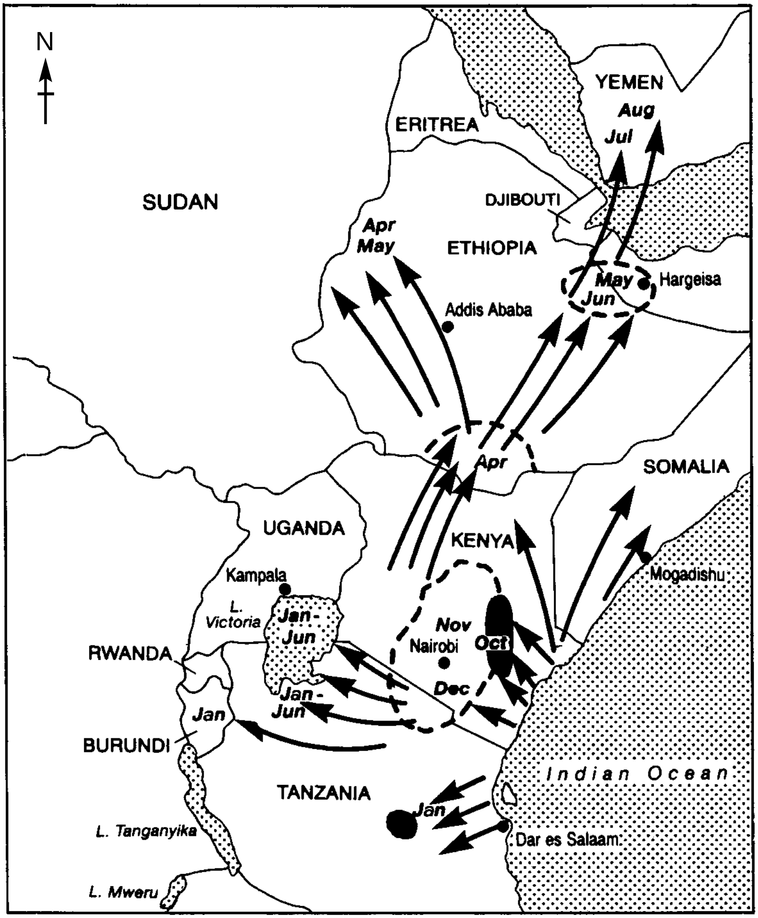


Figure 24 ***cont***. Generalized movements showing how *Spodoptera exempta* outbreaks spread in eastern Africa, from **source areas** in:
(c) Kenya (October 1984).

(d)

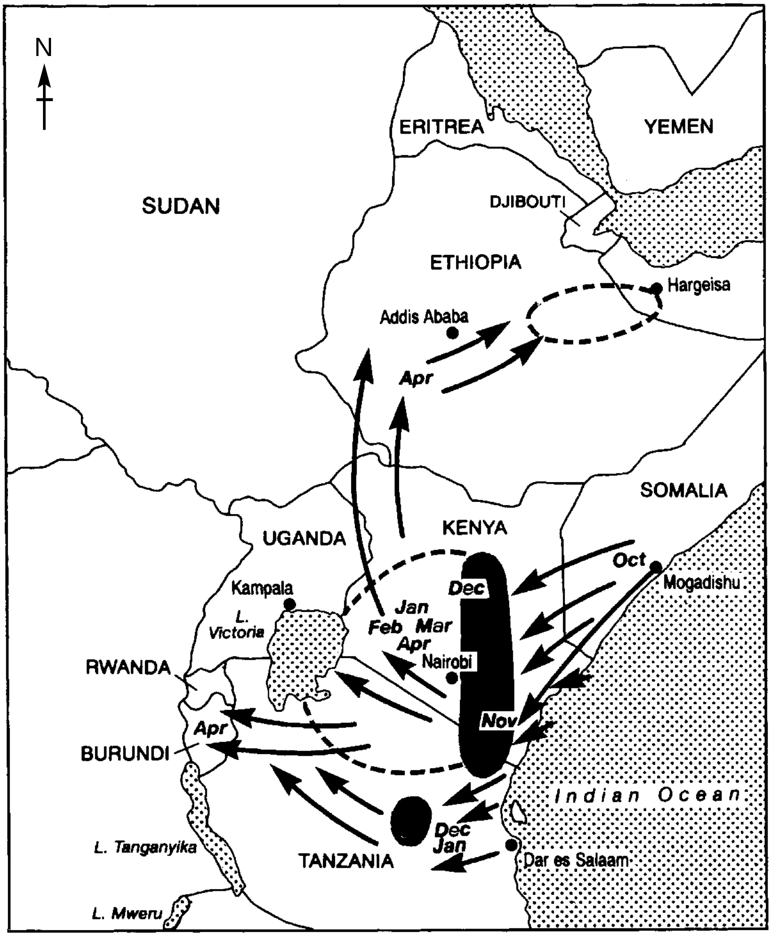


Figure 24 *cont.* Generalized movements showing how *Spodoptera exempta* outbreaks spread in eastern Africa, from **source areas** in:
(d) Somalia (October 1985).

The size and success of the early outbreaks of the season influence the size and extent of the subsequent spread of outbreaks throughout eastern Africa as far north as the Yemen, although their severity may be checked by prolonged rainfall or, conversely, increased by less rainfall and more sunshine.

Low rainfall during the short wet season, particularly during November–December in Kenya and Tanzania, increases the probability of occurrence of primary outbreaks with high survival rates in the following outbreak season.

The southwards spread of outbreaks through southern Africa seems to originate from southern Tanzania, northern Mozambique and Malawi, with moths from these areas invading Zimbabwe, Swaziland and South Africa in a south-westward direction. Moth invasions into Zambia and Botswana are less frequent (Figure 25), although they were extensive in both countries during the 1992/93 season.

There is little evidence that moths produced from outbreaks at the extremes of the geographical range make return migrations southwards from the Arabian Peninsula or northwards from South Africa. It appears that, in some years, moths produced in September–October in southern Ethiopia or Malawi may move respectively southwards or northwards and contribute to the production of primary outbreaks in Kenya and Tanzania, but the main moth source areas are within those countries (see Section 4.2.2).

It has often been suggested that there are cycles in the abundance of armyworm moths and outbreaks, but no clear pattern is obvious (see Section 2.4). The cycles are approximately monthly – equivalent to the generation time, at 4 years and at 10 or 11 years. A 4–5-year cycle might be linked to the El Niño Southern Oscillation (ENSO) cycle, which is the occurrence of high sea surface temperatures off the coast of Peru in the eastern Pacific and has far reaching effects on rainfall patterns within the tropics and possibly throughout the world. A 10–11-year cycle corresponds in length to the sunspot cycle, but the timing of this phenomenon relative to the severity of armyworm outbreaks has not been investigated.

The sources of moths that cause the first outbreaks are believed to be low-density populations of solitary phase larvae. Some of the circumstances which encourage these initial outbreaks are: preceding drought, suitable topography, storm-associated winds for reconcentrating moths, a flush of new food plants, and prolonged periods of sunshine and sufficient rain during the growth period of the larvae. These circumstances regularly occur in the most important primary outbreak areas.

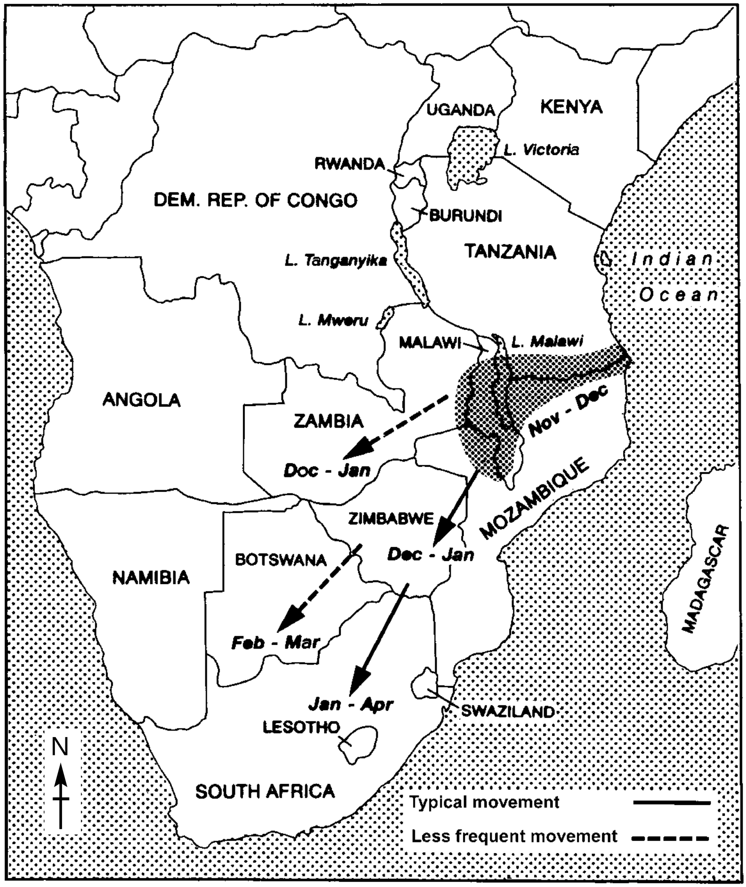


Figure 25. Main suspected source area and generalized spread of *Spodoptera exempta* outbreaks in central and southern Africa.

Key references: Brown *et al*. (1969)
Brown and Swaine (1965b)
Den Boer (1978a)
Haggis (1984, 1996)
Khasimuddin (1977b)
Odiyo (1981, 1984)
Page and Dewhurst (1992)
Pedgley *et al*. (1982, 1989)
Riley *et al*. (1981)
Rose (1975a)
Rose *et al*. (1985, 1987)
Tucker (1984a, 1984b, 1993)
Tucker *et al*. (1982, 1983)

5

MANAGEMENT

5.1 Strategies

5.1.1 Regional co-operation

The migrant nature of armyworm moths and the spread of outbreaks from one country to another necessitate good co-operation between countries to enable farmers and agricultural departments to prepare themselves adequately and in time to take action to reduce losses caused by the larvae to cereals, crops and pasture. National armyworm co-ordinators in neighbouring countries should regularly exchange information on the current armyworm situation in their respective countries. This can be achieved more efficiently if regional organizations are responsible for co-ordinating the exchange of information. Awareness of the development and spread of armyworm outbreaks will then be available to all countries and control strategies can be planned accordingly.

Many national crop protection services, particularly those in eastern, central, southern Africa, and in the Yemen, have departments with special responsibilities for the control of migrant pests, including armyworm. Both DLCO-EA and IRLCO-CSA have regional responsibilities for the management of African armyworm as well as locusts and quelea birds.

Three regions, based on the known migratory patterns of *S. exempta,* are proposed here to improve co-ordination for monitoring, survey, control and training activities concerning armyworm. They are eastern Africa and the Yemen, central and southern Africa, and western Africa, and are illustrated in Figure 26. The countries of these three regions of Africa and the regional organizations currently within them are listed in Table 8.

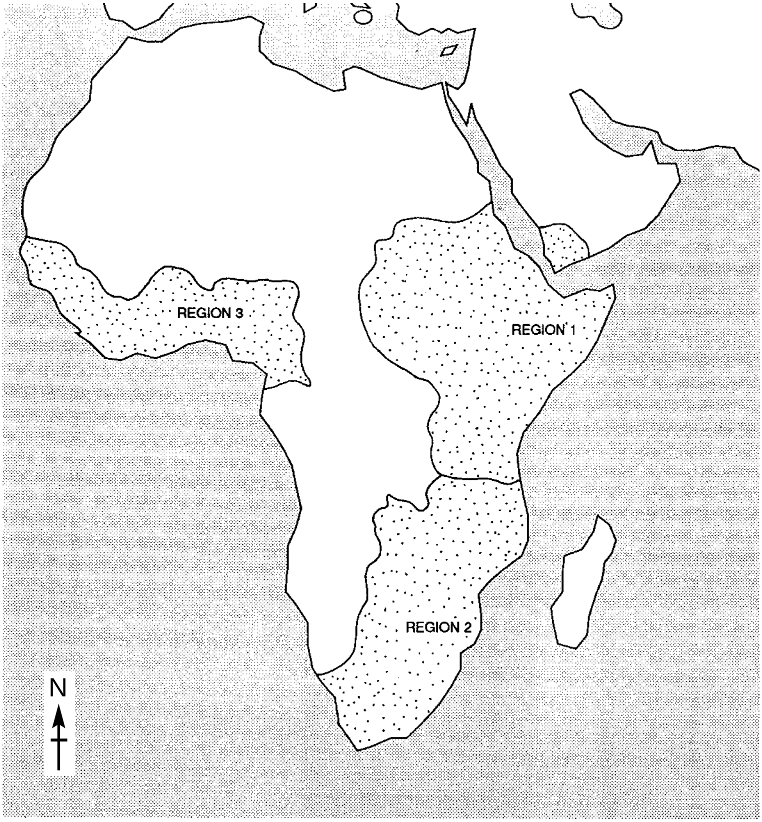


Figure 26. The three regions suggested for armyworm monitoring and control in Africa.

5.1.2 Direct measures to protect crops – crop protection

Farmers are concerned with the direct protection of their crops and pastures, as soon as these are found to be infested.

Criteria for control decisions and methods are discussed in Section 5.4. Heaviest losses to crops and pastures are caused during the last week of larval feeding and growth (see Section 3.1.4), so it is important for farmers to find and control the larvae rapidly, before serious damage

Table 8. Administrative regions suggested for co-ordinating armyworm management in Africa

Region	Countries	Current organization
1. Eastern Africa and Yemen	Burundi, Djibouti, Eritrea, Ethiopia, Kenya, Rwanda, Somalia, Sudan, (Tanzania), Uganda, Yemen	Desert Locust Control Organization for Eastern Africa (DLCO-EA)
2. Central and southern Africa	Angola, Botswana, Lesotho, Malawi, Mozambique, Namibia, South Africa, Swaziland, Tanzania, Zambia, Zimbabwe	International Red Locust Control Organisation for Central and Southern Africa (IRLCO-CSA)
3. Western Africa	Benin, Burkina Faso, Cameroon, Chad, Côte d'Ivoire, The Gambia, Ghana, Guinea, Guinea Bissau, Liberia, Mali, Niger, Nigeria, Senegal, Sierra Leone, Togo	There is no organization at present covering the whole of this region

Notes

Region 1. National Armyworm Co-ordinating Officers have been appointed to liaise with DLCO-EA in Djibouti,Ethiopia, Kenya, Somalia, Sudan, Tanzania and Uganda. The Yemen also has an armyworm monitoring service. National forecasting services are run only by Tanzania and Kenya.

Region 2. Monitoring services have been established in some countries.

Region 3. Armyworm monitoring services are undeveloped. The Organisation Commune de Lutte Antiacridienne et de Lutte Antiaviare (OCLA-LAV) covers only the Sahel countries.

begins. The amount of damage increases exponentially with each day that passes (see Figure 9). Rapid, well co-ordinated control is vital.

In some countries of eastern Africa, where governments have provided assistance to farmers, the presence of armyworm must be reported to the authorities. Insecticides are provided for control and national pest control units assist with the application. Where many hundreds or thousands of hectares are threatened, governments have the option to hire spray aircraft or to request spray aircraft from the regional organizations. Changing economic circumstances may in future affect the availability and extent of government funding to assist farmers with armyworm management.

5.1.3 Measures to reduce the spread of infestations – strategic control

A strategy aimed at reducing the spread of armyworm outbreaks has been developed by DLCO-EA and member countries in Region 1 (eastern Africa). Methods for monitoring and survey, forecasting and control are similar to those for direct crop protection, but aim to intensify all of these procedures at the times and places where primary outbreaks are likely to occur (see Section 4.2.2).

The purpose of strategic control is to eradicate as many larvae as possible early in the outbreak season, in order to reduce the number of moths subsequently available to initiate new outbreaks downwind. This will also reduce the chances of moths becoming reconcentrated and possibly causing critical outbreaks in areas of high agricultural potential.

The aim of controlling these primary outbreaks is to maximize the kill by destroying the largest, oldest and most dense outbreaks first, whether they are on crops or grasses (rangeland or pasture). Where resources are limited, the primary outbreaks selected for priority control should be the ones considered to be critical in space or time for the development of subsequent outbreaks

Primary outbreaks usually occur in recognized areas (see Section 4.2.2), on the windward slopes on the first high ground inland from the

Indian Ocean in eastern Africa (see Figure 22). Predominantly easterly winds blow from the coast towards these semi-arid inland areas at the time when primary outbreaks occur in these areas. These outbreaks often coincide with the first rainstorms of the short rains in Kenya (late October or December (Figure 23) or Tanzania (late November–January), when cereal crops will be planted.

Strategic control of armyworm requires that control operations are mounted against infestations of *S. exempta* larvae **even if they are of no immediate economic importance but when their progeny pose a potential threat to crop areas downwind**. Studies show that strategic control will be worthwhile, particularly during the period October–December. Ground control (see Section 5.4.3 *et seq*.) is also worthwhile on economic grounds in 20% of primary outbreak areas, regardless of season.

Outbreaks in Morogoro district of central Tanzania and Meru district of eastern Kenya can be sources of moths capable of causing economic damage elsewhere downwind of these localities. In eastern Africa strategic control operations should be targeted primarily at areas in northern and central Tanzania and east-central and south-east Kenya.

For example, in October 1984, primary outbreaks in Meru district, Kenya, were identified as being potentially critical and likely to lead to the subsequent spread of secondary outbreaks into agricultural areas (Figure 24c). This was recognized in time for sufficient insecticides and spray equipment to be organized and distributed to agricultural areas for control of the outbreaks that developed subsequently. Outbreaks in one other area were also identified as being potentially critical, but they did not result in the spread of outbreaks because there were no rainstorms downwind to reconcentrate the moths after they emerged and left the outbreak sites (see Sections 3.2.2 and 4.1.1). Five other areas of primary outbreaks were considered not to be critical because of their position in relation to anticipated moth dispersal. This example demonstrates clearly the need to evaluate the potential threat to agriculture of all primary outbreaks by relating their position to the prevailing winds. Evaluations such as these should be carried out by agricultural staff (National Armyworm Co-ordinator) and the National Armyworm Forecaster for every outbreak visited.

Occasionally, as in 1984, primary outbreaks occur in areas where it is recognized that the subsequent spread of outbreaks would be of little importance in terms of a large population increase. For example, subsequent movement to western areas that are wet and, therefore, where survival will be reduced, is unlikely to generate critical outbreaks.

Vigilance is particularly needed at the time the monsoon winds change direction, both from north-easterly to south-easterly during late March to April, and subsequently back to north-easterly during November. This is because moths may be blown towards dry regions where rains are expected to fall as a result of the influence of the ITCZ at the beginning of the rainy season (see Section 4.1.2). Fresh flushes of grasses developing with the rains, coupled with extensive periods of sunshine, favour high larval survival, thus increasing the potential for a rapid increase in the size of the moth population (see Section 4.2.1). Outbreaks occurring at these times must be regarded as being potentially critical in relation to the further spread and possible economic consequences of armyworm outbreaks. For this reason, every effort should be made to control them.

It is almost certain that effective control of selected outbreaks will reduce the spread of armyworm. Results from studies of the pattern of spread of infestations throughout eastern Africa over a period of 28 years indicate this. Typical patterns are depicted in Figure 24. As a general rule, the first two or three generations following a primary outbreak are distinct in their timing; after that generations begin to overlap and become difficult to distinguish.

Evidence that moths emerging from an outbreak are directly responsible for subsequent outbreaks downwind was obtained in experiments in which two outbreaks were ringed with pheromone traps. In one experiment, moths marked with a dye at an emergence site in central Kenya were trapped near the sites of 11 outbreaks which developed downwind in the next generation. On another occasion, also in Kenya, the direction of movement of unmarked male moths was shown to be towards areas where outbreaks in the next generation subsequently developed (from Emali to the Nguruman escarpment in 1983).

Operationally, it has become routine at DLCO-EA to map and predict the movements of moths from the initial outbreaks (see Section 5.3). The reliability of these procedures has been confirmed, and shows that as the spread can be forecast, it should also be preventable by timely control.

Weather is the major factor influencing outbreak severity. When rainfall is sporadic and poor, the early spread of outbreaks in Kenya and Tanzania is enhanced (see Section 4.2.3).

Once the armyworm outbreak season has begun, moths from outbreaks predominate, and those from *solitaria* populations appear to be of minor importance. This is partly because a greater proportion of *gregaria* phase moths are of long flight strains, but also because of the closely synchronized and massive emergence of moths from dense outbreaks (see Sections 3.1.3 and 3.2.1).

The conditions which result in the development of primary outbreaks from *solitaria* populations are seldom found during the wet season. Exceptions may occur, however, when rain falls after an extended dry spell in the wet season, and may also occur in young, late-planted cereal crops when the surrounding grasses have become rank and unpalatable.

Key references: Cheke and Tucker (1995)
Haggis (1996)
Jackson (1979)
Pedgley *et al*. (1989)
Robinson (1991)
Rose (1978a)
Rose *et al*. (1985, 1987)
Tucker (1993)
Wilson and Gatehouse (1992, 1993)

5.2 Monitoring and survey

5.2.1 Traps

The most effective way of monitoring armyworm populations is through networks of moth traps in each country. Information on numbers of moths caught forms the basis from which warnings of invasion and subsequent outbreaks are produced. This information is also needed for understanding and predicting movements of moth populations within and between countries.

Two basic types of trap are in regular use for monitoring *S. exempta* moths, namely light traps and pheromone traps. Pheromone traps are recommended for widespread use in national networks, with light traps restricted to use at research centres that have trained staff for sorting and identifying the catches, and where electricity is available.

Light traps have been used in many locations, principally at research stations, since the 1960s. They catch many different kinds of insects that fly near to the trap, often in large numbers during the wet seasons. All insects caught in light traps have to be sorted and the required species have to be identified. A typical design is shown in Figure 27. Catches are substantially reduced in the absence of a killing agent, presumably because the trapped adults are able to escape. Bright moonlight and the position of the trap site in relation to buildings, lights and trees also affect its trapping efficiency. The catchment area for a light trap is known to vary with environmental conditions.

Light traps catch immature migrating moths and mature and mated adults, both males and females equally, sometimes in large numbers. Males usually predominate in the trap catches when females begin oviposition in the area. Mated females may be recognized by the presence of spermatophore(s) within the bursa copulatrix (see Section 3.2.4).

Light traps require a source of electricity, spare parts are expensive and access to foreign exchange for purchases may be required. Trained staff are needed to identify the pest species from the often large insect samples caught (particularly during the wet season).

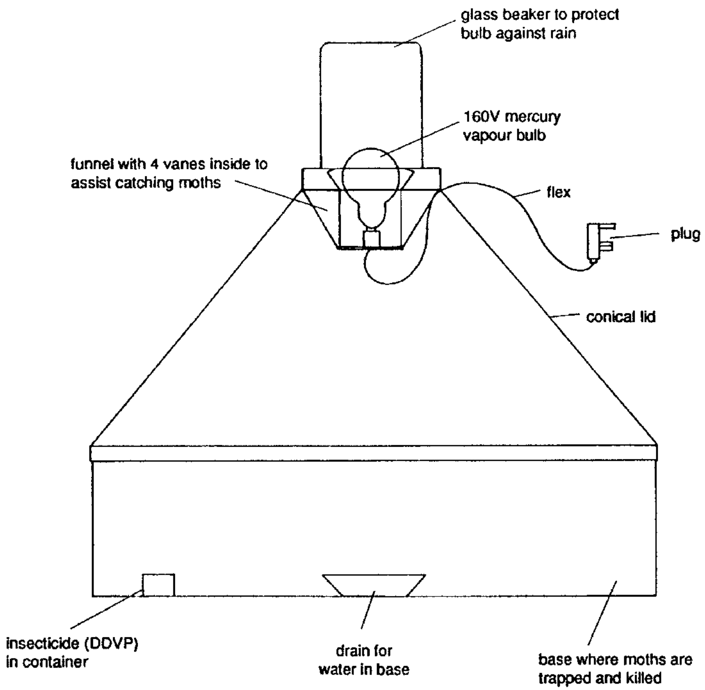


Figure 27. The design of a light trap (after Robinson and Robinson, 1950).

Pheromone traps utilize artificial sex attractants (pheromones) which are species specific. Only sexually receptive males are attracted by the pheromone. Once they have detected the pheromone molecules in the air they orientate themselves to fly upwind describing a zigzag flight path to intercept the odour plume repeatedly, its strength increasing as they approach the pheromone source, i.e. the trap. Evidence of this upwind movement can be simply demonstrated by the distribution of moths on a sticky disc trap: all the male moths caught on a night in which the wind direction is constant will be found on the downwind side of the lure.

As with light traps, the area over which the pheromone trap is effective varies with environmental conditions, particularly with wind speed and trap site, but it is thought to be within at least a 100 m radius.

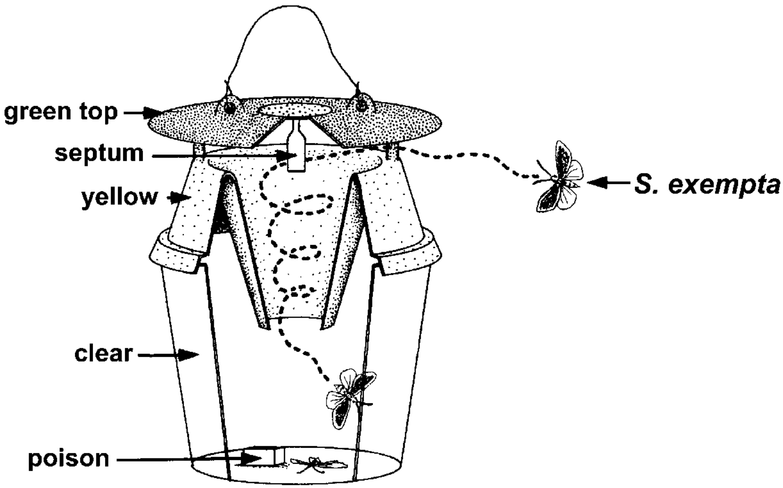


Figure 28. A pheromone trap (funnel type UNI-trap).

The standard armyworm pheromone trap now recommended, the UNI-Trap, is shown in Figure 28. It can be purchased commercially (see Appendix 5) and it is cheap to operate. This funnel trap has been shown to be six times more efficient at trapping moths than other designs tested. It is made from a 10 cm diameter yellow plastic funnel with a green plastic top attached 3 cm above the funnel rim. This type of pheromone trap is cheap to buy and easy to operate requiring only minimal training for the operator. The synthetic sex pheromone impregnated into the septum (**lure**) is suspended 1 cm below the centre of the top of the trap (see Figure 28). Male moths enter the funnel and are caught in a collecting container attached to the base of the funnel. The container can be made of any material, even a strong plastic bag. Ideally the container should be clear (to aid observation) and have a number of small drainage holes to ensure that rainwater can escape. A killing agent is also required such as ‘Nogos’ (dichlorvos) impregnated into a plastic strip (‘Vapona’), or ‘Baygon’(propoxur) dust. Catches are substantially reduced in the absence of a killing agent, presumably because the trapped adult moths are able to escape back up through the funnel.

Pheromone lures have an effective field life of about 3 months (this figure is used for armyworm moth monitoring purposes although the active life of the lure is really much longer). **Used lures should always be destroyed and not thrown away at the site of the trap** because the residual pheromone in them will distract male moths when they approach the trap, disorientating them and thus reducing the effectiveness of the trapping system.

As pheromone traps catch only sexually receptive males, large numbers of male moths in a pheromone trap imply that mature females are present in the vicinity, unless, as has been observed occasionally, the moths are on migration.

Despite the uncertainty of the true size of the trap catchment area, high numbers of *S. exempta* in light traps and pheromone traps have been shown to be reliable indicators of the presence of large moth populations in areas up to 100 km of the trap site and within which outbreaks are particularly likely to occur.

NB. If more than 30 males are caught in one night in a pheromone trap, farmers should be alerted to the possibility of an outbreak in the area. When high numbers are caught over 3–5 nights, females are almost certain to be laying eggs in the area, and outbreaks are likely to follow.

How many moths constitute a high catch in a light trap varies widely from one location to another (see Section 5.2.4).

Key references: Betts and Odiyo (1968)
Bowden (1973a)
Murlis *et al*. (1986)
Robinson and Robinson (1950)

5.2.2 Pheromone lures

Pheromone structure. Two components of the female *S. exempta* sex pheromone were identified in 1975. These were (*Z*)-9-tetradecenyl acetate (Z9-14:Ac) and (*Z,E*)-9,12-tetradecadienyl acetate (ZE9,12-

14:Ac) found at a ratio of 100 : 5. A synthetic mixture based on these two components was found to be highly attractive in the field.

Further extensive re-examination identified four more compounds:

(i) (*Z*)-9-tetradecenal (Z9-14:Ald)
(ii) (*Z*)-11-tetradecenyl acetate (Z11-14:Ac)
(iii) (*Z*)-9-tetradecen-1-o1 (Z9-14:OH)
(iv) (*Z*)-11-hexadecenyl acetate (Z11-16:Ac).

Each component constituted less than 5% of the major component, Z9-14:Ac.

Field tests of these components in Kenya showed that Z9-14:OH reduced the synthetic pheromone's attractiveness to male moths. Z9-14:Ald and Z11-14:Ac had no effect on trap catch, but the addition of Z11-16:Ac increased catches significantly.

The present synthetic attractant used in the regional monitoring system is composed of a binary mixture of Z9-14:Ac and ZE9, 12-14:Ac in the ratio of 100 : 7.5. The quantity of ZE9,12-14:Ac has been increased by 50% over the natural pheromone to compensate for material lost by degradation in the field due to sunlight. Oxidation can also degrade the pheromone components in the high temperatures experienced in the field. In order to combat this, the antioxidant butylated hydroxytoluene (BHT) is added to the lure. This compound has no effect on the attractiveness of the synthetic lure.

The pheromones produced by female moths collected in Kenya, Tanzania and Malawi have been examined and found to contain essentially the same pheromone mixture. The sex pheromone produced by *S. exempta* moths reared from *gregaria* and *solitaria* larvae have also been shown to be essentially the same, in both quantity and composition.

The pheromone blend is currently formulated and marketed by NRI, Chatham, UK. Rubber septa are loaded with pheromone blend and distributed regionally by the DLCO-EA, Nairobi, Kenya. Blank septa may also be purchased commercially (see Appendix 5).

Field use. Although the pheromone lure (septum loaded with pheromone) specifically attracts male *S. exempta* moths, other insects are sometimes found in the traps. An early formulation of *S. exempta* pheromone occasionally attracted *S. triturata*, but this does not now happen. If there is doubt about the identity of the moths caught, sample catches must be retained and forwarded to the national armyworm co-ordinator for detailed examination. There is no indication that *S. cilium*, *S. littoralis* or *S. exigua* are attracted by the *S. exempta* pheromone.

For the synthetic pheromone to be useful in the field it is necessary for the component compounds to be released in a controlled manner over a long period of time. The physical characteristics of the dispenser provide the necessary environment for this to take place. At present, small (19 mm long by 8 mm outside diameter) rubber septa are used for this purpose. Lures are prepared by applying a solution of the synthetic pheromone inside the narrow neck of the septum and allowing the solvent to evaporate. The pheromone is retained on the rubber and slowly diffuses from its surface at a controlled rate. The release rate from a typical rubber septum, containing 5 mg of pheromone, in a wind tunnel at 28 ^{o}C in 8 kph wind speed, is shown in Figure 29.

Half of the pheromone is released in approximately 26 days. However, because release rate is proportional to loading, even after 90 days approximately 10% of the original pheromone loading remains. Septa loaded with 2 mg of pheromone that had been exposed under field conditions in Kenya for 1 or 3 months were found to contain respectively 84% and 56% of their original loading, when subsequently analysed. Thus the lures presently supplied for the monitoring system, which contain 4 mg of pheromone, should have a useful field life in excess of 3 months.

The pheromone release rate is increased by increases in temperature and wind speed, therefore, the highest release rate occurs during daylight hours. If the lures are not stored under suitable conditions prior to their installation in the field, then their effective life will be dramatically shortened. For this reason they should not be used in the

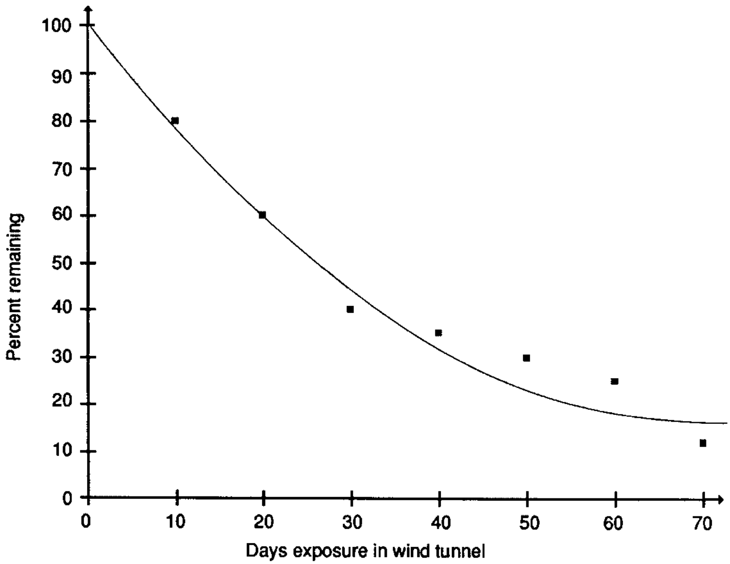


Figure 29. Release rate of pheromone from a typical rubber septum.

field for more than 3 months, even though residual activity may be apparent. Ideally, prior to use, they should be stored at <5 °C, in a refrigerator. Alternatively they can be stored in sealed **dark** glass containers at room temperature. The lures will then retain their activity without deterioration of the rubber. Laboratory tests with glass blood bottles kept in the dark indicate that the lures will retain at least 80% of their active ingredients for a year when kept at 30 °C. A new lure is being developed which will remain viable for the whole armyworm season of 6–8 months.

In order to minimize the problems associated with the use of aged pheromone lures, batches should be dated when dispatched and recipients should keep a record of the storage history of their lures, to ensure that they are viable when placed in the field.

Key References: Beevor *et al*. (1975)
Campion *et al*. (1976)
Cork *et al*. (1989)

5.2.3 Trap operator responsibilities

The nightly catch of *S. exempta* moths is recorded on a special record form (Figure 30). **The catch is dated for the evening the trap was working** (not the date when it was cleared), for example, if the trap is emptied on the morning of 23 December, the catch is entered on the record form for 22 December. This convention was established because more insects fly at dusk than at dawn.
It is essential:

- that trap operators keep accurate records of each night's moth trap catches on the trap record forms;
- that record forms are transmitted promptly every week to the national centres;
- that pheromone lures are changed at the recommended intervals;
- that used lures are removed from the vicinity of the trap.

5.2.4 Selecting a site for a moth trap

Pheromone traps are most widely used in trap networks because of the simplicity of their use, as they catch only mature *S. exempta* males. The trap should be easily accessible to an operator to minimize the work involved in daily recording the number of moths caught.

Pheromone traps should be set up inside a protected area, away from disturbance by children or animals and clear of any obstacles which might impede the flight of moths towards the trap (e.g. hedges or buildings). The best sites for traps are climatological or synoptic meteorological stations. Permission to place traps at these sites should first be sought from the national meteorological service.

The trap should be placed on an isolated, free-standing structure at a height of about 1.5 m above the ground, giving unimpeded air flow to the trap. If the trap must be suspended from an existing object, such as a fence line, it is important that this object does not obstruct the prevailing direction of airflow upwind, or downwind, from the trap. Do not place the pheromone trap beneath, or hang it from a tree. Site it at least 50 m from hedges, shrubs, tree lines or other windbreaks, such as earth mounds or banks, which could affect downwind drift and disrupt the shape of the pheromone plume. In growing crops, the trap height may have to be adjusted as the crop grows.

Light and Pheromone Trap Record for *Spodoptera exempta*

Month and year____________

Country____________ Station____________ No________

Date	Light trap			Pheromone trap						Weather			Relative Humidity	Rain mm	Direct Wind	Notes
				I			II									
	Male	Female	Total	Male	Female	Total	Male	Female	Total	Max	Min	Mean				
1																
2																
3																
4																
5																
6																
7																
8																
9																
10																
11																
31																

Figure 30. Example of reporting form used for moth trap catches.

Light traps should be situated in places sheltered from the wind but not overshadowed by buildings or trees, and also be at least 100 m away from artificial lights. Light traps capture both immature and mature male and female moths, therefore, the operation of some light traps within a network is useful to obtain a better picture of moth migrations, if the operating costs can be met.

Trap factor. Once a trap, light or pheromone, has been set, it should not be moved, as the number of armyworm moths caught depends on trap position. This is called the 'trap factor'. When data have been collected for a number of years, the trap factor is known and is used in evaluating and comparing catches from other traps in the trap network in conjunction with the occurrence of local outbreaks of larvae (see Section 5.3.2). Low numbers in one trap may be as important as high numbers in another.

Key references: Campion *et al*. (1976)
Odiyo (1979a)
Taylor (1986)
Taylor and Brown (1972)
Tucker *et al*. (1982)

5.2.5 Monitoring outbreaks and sampling larvae

Accurate monitoring and prompt reporting of armyworm outbreaks are essential for forecasting and control. Surveys are important for understanding the population dynamics and the control options available to deal with them. If at all possible, surveys should be carried out at every outbreak reported and full details recorded for the computerized data management system now operational in many countries (see Section 5.3.2). The following procedures are based on the experience gained by national crop protection services in member countries of DLCO-EA.

Individual farmers and other local people usually report initial sightings of outbreaks. Agricultural personnel cannot possibly monitor the situation throughout their whole country, they can only check and assess reported outbreaks to recommend control requirements and in some cases assist. Assessment of outbreaks is carried out by ground survey, but speed of access may be considerably improved by the use of a helicopter, which is a valuable tool for survey and control purposes, as outbreaks may be patchy, widely scattered and difficult to reach by land, and travel may be impeded during the rains.

Searches for newly hatched larvae should be made about 1 week after the first high catches in local pheromone traps, particularly when the catches are associated with the first heavy rains after a drought/dry period (see Sections 4.2.2, 5.2.1 and 5.3.3). Checks should also be made when warnings have been received (see Section 5.3.3). The best way of searching for young larvae is to tap, or shake, grasses or cereals, and to look for larvae suspended by silken threads (Plate 9), as well as the typical 'windowing' on the leaves (Plate 10).

Samples of larvae are essential for determining the age of the outbreak, and the dates of moth arrival, pupation and moth emergence. Density estimation should also be made using a quadrat. Samples of larvae should be taken at regular intervals along a transect line or lines through the outbreak, and not at points where the larvae are most dense and, therefore, easier and more rapidly sampled. This will mean that many larvae are collected at some sampling points and only a few at others, on account of the clumped distribution of larvae within an outbreak (see Section 2.4).

Samples of at least 50 larvae should be made at each outbreak by collecting **all** larvae, both large and small, within a defined area such as a quadrat of standard size. A quadrat encloses a known area; it is usually square, made of wood and of dimensions 50 x 50 cm, which may be sub-divided into four 25 x 25 cm quadrants to make counting large numbers of larvae easier. Alternatively, a quadrat may be circular and made from wire. A circle with a circumference of 3.54 m will enclose an area of 1 m^2. To estimate the area of a circle of smaller circumference, apply whichever of the following formulae is appropriate:

circumference = 2 x 3.142 x radius
or
area = 3.142 x $radius^2$

Counts made during sampling should be used as examples of the range of larval densities within the area sampled. They should **not** be extrapolated to apply to the whole outbreak, on account of the varying density and clumped distribution of the larvae.

Another method of sampling larvae is to use a sweep net. This method will yield qualitative figures only and should not be used in very short grass. It is, however, good for sampling larvae for ageing as it eliminates the chances of missing smaller larvae. The method of sweeping may vary but should be the same throughout each outbreak sampled and a standard number of sweeps should be taken for each transect line.

When making density estimations a **minimum** of 10 quadrat counts should be taken at random within the outbreak, however, because of the clumped nature of outbreaks, counts should be made from areas within which larvae can be seen.

All larvae collected must be preserved in 70% alcohol or methylated spirits, with a label in the bottle to record the date and place of collection, the crop attacked and the name of the collector. The **label must be written in pencil** because the preserving fluid dissolves ink from nibbed or ballpoint pens.

Outbreaks should be reported, preferably with samples of larvae, to the local district agricultural offices and national plant protection services **immediately they are found**. Samples must then be forwarded to the national armyworm co-ordinators for analysis. Pre-paid postcards printed for recording all outbreak details (Figure 31) are distributed to all agricultural officers in eastern Africa (Region 1) and increasingly in central and southern Africa (Region 2). The cards should be completed and returned immediately to national crop protection services, with any samples of larvae.

Assessment of larval age is carried out at national plant protection service laboratories. The head capsule widths of the larvae are measured, either with vernier callipers, or using a stereomicroscope fitted with an eyepiece micrometer. Figure 32 shows the correct

(a)

URGENT

ARMYWORM OUTBREAK REPORT

Province .... District .... Division .... Location ....

Date .... Reported by .... Total area infested .... hectares

Are control measures being taken: Yes/No; if Yes then: Insecticide used ....

Quantity/rate applied .... How applied ....

Colour of larvae (majority) .... Were samples of larvae collected .... (minimum 50 needed)

Size of larvae (indicate below):

I

II

III

IV

V

VI

Crop attacked	Area attacked (ha)	No of larvae on 10 different plants									
		1	2	3	4	5	6	7	8	9	10
Maize											
Sorghum											
Millet											
Wheat											
Rice											
Tef											
Sugar cane											
Other											
	Area attacked	No. of larvae in 10 different square metres									
Pastures / Grasses											

Post this report immediately, even if control is not yet finished

(b)

TAARIFA YA KUTOKEA KWA VIWAVI-JESHI

Mkoa .... Wilaya .... Tarafa .... Kata .... Kijiji ....

Tarehe .... Imetumwa na .... Mazao yalioshambuliwa

Urefu wa mazao .... Eneo lilioshambuliwa .... hekta

Hesabu ya viwavi (kwa mmea/kwa eneo ft²) 1) .... 2) .... 3) .... 4) ....

5) .... 6) .... 7) .... 8) .... 9) .... 10) .... 11) .... 12) ....

13) .... 14) .... 15) .... 16) .... 17) .... 18) .... 19) .... 20) ....

Ukubwa wa viwavi

(weka ✔)

I

II

III

IV

V

VI

Rangi ya viwavi ....

Dawa iliotumiwa ....

Uingi .... lita ....

PELEKA RIPOTI KWA DADO HARAKA
PELEKA RIPOTI HII MARA MOJA HATA KAMA VIWAVI HAWAJAZUIWA

FOR OFFICE USE

Date received .... Lat. .... Long. ....

Estimated density ....

Estimated date of oviposition ....

Estimated date of emergence ....

Figure 31. Armyworm outbreak reporting cards.
(a) English.
(b) Swahili.

positioning of the larva for head capsule measurement. This is essential to obtain accurate measurements.

Head capsule width measurements should be graphed by grouping together all those that are the same size. Examples of typical measurements are given in Table 4. A frequency histogram is then prepared. The **mean head capsule width** is calculated and compared with Figure 33 to determine the average age of the larvae and to estimate the arrival and departure dates of successive generations of moths. This information is needed for providing accurate forecasts.

Early in the season, the frequency histogram of head capsule widths is most likely to show a normal distribution (i.e. a single peak trailing off at both edges). As the season progresses and generations of armyworm overlap, histograms may show multiple peaks.

Key references: Page and Dewhurst (1987, 1992)
Rose (1975a, 1979a)

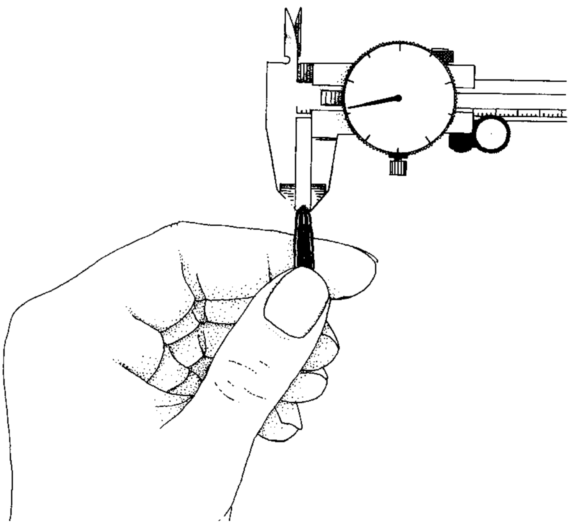


Figure 32. The correct position of a larva for measuring its head capsule with vernier callipers.

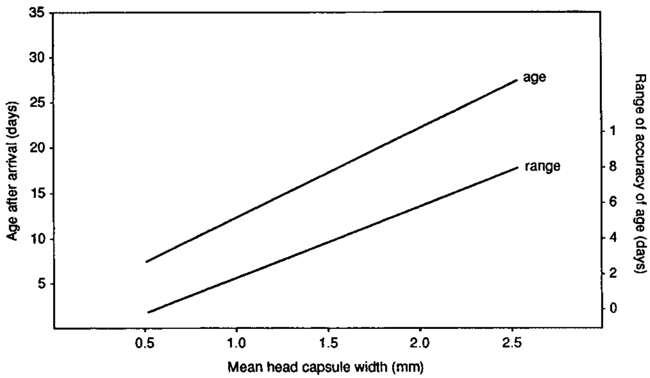


Figure 33. Rate of development of a population of *Spodoptera exempta* larvae as shown by mean head capsule width. Vertical scales show dates of moth arrival and estimated rates of development.

5.3 Forecasting outbreaks

5.3.1 Weather and data interpretation

Regional and national armyworm forecasting services should monitor wind fields, rainstorms and rainfall distribution to help interpret armyworm data and to aid forecasting. Close co-operation between the forecasting services and the national meteorological services is essential.

Climatological stations record daily rainfall totals and maximum, minimum and wet bulb air temperatures. Synoptic* stations also record these parameters and in addition wind speed and direction, air pressure, cloud amount and type, present and past weather, at internationally agreed times each day. These data are plotted on synoptic charts and analyses are drawn of the wind field and weather systems. Daily rainfall data, synoptic charts and wind streamlines are essential to the armyworm forecaster for the analysis of moth migration and subsequent forecasting. This will require an agreement between the armyworm forecasting service and the national metereological

* Synoptic meteorology is concerned with the analysis of weather systems typically hundreds to thousands of kilometres across, depicted on geographical charts using near instantaneous observations made at synoptic weather stations at specific times.

service possibly involving regular weekly visits by a member of the armyworm forecasting service to the central meteorological office to collect the data.

Imagery received from Meteosat, a geostationary meteorological satellite which has a spatial resolution of 5 x 5 km (Plate 23), can be used to identify the location and duration of nightly storm clouds whose associated winds have the potential to concentrate moths (see Section 4.1.1). The number of hours in a given period during which Meteosat infra-red images indicate cloud top temperatures of less than a certain threshold is termed Cold Cloud Duration (CCD). Values of CCD are calculated for each pixel (5 x 5 km) over the whole of the 512 x 512 pixel infra-red image on the Bradford Meteosat Primary Data User System (PDUS). Cloud top temperatures of lower than -50 °C are correlated with the presence of large cumulus or cumulo-nimbus clouds that are liable to give heavy rain and rainstorm outflow winds (Plate 26). A threshold CCD of -50 °C over 12 h is normally used for armyworm forecasting, because such storms cause meso-scale* zones of wind convergence that can concentrate flying moths (see Section 4.1.1).

Although CCD is not a direct measure of rainfall, it provides a good guide to weather conditions, for example, prolonged periods with little or no cloud, or how widespread or scattered early rainstorms may be over the region. For practical purposes, this visual assessment is directly available at various centres in eastern and central Africa, whereas rainfall records from meteorological stations take longer to obtain.

Key references: Pedgley (1982)
Pedgley *et al*. (1982, 1989)
Robinson (1991, 1995)
Tucker (1983, 1984a, 1984b, 1997)
Tucker *et al*. (1982)

* Meso-meteorology is the study of weather phenomena typically at a scale of tens of kilometres across, such as individual storms.

5.3.2 Historical data archives

Archives of armyworm trap and outbreak data continue to be built up nationally and regionally in eastern Africa. These data are stored at DLCO-EA in Nairobi and at NRI at Chatham, UK.

A specially written menu-driven data management system called WormBase has been developed and is regularly used by forecasting officers in East Africa. It is currently the only operational data handling system for pest management in regular use in Africa and was originally developed for DLCO-EA. A modified WormBase is increasingly being used in the IRLCO-CSA countries of central and southern Africa and the Yemen. The latest version of the programme (WormBase v2) is adapted for use in any country, however, there is an increasing need for the development of a data management system such as that in use for the desert locust.

Historical data are needed to provide analogues of outbreak distribution and trap characteristics (location, altitude, details of the site, years of operation – see Section 5.2.4) against which the current armyworm situation can be evaluated.

Historical meteorological data are available from the national meteorological services in each country.

5.3.3 Forecasts and warnings

Forecasting and warning systems should continue to be developed nationally and regionally to alert farmers and governments to the possibility of imminent outbreaks. Forecasts and warnings have different levels of urgency.

A **forecast** is prepared weekly, or every 2 weeks, based on information received at the national or regional offices from the monitoring systems, and describes expected future armyworm developments.

A **warning** is issued as an alert to the likelihood of the imminent occurrence or redistribution of armyworm infestations, issued while they are still at the moth or early larval stages. Warnings are of greater importance than weekly forecasts and should be sent by the fastest possible means to agricultural officers in the areas likely to be invaded. Telephone and radio networks should also be used. In some countries warnings are broadcast on local radio, through the national press and through local gatherings, such as church services.

Warnings must be acted upon immediately.

All forecasts and warnings are prepared by integrating information on each of the following:

- the distribution of armyworm populations as currently reported through the monitoring system (see Section 5.2);
- the distribution of previously reported armyworm populations inferred as still active, allowing for their development with time, for example, oviposition by moths sampled in large numbers in traps and the subsequent hatching and development of larvae, or emergence of moths from earlier outbreaks (see Section 3.1.4);
- the dominant winds through the period under review, on which moths could migrate, and any synoptic or meso-scale zones of wind convergence in which moths could become concentrated (see Section 4.1.1);
- the distribution of rainstorms, particularly early in the armyworm outbreak season, around which local wind convergence could concentrate moths (see Section 4.1.1);
- the historical precedent for the development of outbreaks anticipated (see Section 5.3.2 and Figure 4).

The last criterion applies particularly to countries with a long history of data collection and experienced armyworm forecasting officers.

Positive forecasts and warnings are issued when moth population numbers reach potentially dangerous levels. These are recognized by comparison with historical data and vary with time, location and type of trap (see Sections 5.2.1 and 5.2.4).

For any infestation of known age (estimated from head capsule measurements – see Section 3.1.4), the date of oviposition is inferred by subtracting the appropriate number of days from the sampling date, and the emergence date estimated by adding the reciprocal number of days, using the life table most appropriate to the location of the sampling site (Tables 6 and 7). For example, IV instar larvae collected at Athi River near Nairobi on 10 April would have been in the egg stage 12–14 days earlier, during the period 28–30 March, and peak moth emergence would be expected 22–24 days later, during 2–4 May. Where no samples are collected, the larvae are assumed to be in IV to VI instar, their most conspicuous stages, and dating is accordingly less precise. For the same example, oviposition would have taken place during 16–30 March and emergence during 21 April–4 May. This type of calculation has been built into the WormBase forecasting system and is generated automatically for every outbreak recorded in the database.

Computerized databases have been established within the Crop Protection Branch, Kenya, and PCS, Tanzania, in member countries of DLCO-EA and IRCLO-CSA, and programmes for manipulating the data have been written and upgraded as WormBase v2. In addition, two Expert Systems have been developed to produce computer-assisted forecasts. Such databases may also be used for generating models to predict the overall population dynamics of the African armyworm in East Africa.

An example of a National Forecast is given in Figure 34a. The Regional Forecast (Figure 34b) is a situation report based on information received from member countries in the region. It also provides a forecast of expected outbreaks within the region. Its particular value is to provide an overview of armyworm population movements within and between countries, as well as the current weather conditions over a wide area. Forecasting the dates and locations of outbreaks in detail is better achieved by the national plant protection services, where they exist.

KENYA AGRICULTURAL RESEARCH INSTITUTE

Telephone: Karuri 0154-32880/1-66 for National Armyworm Forecasting Service
Telegrams : Kari/NARC Nairobi
Address: NARC Muguga, P.O. Box 30148, Nairobi

ARMYWORM REPORTS AND FORECASTS Nos 19,20, 21 OF 1990/91 SEASON.
The general situation during weeks 4-24 February 1991

1. LARVAE: Kenya continued to remain free of armyworm infestations during 4-24 February 1991

MOTHS: The current persistent dry weather conditions in Kenya continue to put the population of *Spodoptera exempta* moths in the country under check. The following were the trap catch reports received.

(i) Week 4-10 February 1991
4 moths were caught during the week at Njoro (light trap: 1) and Ruiru (Pheromone trap: 3). Muguga, Msabaha, Eldoret DAO's Office and Isiolo reported nil.

ii) Week 11-17 February 1991
13 moths were caught at Ruiru and Eldoret DAO's Office pheromone traps (2 and 11 moths respectively). Muguga and Isiolo had nil.

iii) Week 18-24 February 1991
Only Muguga's report was available, with a catch of 1 moth in the pheromone trap.

2. FORECAST FOR WEEK 1-7 MARCH 1991
Kenya remained free of armyworm infestations during the Forecast weeks 15-28 February 1991. During the coming first week of March 1991 there is a low probability of fresh infestations occurring at suitable localities in Nyanza and Western Provinces, including Narok District. The rest of the country is expected to remain free of armyworms.

Figure 34a. Example of national armyworm forecast.

Radio message to: All DLCO-EA Base Managers and PCLO (Addis Ababa).

1. ARMYWORM SITUATION FOR WEEK No. 27 DATE 13-19/4/91

TANZANIA:	Unconfirmed outbreak reported in Moshi. Moth catches awaited.
ETHIOPIA:	No outbreaks reported. Moth catches reported in Dire Dawa 2 P. and 18 L. Other areas' reports awaited.
KENYA:	No outbreaks reported. No moth catches reported.

UGANDA, SOMALIA: Reports awaited.

2. FORECAST FOR WEEK 20-26/4/91

KENYA:	High probability of outbreaks in Coast Province (Malindi, Lamu, Kilifi, Taita Taveta and Kwale) and a medium probability of outbreaks in Tana River and the Lake region (from CCD)
TANZANIA:	High probability of outbreaks in Morogoro region, Kilimanjaro region, and Central Tanzania (Singida and Arusha regions) (from CCD)
SOMALIA:	Medium probability of outbreaks in western Somalia (from CCD).
ETHIOPIA:	Low outbreak probability in Sidamo and Gamu-Gofa regions (from CCD).
UGANDA:	Expected to be free of armyworm outbreaks.

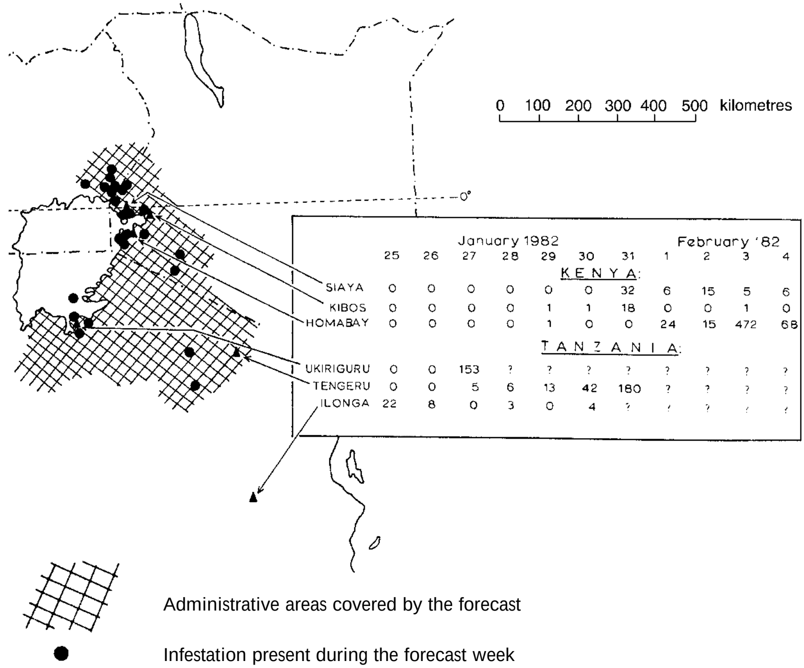
Date and time sent, radio operator to sign

Ethiopia..................	Kenya.....................	Somalia.................
Tanzania.................	Uganda...................	Sudan....................

Figure 34b. Example of regional armyworm forecast.

5.3.4 Long-term forecasts

There is an inverse correlation between the rainfall in November and December in Kenya and Tanzania and the severity of the subsequent outbreak season in those countries. This negative relationship can be utilized to give a long-term warning and enable crop protection services to prepare for control operations.



	January 1982							February '82			
	25	26	27	28	29	30	31	1	2	3	4
KENYA:											
SIAYA	0	0	0	0	0	0	32	6	15	5	6
KIBOS	0	0	0	0	1	1	18	0	0	1	0
HOMABAY	0	0	0	0	1	0	0	24	15	472	68
TANZANIA:											
UKIRIGURU	0	0	153	?	?	?	?	?	?	?	?
TENGERU	0	0	5	6	13	42	180	?	?	?	?
ILONGA	22	8	0	3	0	4	?	?	?	?	?

Figure 35. Armyworm forecast verification, showing sites of light traps, catches of *Spodoptera exempta* moths (tabulated) associated with the first seasonal migration of populations between Tanzania and Kenya in 1981–82, areas (hatched) for which outbreaks forecast for 9–15 February 1982, and infestations present during the forecast week (after Odiyo, 1986).

A simple 'rule of thumb' method for outbreak prediction based on this negative relationship has been established.

5.3.5 Forecast verification

The reliability of forecasts and warnings must continue to be routinely verified to enable their accuracy and value to be improved. This is done by plotting maps of the locations of reported outbreaks in relation to areas predicted (Figure 35).

The reasons for any omissions or overstatements in the forecast need to be identified and steps taken to avoid repetition of errors in the future.

Key references: Betts (1976)
Blair and Catling (1974)
Blair *et al*. (1980)
Haggis (1984, 1986a, 1996)
Odiyo (1979a, 1981, 1984, 1989)
Tucker (1984a)

5.4 Control

5.4.1 Agricultural practices

Maize crops greater than 30 cm in height are unlikely to become seriously infested by newly hatched armyworm, as the maize leaves are too tough for the small larvae to eat (see Section 3.1.4). Farmers are advised to keep crops free of grass weeds to avoid infestation, however, if fields do become infested, grass weeds should be left as an alternative (sometimes preferred) food source for the larvae which would deter them from moving into the crop. Such a practice constitutes a type of cultural control and falls within the scope of integrated pest management (IPM).

Some varieties of maize are more readily eaten by armyworm than others. For example, ‘Katumani’, a dryland variety developed and widely grown in Kenya, is preferred to other hybrid varieties. In areas where there is a high probability of armyworm infestation, varieties such as this are most at risk. They are usually varieties that mature rapidly in the dryland farming areas where rainfall is unpredictable.

5.4.2 Biological control

None of the predators and parasitoids of armyworm are numerous enough to be effective for the natural control of outbreaks. However, the naturally occurring pathogen, SpexNPV, has been used to control armyworm infestations successfully. The virus is slow acting, taking several days to kill the larvae and it is, therefore, important to find outbreaks and apply the virus suspension to early stage larvae before serious crop damage occurs.

Control has been achieved by spraying infestations of *S. exempta* larvae with a water suspension of diseased larvae that were collected at previous infected outbreaks. However, this is a time consuming and inefficient method of applying the virus as the liquid is heavily contaminated with larval remains, although it may have uses in the future. Laboratory formulation of the virus from specially reared *S. exempta* larvae should improve the ease of application and its effectiveness. Following a lengthy development programme, a feasibility study on virus production and formulation was undertaken; it was concluded that SpexNPV could be readily produced and formulated in the laboratory, if funding was made available.

Promising new products include some of the insect growth regulators (IGRs), and a bacterial preparation using a strain of *Bacillus thuringiensis* (Berliner). These, like the species-specific SpexNPV, require full laboratory and field evaluation against *S. exempta* under differing field conditions before their suitability for control of armyworm can be advocated.

If these products are cost-effective, confirmed as being safer to non-target organisms, man and the environment, they are likely to be adopted in the future.

Key references: Brown (1966b, 1972)
Brown *et al*. (1984)
Brown and Swaine (1965a)
Broza *et al*. (1991a)
Fisk *et al.* (1993)

5.4.3 Chemical control

It is as larvae that *S. exempta* causes damage to crops and pasture, and this is the only stage in the life cycle readily susceptible to control by pesticides. The eggs are not found easily and the pupae are subterranean. The moths cannot easily be controlled because they are dispersed as they fly downwind at night (see Section 3.2.2), unlike the cohesive day-flying swarms of the desert locust which can be sprayed directly by aircraft.

S. exempta larvae are susceptible to a wide range of pesticides. The major problem is that larvae are not normally noticed until they turn black at the III–IV instar moult, by which time they have already been in the field for 7–10 days and feeding is reaching a damaging level (see Section 3.1.4 and Figure 9). This means that there are only 8–12 days left to undertake control before the larvae pupate, and each day control is delayed allows further damage to the host plant. Rapid response is essential, therefore, and can only be achieved by acting on warnings or forecasts (see Section 5.3.3), checking young cereal crops for infestations 1 week after moth invasion (as indicated by local traps – see Section 5.2.1) and by being well prepared and equipped for control operations. In the absence of warnings or trap data, it is worth checking crops for newly hatched larvae 1 week after storms, particularly the first storms of the wet season in the area (see Section 4.1).

A list of effective insecticides is given in Table 9. This includes dosage rates for different products and types of formulation, as well as withholding (pre-grazing) periods. Improvements in application technology have meant that **volume application rates (VAR)** for some insecticides, especially fenitrothion and cypermethrin, can be reduced to 0.3–0.4 l/ha in many situations, i.e. half the quantities recommended in earlier publications, such as the armyworm booklet on which Table 9 is based, and still be effective against *S. exempta* larvae. Although no recent definitive field trials have been carried out to determine effective rates, it is reasonable to assume those recommended for desert locust control will be effective against armyworm and it is likely that rates could be reduced even further without affecting their efficacy against armyworm. Further trials with new pesticides are much needed.

It should be noted that for environmental reasons and human safety, persistent and more toxic pesticides are not included in Table 9 and must **not** be used. These include all the chlorinated hydrocarbons such as DDT and BHC. A recent ruling (2000) by the EU Standing

Table 9. Some insecticides suitable for armyworm control, their application rates and with-holding (pre-grazing) periods,based on manufacturers' 'all purpose' recommendations

	Water-dispersible formulations (WDF)		Ultra-low-volume formulations (ULV)			With-holding period
Common name	**Trade name**	**Formulation concentration**	**Volume (litres) of formulation in 100–250 l[†] of water (w)**	**Formulation concentration**	**Volume Application Rate (l/ha)**	**Weeks or days (d)**
Organophosphates						
Malathion	Kilpest	50% m.l.	1.3–3.0	95% Tech.	1.25	1 w
	Cythion	50% e.c.	1.3–3.0	95% Tech.	1.25	1 w
Fenitrothion*	Accothion	50% e.c.	1.3	96% Tech.	0.5	7–10 d
	Agrothion	50% e.c.	1.3	96% Tech.	0.5	7–10 d
	Folithion	50% e.c.	1.3	96% Tech.	0.5	7–10 d
	Simbathion	50% e.c.	1.3	96% Tech.	0.5	7–10 d
	Sumithion	50% e.c.	1.3	96% Tech.	0.5	7–10 d
Trichlorphon	Dipterex	50% e.c.	1.0–2.0	250 ulv	2.0-2.5	7–10 d
Phoxim	Volaton	50% e.c.	1.3	900 ulv	1.0	2 w
Chlorphyriphos	Dursban	48% e.c.	0.5	24% ulv	1.0	2 w

Quinalphos	Ekalux	25% e.c.	2.0	30% ulv	1.5	2 w
Tetrachlorvinphos	Gardona	24% e.c.	3.0	35% ulv	2.0	1 w
Primiphos methyl	Actellic	25% e.c.	3.0–4.0	500 ulv	1.5–2.0	2 w
Carbamates						
Carbaryl	Sevin	85% w.p.	1.2	25% ulv	2.5	1 w
Synthetic pyrethroids						
Cypermethrin*	Ambush CY	5% e.c.	1.0	2.5% ulv	1.0	2 d
	Ripcord	6% e.c.	0.85	2.5% ulv	1.0	2 d
	Sherpa	6% e.c.	0.85	2.5% ulv	1.0	2 d
Deltamethrin	Decis	2.5% e.c.	0.4	0.5% ulv	1.5	2 d
Fenvalerate	Sumicidin	7% e.c.	1.0	3.0% ulv	2.0	2 d
Permethrin	Ambush	10% e.c.	0.6	4.0% ulv	1.0	2 d

e.c.= emulsifiable concentrate; w.p. = wettable powder; m.l. = miscible liquid ulv = ultra-low-volume; Techn. = Technical material.

*Recommended rates for fenitrothion and cypermethrin can be reduced to 0.3–0.4 l/ha in many situations, particularly young cereals and short grassland.

† This is the approximate volume of spray per hectare; in practice, target spraying may enable a greater area to be covered.

Committee on Plant Health has recommended a ban on permethrin by July 2003.

The application of dusts is not recommended, as pesticides in this form are difficult to apply efficiently and it is difficult for operators to avoid inhaling the dust. If application of dusts is unavoidable, then operators **must** wear masks. Safety precautions are fully described below (see Sections 5.4.10–5.4.13).

In selecting the type of equipment and application method to be used in any control operation, the area to be treated, safety (human and environmental), time available, cost and insecticide efficacy all have to be taken into account. Pesticide may be applied by using portable sprayers, vehicle-mounted sprayers, or aircraft-mounted sprayers. The principle for all these spray platforms is the same and the choice is made according to the scale and accessibility of the outbreak. The approximate time needed to treat areas using different spray equipment is given in Table 10.

Table 10. Speed of treatment with insecticide using different application techniques

Size of outbreak and type of site	Control equipment	Estimated coverage per sprayer
<1–5 ha Small plots, patchy of larvae	Knapsack sprayer, dusting tins/bags (dusting is **not** recommended)	5 ha/day 5 ha/day
1–100 ha Small and medium size farms	Hand-held ULVA* sprayers	6 ha/h (30 ha/day)
>100 ha Medium to large farms/ rangeland	Vehicle-mounted ULVA* sprayers; Aircraft with ULV spray gear	70–140 ha/h (350–700 ha/day) 1000 ha/h (4000 ha/day)

* Ultra-low-volume applicator.

Ultra-low-volume (ULV) spraying is the most efficient method for armyworm control, as infested areas can be treated much more rapidly and with less volume of insecticide than with other methods. Application techniques should be chosen to suit the local situations, for example, in eastern Africa, ULVA use is common while in the Great Lakes region knapsack sprayers are more commonly used.

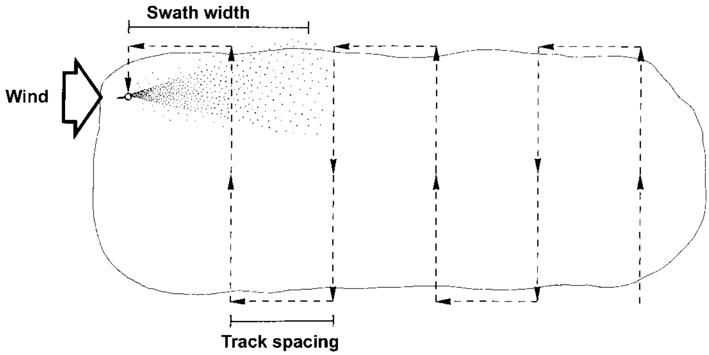
Key references: Brown (1970b)
Page and Dewhurst (1987, 1992)
Rose (1978a)
Yeates (1973b)

5.4.4 Ultra-low-volume (ULV) spraying

Principle. ULV spraying is the application of a small volume (0.3–3.0 l/ ha) of oil-based formulation (to stop evaporation) in small and relatively uniformly sized drops of concentrated pesticide. ULV spraying relies on the wind to carry small droplets of pesticide over the target area. This applies to all sprayers, including those which generate an air blast, such as the Micronair AU8110, where the initial velocity is used to propel the spray away from the operator and, when directed upwards, takes it to a greater effective emission height, giving a wider swath. Wind is still needed to carry the drops to the target over a swath width of 20–30 m (hand-held sprayer), 50–100 m (vehicle-mounted sprayer), or 200–300 m (fixed-wing aircraft) (see Figure 36a). Since droplet deposit within these swaths is not uniform (a peak close to the sprayer and a long tail of low level deposit further downwind), the swaths must be overlapped in order to improve deposit uniformity. As a result, track spacings must be set considerably less than swath widths in order to achieve this overlap.

Conditions for ULV spraying. The wind must be steady, with a wind speed between 1m/s and 10 m/s (at the minimum wind speed of 1 m/s, a distinct breeze can be felt on the face – see Appendix 4). **Never spray under calm conditions**. If wind speeds are greater than 10 m/s, or if the wind direction is variable (as in conditions of strong convection), ULV spraying should **not** be undertaken.

(a)



(b)

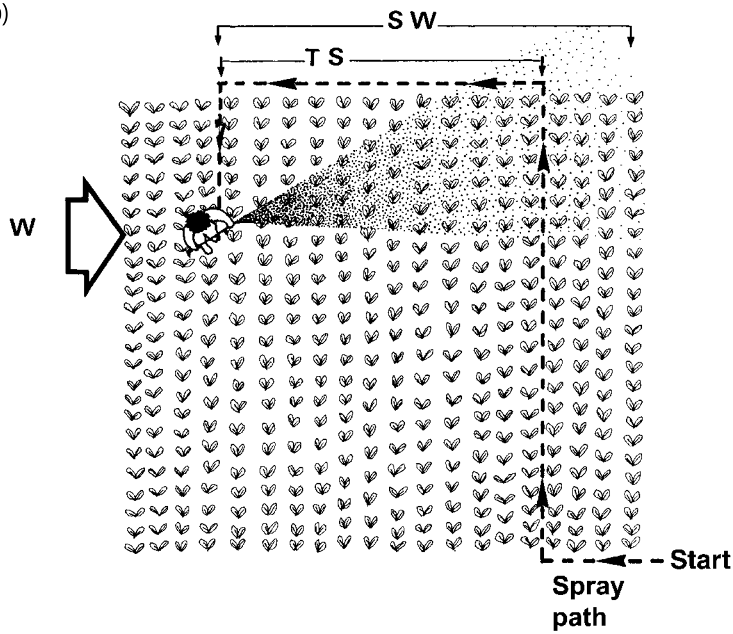


Figure 36. Route to be followed by spray operator in relation to wind direction during ULV spray application.
(a) Plan view showing swath width and track spacing, applicable to all types of sprayers.
(b) Plan view, illustrating 12-row (10 m) track spacing (TS) between spray paths and position of spray head of hand-held sprayer relative to operator and wind (W). Swath width (SW) also shown.

(c)

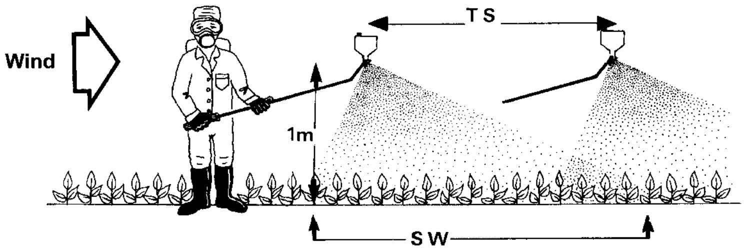


Figure 36 *cont.* Route to be followed by spray operator in relation to wind direction during ULV spray application.
(c) Front view, illustrating overlapping swaths (SW) from successive spray paths at track spacing (TS), position of spray head of hand-held sprayer relative to operator and wind, and protective clothing worn by operator.

Spraying must not be carried out when there is strong sunshine, which causes convective upcurrents to develop and will carry small drops skywards and away from the area being treated. As a general rule, spraying should be carried out before 10.30 h or after 16.00 h although these limits may be relaxed if there is a strong, steady wind. Conditions will vary with locality and season and operational decisions will have to be made, bearing in mind variables such as urgency of control and insecticide efficiency.

5.4.5 Types of spray equipment

Hand-held spinning disc sprayers such as the Micro-ULVA or Micron Ulva+ are widely used for armyworm control. These sprayers are powered by 4–7 1.5 volt batteries which provide power to rotate the atomizer. A set of good standard batteries (‘D’- type) should last for more than 8 h of spraying before requiring replacement. The cost and availability of batteries in some parts of Africa is a limitation to the use of ULV sprayers by farmers. The supply of batteries should be ensured by pre-season planning at government level by national crop protection units.

Trained teams of ULVA operators can spray hundreds of hectares per day in areas not accessible to vehicles (see Table 10). This rate of work cannot be achieved with knapsack sprayers that are widely owned and used by farmers in many parts of Africa. They are slow, cumbersome and much too heavy, requiring large quantities water, for use in armyworm control.

Figure 37 illustrates a hand-held ULVA sprayer.

Knapsack sprayers (lever-operated), of which several types are available, use hydraulic nozzles whose function is to break up the liquid under pressure (the drops produced by fine nozzles are not as small as those used in ULV spraying). The different nozzle sizes are usually colour-coded and the local plant protection services should be

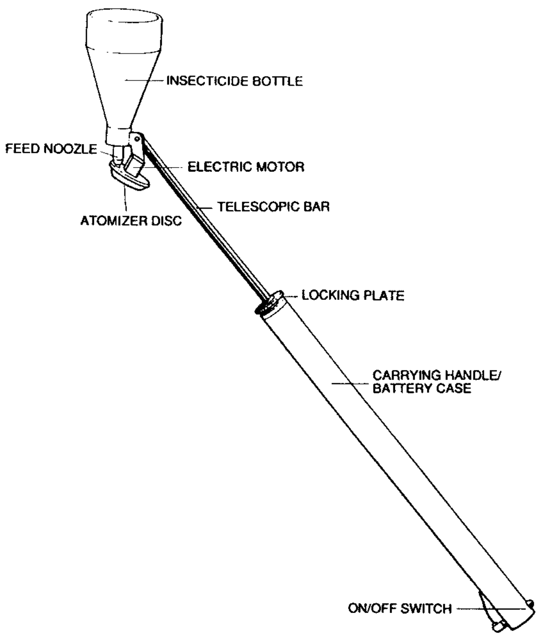


Figure 37. An ultra-low-volume applicator (ULVA) (original design). Modification by manufacturer allows refuelling without removing the redesigned reservoir from the sprayer.

able to advise which nozzles are suitable for armyworm control, and where the sprayers and nozzles are available.

Vehicle-mounted spray equipment. In ULV systems the spray is atomized through ULV rotary cage atomizers. Common examples are the Micron Ulvamast and the Micronair AU 8110. The Micron sprayer is a ‘passive drift’ sprayer meaning that the spray is released directly into a crosswind. The Micronair sprayer is an airblast sprayer which means that the spray is initially propelled upwards or sideways for a few metres by an airblast before this dissipates and releases the spray into the cross-wind. Vehicle-mounted sprayers are useful where the area is too large for treating with hand-held spraying equipment or when aircraft are not available. A locally made vehicle-mounted sprayer is illustrated in Figure 38 and has all the usual features of a passive drift sprayer.

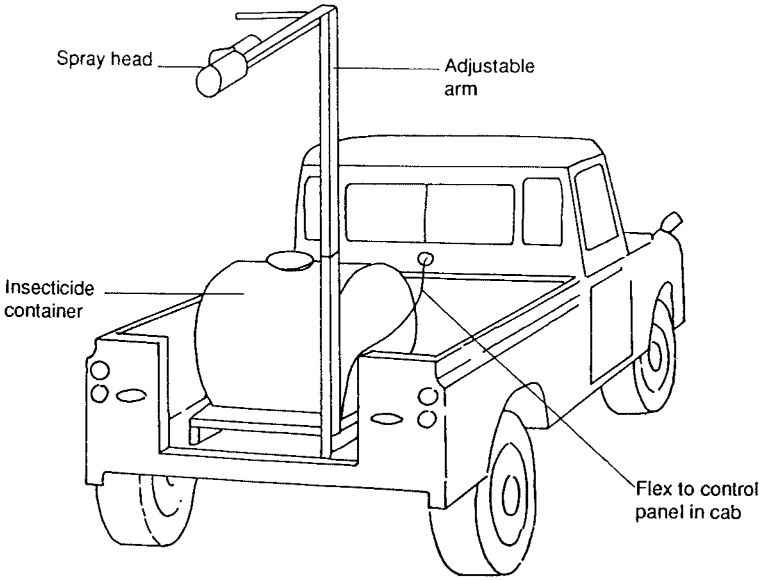


Figure 38. A vehicle-mounted ULV spray applicator developed by DLCO-EA.

Aircraft-mounted spray equipment. Most ULV aerial spraying is carried out using rotary cage atomizers. These are usually rotated by the action of the aircraft's forward speed through the air on the atomizer's windmill blades, but there are hydraulically and electrically powered versions available for fitting to helicopters where the airspeed may fluctuate. The rotating heads are typically mounted beneath the wings or the fuselage or sometimes on pods suspended beneath the wings of the aircraft.

5.4.6 Calibration of equipment

Purpose and principles. All sprayers require calibration for all methods of application and all classes of pesticides. It must be done to check and adjust equipment settings in order to obtain the desired spray result. Good results can be safely and efficiently achieved with the recommended dose of pesticide, provided that three important calibration factors are taken into account.

(i) **Droplet size.** It is necessary to check that the sprayer is adjusted to produce a droplet size that will give a good spray distribution and effective kill. The volume median diameter (VMD) of the spray spectrum recommended for armyworm control is 70–90 µm. In practice, checking droplet size on most ULV machines means checking that the sprayer is set to give the correct rotational speed of the atomizer, for example, correcting the blade angle on a Micronair or the number and condition of batteries in the Micro ULVA.

(ii) **Emission height.** The height at which the cloud of spray droplets is released affects the swath width. Generally the greater the emission height, the wider the swath. However, if the emission height is too great there is a risk that the droplets will not come down in the target area. A hand-held sprayer with an emission height of about 1 m might give a swath width of about 25 m, whereas an aircraft flying at 10 m could produce a swath width of 250 m. The sprayer height must always be adjusted for the prevailing conditions.

(iii) **Dosage rate.** This is the quantity of active ingredient of pesticide (expressed in grams of active ingredient – g a.i.) to be applied per hectare. It is the dose that will be sufficient to kill the armyworm without waste and without damage to the environment, and which varies between products (Table 9). Recommended dosage rates and formulation concentrations are normally given on the label on the pesticide package. Further field trials are continually needed to confirm effective doses against armyworm using new pesticides as these become available.

Regulation of the dosage rate and volume application rate. To achieve the correct dosage rate for a given pesticide, it is necessary to regulate the **volume application rate (VAR).** This is the volume of pesticide applied per unit area and is usually expressed in millilitres per hectare (ml/ha). The required VAR can be calculated from the area dosage and the concentration of the pesticide formulation (g a.i. per unit volume) using the following formula:

volume application rate (ml/ha) =

$$\frac{\textbf{recommended dosage (g a.i./ha) x 1000}}{\textbf{formulation concentration (g a.i./l)}} \qquad \text{(Formula 1)}$$

In order to achieve this correct VAR, three components must be regulated:

flow rate of the sprayer (also called the emission rate) – if the flow rate increases, the VAR increases;

forward speed (of the sprayer, not the rotational speed of the atomizer) – if the forward speed increases, the VAR decreases;

track spacing (the distance between spray passes) – if the track spacing increases, the VAR decreases.

It is important to understand the relationship between these components and to be able to calculate the correct value for them for any pesticide, in order to achieve the recommended VAR and area dosage. Operators must be advised on the flow rate, forward speed and track spacing to use.

The relationship of the three components is shown by the formula:

flow rate (ml/min) =

$$\frac{\textbf{VAR (ml/ha) x speed (m/min) x track spacing (m)}}{\textbf{10 000}} \qquad \text{(Formula 2)}$$

The formula can be rewritten to calculate the other variables as follows:

$$\textbf{VAR (ml/ha)} = \frac{\textbf{flow rate (ml/min) x 10 000}}{\textbf{speed (m/min) x track spacing (m)}} \qquad \text{(Formula 3)}$$

$$\textbf{speed (m/min)} = \frac{\textbf{flow rate (ml/min) x 10 000}}{\textbf{VAR (ml/ha) x track spacing (m)}} \qquad \text{(Formula 4)}$$

$$\textbf{track spacing (m)} = \frac{\textbf{flow rate (ml/min) x 10 000}}{\textbf{VAR (ml/ha) x speed (m/min)}} \qquad \text{(Formula 5)}$$

Calculations. Before using a new pesticide or changing the application technique, calculations must be made to ensure that the correct area dosage rate is applied. Three steps are required for this process.

(i) **Determine** the required area dosage in g a.i./ha by consulting the label on the pesticide container, the published list of recommended doses, or the results of recent field trials.

(ii) **Check** the concentration of the pesticide (from the label) and calculate the VAR in ml/ha, using Formula 1 above.

(iii) **Select** an appropriate forward speed and track spacing, and calculate the flow rate required to achieve the VAR, using Formula 2 above.

The **forward speed of the sprayer** is usually measured in metres per second or, in the case of aircraft, kilometres per hour, miles per hour or nautical miles per hour (knots). Typical forward speeds are 160 km/h for a fixed-wing aircraft (equivalent to 100 mi/h or 90 knots) and 7 km/h for a vehicle. **Remember** to convert speeds from km/h, mi/h or knots to m/min (see Appendix 4) before making any calculations.

With a hand-held sprayer, the time taken for an operator to walk a distance of at least 100 m must be determined. To do this, the person should carry the sprayer along the measured line several times, noting the time taken for each traverse. The average time and forward speed can then be calculated:

$$\textbf{Speed} = \frac{\textbf{Distance}}{\textbf{Time}} \qquad \text{(Formula 6)}$$

Track spacing is the interval between spray tracks, measured in metres. This is decided upon before spraying, taking into account the method of application, the target, the terrain, the weather and how uniform the deposit must be. Track spacing is typically 10 m for hand-held spinning discs, 30 m for vehicle passive drift sprayers, 50 m for vehicle-mounted airblast sprayers and 100 m for aircraft (see Figure 36), although these can be modified according to prevailing conditions.

In practice, the speed is often limited by external factors such as the flying speed of the aircraft or the walking speed of a person, and the track spacing also is limited by the swath width produced by the sprayer.

Swath width is the distance the spray is carried downwind from the spray head and is not to be confused with track spacing which is predetermined for each operation (see Section 5.4.7 and Figure 36).

Consequently, the flow rate is the easiest variable to adjust to achieve the correct dosage rate.

Whatever the type of sprayer, it is important to measure and adjust the flow rate before spraying starts. The figures given by the equipment manufacturer are only a guide because the flow rate will vary according to the type of formulation and the temperature during the control operation, both of which influence the viscosity, which has a significant effect on flow rate.

Flow rate measurement and adjustment. If the spraying equipment does not have an automatic flow meter, measure the flow rate either by collecting and measuring the volume emitted from the sprayer over a given time (the ‘collection method’), or by measuring the reduction in volume in the tank after a given period of spraying (the ‘loss method’). The first of these methods is suitable for lever-operated knapsack sprayers and rotary atomizers (with the atomizer stationary), but it is not suitable for air blast atomizers like the exhaust nozzle sprayer (ENS). The ENS is not recommended for armyworm control, but if it is used, the flow rate may be calculated by measuring the amount needed to refill the tank to a predetermined level after a known time.

Calibration of spraying equipment, using pesticide or diesel must be undertaken well away from human habitation, buildings, open water or food crops or stores.

To check the flow rate of a hand-held sprayer use either of the following methods.

The collection method

(i) Fill the sprayer with spray liquid (pesticide).
(ii) Place a funnel and measuring cylinder underneath the atomizer (spray head), in a bucket or similar container to collect spillage.
(iii) **Without switching on the disc,** invert the atomizer (ULVA), or pump the lever (knapsack sprayer) which takes the pesticide from the reservoir to the atomizer, and allow the liquid to collect in the measuring cylinder for a predetermined time (e.g. 3 min), using a watch or stopwatch.
(iv) Measure the volume of liquid collected.
(v) Calculate the flow rate in ml/min by dividing the volume collected by the number of minutes taken.
(vi) Repeat stages (i) to (v) twice more to obtain the average flow rate of the sprayer.

The loss method

(i) Measure an **exact** volume of liquid (e.g. 500 ml for a ULV sprayer or 3 l for a high volume sprayer) and put it into the reservoir of the sprayer.

(ii) In the open, spray for a predetermined time (e.g. 3 min), using a watch or stopwatch.
(iii) Measure the volume of liquid remaining in the reservoir.
(iv) Calculate the flow rate in ml/min by dividing the volume **used** by the number of minutes taken to spray the measured liquid.
(v) Repeat stages (i) to (iv) twice more to obtain the average flow rate of the appliance.

If the flow rate measured in this way will not achieve the desired VAR, it will be necessary to make an adjustment to the sprayer by using a different restrictor or nozzle or, if this is not possible, making some adjustment to the track spacing (Formula 5) or forward speed (Formula 4) to compensate.

For example, using cypermethrin 2.5% ULV (one of the insecticides considered effective for the control of armyworm – see Table 9), a normal walking speed and a track spacing of 10 m will result in an application rate of 1 l/ha with most ULV products if the red restrictor is fitted to the ULVA. To apply the insecticide at 0.5 l/ha, the walking speed would have to be doubled, or a smaller restrictor fitted. A suitable swath width to be achieved is 20–30 m.

Aircraft equipment. Calibration of aircraft equipment can be carried out in flight or on the ground if the pesticide pump is electric. If the pump is a windmill type, calibration on the ground is usually not possible due to the difficulty of achieving the correct pump pressure solely from the slipstream of the aircraft propeller. Calibration should be carried out with the particular pesticide to be sprayed. This is not always considered acceptable due to environmental, human safety and financial constraints. If calibration is carried out with diesel fuel, or other liquid, the relationship between its flow rate and that of the pesticide must be established through at least one calibration with the pesticide itself – sometimes it can be significantly different.

Procedure for calibration in flight.

(i) Load 50 l of pesticide (or diesel) into the tank.
(ii) Set the blade angle by reference to the manufacturer’s handbook using known flying speed and required droplet size.

(iii) On the ground, with the aircraft engine running, turn on spray in the normal way (collect the emitted liquid for recycling) until the pump pressure falls on the cockpit pressure gauge. At this moment, all spray lines are primed with liquid, but the tank and pipes leading to the pressure gauge are empty.
(iv) Load the amount of pesticide (or diesel) to be released in 1 min, according to the sprayer manufacturer's recommendations.
(v) Take off and spray again until the pressure falls, noting the time taken to reach this point. Return to the airfield.
(vi) If the time taken to emit the spray was less than 50 s, on each rotary cage (such as a Micronair) close the restrictor slightly, and if more than 70 s, open it a little.
(vii) Repeat (iii)–(v) until the time is between 55 s and 65 s. (This method can also be used to check whether a flow meter is functioning properly.)

Always estimate the amount of pesticide to be used and compare the figure with what has been sprayed, for example, 50 l in the tank should treat 100 ha at 0.5 l/ha.

If the amount of pesticide used is greater or less than expected, check that flow rate and track spacing are correct.

5.4.7 Application techniques

Principle of ULV application method. Figure 36a illustrates the spraying pattern for ULV applications. It applies to all methods, whether walking with a hand-held rotary atomizer, driving with a vehicle-mounted sprayer, or spraying from an aircraft. When ULV spraying, the operator must proceed **across the direction of the wind** (at right angles (90°) to it). At the end of each spray pass, the turn must be made **into the wind** (upwind), **not in the direction of the wind** (downwind).

The target at the extreme downwind end of a swath may not receive a sufficient dose to kill the insects. To overcome this, spraying is done incrementally, i.e. with **a track spacing less than the swath width**, so that spray deposition in the area of overlap is augmented from two or three successive passes.

Spray pattern. The spray area is traversed at intervals of the track spacing to ensure that the recommended area dosage is applied (see Section 5.4.6). After each spray pass, the sprayer is taken through the area in the opposite direction to the previous edge at a distance **upwind** of one track spacing (Figures 36a and 36b). **Spray flow must be switched off while moving between passes**. To avoid travelling through the area where spray has been carried from previous passes it is essential to **start spraying at the downwind edge of the area to be treated**. When using hand-held sprayers, the atomizer should always be held **downwind** of the operator so that the spray drops are carried away from the operator by the wind.

Hand-held rotary atomizer sprayers (e.g. the battery-powered Micro-ULVA). The speed of rotation of the atomizer and, therefore, the droplet spectrum, depends on the voltage. As the batteries become discharged, the atomizer slows down. When this happens the batteries must be replaced. Although an experienced operator can tell from the noise of the electric motor when batteries need replacement, it is better to check the speed of the disc using a device such as a vibrating needle tachometer (e.g. Vibratak).

When spraying in wind speeds of 1–3 m/s, the ULVA should be held about **1 m** above the vegetation. At higher wind speeds, the head of the sprayer should be held nearer to the ground. If no means of measuring wind speed is available, follow the Beaufort wind scale (see Appendix 4).

Knapsack sprayers (lever-operated). Coarser nozzles (with bigger holes) make larger drops and the flow rate is greater. It is usually necessary to apply a higher VAR (see Section 5.4.6) to the area when using a coarse nozzle because there are fewer larger drops. Conversely, using a finer nozzle that produces smaller drops can make some reduction in the VAR. When nozzle orifices become wider through wear, the nozzle should be replaced because a coarse spray with a poor droplet spectrum will waste the operators' time and decrease the effectiveness of the treatment.

Knapsack sprayers require clean water for pesticide dilution. **Never** try to fill the spray tank more quickly by removing the filter that is fitted in the filler opening, as vegetation or dirt will clog filters and nozzles of the sprayer.

Vehicle-mounted sprayers. Techniques are similar to those used for hand-held sprayers. However, care should be taken to ensure that speeds, quantities and application rates are adjusted by reference to operation manuals (see Section 5.4.6).

Areas of infestation of this scale should be marked with flags prior to spraying. The accuracy of spray placement is improved by clearly marked track spacing. (See Section 5.4.10 for safety precautions for flag marker teams.)

If Differential Global Positioning System (DGPS) equipment is available, there is no need for flag marking. The correct use of a DGPS must, however, be clearly understood.

Aerial applications. The area to be treated must be marked out in advance by putting out coloured marker flags at the corners of the area to be sprayed and, if possible, at the end of each spray run (pass). (See Section 5.4.10 for safety precautions for flag marker teams.)

If DGPS equipment is available in the aircraft, there is no need for flag marking, but target boundaries may still need to be marked by ground teams.

5.4.8 Assessment of kill

Assessing the effectiveness of the control operation can be undertaken at each target site at various times after spraying (3, 6, 12 and 24 h if possible). Assessment requires taking a number of quadrat samples (see Section 5.2.5). In each quadrat the ratio of dead larvae to the sum of those alive plus dead gives the proportion killed. Thus:

$$\textbf{percentage kill} = \frac{\textbf{dead} \times \textbf{100}}{\textbf{alive + dead}} \qquad \text{(Formula 7)}$$

This will give a good idea of the efficacy of the pesticide, assuming that the correct application technique has been used. Where the result is widely different from that expected, spraying method, pesticide used, application rates and weather conditions all need to be checked.

5.4.9 Environmental considerations

The application of pesticides is potentially damaging to the environment both terrestrial, aerial and aquatic. It is, therefore, the responsibility of all field officers to **ensure that every possible precaution is taken to avoid contamination**. Spray operators should be aware of the potential danger not only to humans and animals, but also to standing water, streams and rivers, water catchment areas, grazing land, to bees and other beneficial insects such as parasitoids and predators, and to non-target species such as birds, fish, crustacea and other wildlife.

Detailed planning for control operations should also include eco-toxicological monitoring both before and after the treatment to identify any environmental impacts. Techniques are well described in the literature for use in the field and advice on this important subject can be obtained from NRI (see Appendix 5).

Key references: Brown and Odiyo (1968)
Brown, Stower *et al*. (1970)
Page and Dewhurst (1987, 1992)
Southwood (1978)
Yeates (1973a, 1973b)

5.4.10 Safe use of pesticides – operator safety

Hazards. Insecticides (chemical and biological) are, by definition, poisonous to insects. They can also, if used carelessly, kill or poison people and non-target organisms including domestic and wild animals, birds, fish or beneficial insects, and damage the environment (see Section 5.4.9).

Time-expired pesticides should never be used.

Some insecticides (organophosphates and carbamates) inhibit the formation of an enzyme in the blood of mammals known as

Table 11. Hazard classification of insecticides used for control of armyworm

Insecticide	Oral LD50	Dermal LD50 (mg/kg)	WHO hazard (mg/kg)	Formulation % a.i. class	Liquid/ solid	Formulation hazard oral	Formulation hazard dermal	Comments
Organophosphates								
Malathion	1200	4100	III	50%	liquid	III	III	
				95%	liquid	III	III	
Fenitrothion	503	890	II	50%	liquid	II	III	
				96%	liquid	II	III	
Trichlorphon	560	>2000	III	50%	liquid	III	III	Mutagen
Phoxim	1976	1975	II	50%	liquid	III	III	
Chlorphyriphos	135	2000	II	48%	liquid	II	III	Eye irritant
				24%	liquid	II	III	
Quinalphos	62	1250	II	25%	liquid	II	III	
				30%	liquid	II	III	
Tetrachlorvinphos	4000	>5000	III	24%	liquid	III	III	
				35%	liquid	III	III	
Pirimiphos methyl	2018	>2000	III	25%	liquid	III	III	Skin and eye
				50%	liquid	III	III	irritant; mutagen

Carbamates								
Carbaryl	300	>4000	II	85%	solid	II	III	Mutagen
				25%	liquid	II	III	
Synthetic pyrethroids								
Cypermethrin	>4000		III	5%	liquid	III	III	Skin and eye irritant
				2.5%	liquid	III	III	
				6%	liquid	III	III	
Deltamethrin	>2200		III	2.5%	liquid	III	III	Eye irritant
				0.5%	liquid	III	III	
Fenvalerate	3200	>5000	III	7%	liquid	III	III	Skin and eye irritant
				3%	liquid	III	III	
Permethrin	4000		III	10%	liquid	III	III	
				4%	liquid	III	III	

II = moderately hazardous; III = slightly hazardous.

cholinesterase. This enzyme controls the formation of acetylcholine, an impulse transmitter substance in the nervous system. Blood cholinesterase level is a useful indicator of the degree of exposure to such insecticides. As the level can be depressed even in the complete absence of external symptoms of poisoning, the level of cholinesterase in the blood should be checked in users of organophosphorus and carbamate insecticides before exposure and checked again at regular intervals. This is particularly important for operators who are engaged in prolonged spray operations and for storemen who handle or decant pesticides regularly.

Synthetic pyrethroids also are dangerous if handled carelessly; they affect the nervous system.

Always read the pesticide label before starting to handle pesticides, and follow the instructions carefully.

Table 11 shows the hazard classification of various insecticides. Those listed as effective for spraying armyworm are classified as class III (lowest risk) with respect to dermal contact (through the skin), and most are also class III for accidental oral ingestion (by mouth). Nevertheless, it must be remembered that **all** pesticides are toxic and some may even be mutagenic. Even the solvent or oil-based carriers may be harmful, so that contact without sufficient protective clothing should be avoided.

Protection. Always wear protective clothing when handling or applying pesticide. In the tropics, gloves, overalls, respirators, face masks and goggles (the recommended range of protective clothing) may be uncomfortable and unpleasant in hot and humid conditions. An uncomfortable spray operator can become a dangerous or careless user unless rest periods are taken during prolonged spraying activities. The selection of protective clothing can be made with reference to the degree of protection required for safe practice and the comfort of the wearer.

All protective clothing must be free from holes, and made from materials which should be as light as possible. Note that gloves and overalls are needed for **all** the pesticides recommended for armyworm control. A general guide to clothing recommended for handling and spraying pesticides is given in Table 12.

- **Overalls** made of cotton fabric are preferable, but a long-sleeved shirt and long trousers (without turn-ups) may be sufficient. All clothing should be washed thoroughly after use, and kept separate from other items being washed. If they do have to be washed at the same time as other clothing, they should be put through the water last, to avoid contaminating ordinary clothes. Clothing used in control operations should be hung in full sunshine to dry so that the UV light can speed up the breakdown of any remaining pesticide, and should be stored separately from everyday clothing.

Table 12. Minimum protective clothing requirements when using insecticides

Activity	**Protective clothing**	**Insecticide classification***			
		Class Ia	**Class Ib**	**Class II**	**Class III**
Handling and mixing concentrate	Overalls	+	+	+	+
	Gloves (neoprene)	+	+	+	+
	Boots	+	+	+	
	Face shield/eye protection	+	+	(+)	
	Apron (neoprene)	+	+		
	Respirator/face mask	+	+		
Hand spraying	Overalls	+	+	+	+
	Gloves (neoprene)	+	+	+	
	Hat	+	+	+	
	Boots	+	+		
	Face shield/eye protection	+	+		
	Respirator/face mask	+	(+)		

* Insecticide classification:

Ia = extremely hazardous
Ib = highly hazardous
II = moderately hazardous
III = slightly hazardous
\+ = requirement
(+) = recommendation

- **Gloves** should be worn, particularly when decanting pesticides. They should be made of nitrile or some other material that is not damaged by pesticides or their carrier (solvents) and is impervious. Heavy-duty PVC gloves may be used as an alternative in the absence of nitrile. **Do not wear cotton or leather gloves**.
- **Boots** or **shoes** (not sandals) should always be worn. It is particularly important to wear rubber boots when decanting pesticides and to have the overall or trouser legs outside the boots (not tucked in) so that if there is spillage pesticide cannot enter the top of the boot.
- **Goggles** or **protective glasses** should be worn, if available, during control operations.
- A **hat** should also be worn to help reduce contamination from pesticide during spraying operations.

To prevent pesticide trickling inside clothing, cuffs of overalls or shirt sleeves should be worn outside the gloves if the spray head is held low. The sleeve should be tucked inside the long sleeve of the glove when the sprayer is to be held above waist level.

If dust or fine powder formulations are used, always wear a respirator or dust mask. A handkerchief or cloth tied to cover the nose and mouth will reduce inhalation of dust if a dust mask is not available.

Precautions. The above precautions apply equally to storemen, operators and flag marker teams involved in both aerial and ground spraying operations.

Where protective clothing is not available, only use insecticides in the **lowest** hazard categories.

Always have plenty of clean water and soap available near the site where pesticide is handled and always wash thoroughly after handling or using pesticides.

- Pesticides should **never** be transported in open or leaky containers.
- Pesticides should **never** be transported with foodstuffs.

5.4.11 Safe storage of pesticides

Any store used for storing pesticides should be purpose built and used solely for that purpose. It should be located on a site away from water sources and other buildings. The site should be well drained, not subject to seasonal or other flooding, and preferably be in a shaded situation to keep store temperature down (high temperatures reduce the shelf-life of some pesticides).

There should be easy access for delivery vehicles and emergency services.

Items such as food, drink, clothing or fuel should **never** be stored with any pesticide, to avoid contamination and fire hazard.

A permanent pesticide store should be operated so that materials are issued on a first-in, first-out basis. This will prevent the build-up of stocks of old, out-of-date products that will present a disposal problem and represent money wasted. The date of receipt should be written on the labels of new stocks as they arrive. Labels should not be allowed to rub, drop off, or fade to illegibility. **Any labels that become torn, stained or otherwise difficult to read should be replaced with new, clearly written ones.**

The **quantity of pesticide** and date received, storage notes (shelf-life, etc.), the date and quantity issued and the balance in stock should be recorded in a ledger, and kept up to date.

All **pesticides** have a recommended shelf-life and methods of storage should be designed to prolong this as far as possible. Shelf-life rapidly declines once a container has been opened. Disposal of outdated, or unusable, insecticides presents many problems, both logistical and financial. These can be avoided by carefully following recommended procedures and manufacturer's guidelines.

Packages, bottles and drums should be carefully stacked so that they cannot fall and become damaged or cause spillage. Stacks should be kept to an easily accessible height.

Pesticide containers should be stored off the floor, on pallets, timber, bricks or shelving where appropriate, so that any leaks are easily seen, and corrosion caused by damp floors, or leaking pesticide, is minimized.

Stores must be kept clean and tidy, with the floors clear of debris. There should be adequate gangways between stacks for easy access and circulation of fresh air.

Always wear gloves, boots, overalls and preferably, goggles or protective glasses, when handling or decanting pesticides.

5.4.12 Safe disposal of pesticides

Do not attempt to recover spilled pesticide for subsequent use, as the debris inevitably entrained will damage the application equipment. There are three important steps to dealing with a spillage: **clear up; clean up; disposal**.

Clear up. Liquid pesticide spills should be covered with suitable absorbent material such as sawdust, sand, or soil, which should then be swept up and placed in a clearly labelled container for disposal. Supplies of containers and absorbent material should always be kept in the store.

For solid spills, to avoid stirring up dust, add damp sand or sawdust to the spill and sweep up carefully. The waste should then be disposed of (see below).

- **Wear full protective clothing when dealing with pesticide spills.**

Clean up. The affected area should be scrubbed with detergent, or strong soap and water, not hosed down, as this merely disperses the spill. Dispose of the contaminated cleaning water in the same way as the pesticide (see below).

Disposal. A practical method of disposal of a few litres or kilograms of pesticide is by burying it, preferably at least 0.5 m deep. Burial sites should be permanently fenced and marked. In choosing a burial site, which should be well away from watercourses or dams, take care to avoid problems with public health and environmental contamination, especially of the water table.

Cardboard or paper packaging should be burned, again choose a site well away from habitation.

Empty pesticide containers must never be used for domestic or agricultural purposes. If they are not going to be recycled for insecticide use, they should be perforated and buried.

5.4.13 Code of conduct for pesticide users

(a) **Mixing, decanting and applying pesticides**

1. Wear appropriate protective clothing.
2. Have plenty of soap and water immediately available for washing.
3. Read the instructions on the label and follow them.
4. Avoid contamination by pouring liquids carefully without splashing, and by transferring powders without spillage or puffing them up into the face, or over the hands and arms. Transfer powders and dusts only where it is sheltered from the wind.
5. Avoid inhaling toxic dusts and vapours by working in the open air as much as possible. Take care not to let dust blow about in the wind.
6. Never eat, drink or smoke and avoid touching the eyes, mouth or any broken skin when handling pesticides.
7. Never work alone when handling very toxic substances.
8. Keep unauthorized people (especially children) and animals away from pesticides.
9. Never leave opened containers of pesticide unattended.

10. Do not apply pesticides if weather conditions (wind and/or temperature), time of day or season are unsuitable for spraying.
11. Do not spray near water (streams, ponds or dams).
12. Ensure that operators are adequately instructed and supervised and have sufficient rest periods between spray sorties.
13. Only use pesticides in strict accordance with instructions or approved recommendations.
14. Never blow out blocked nozzles or holes by mouth (use a bicycle pump).
15. If an operator, through accident or misuse, does become contaminated, immediately take him away from the pesticide spillage, remove his contaminated clothing and make sure the whole body is washed thoroughly with soap and water, and that clean clothing is put on.
16. If anyone becomes contaminated with pesticide, seek medical advice as soon as possible, giving the doctor full details which are described on the label of the pesticide container.
17. During the spray operation and for as long as recommended afterwards, keep people and animals out of the area being treated.
18. Have all regular users of insecticides checked regularly for blood cholinesterase levels.
19. Do not continue to work or remain in contact with pesticides if blood tests show that your cholinesterase level is below normal.
20. Always wash thoroughly after handling pesticides.

(b) **After applying pesticides**

1. Unused pesticides must be removed from spray equipment, and all equipment cleaned thoroughly

with a suitable solvent (e.g. water for e.c. and diesel or paraffin for ULV) before returning it to the store.

2. Return unused pesticides to the store and keep them locked away from unauthorized people and out of reach of children.
3. **Never** transfer pesticides into bottles used for drinks (e.g. soda, beer bottles or edible oil cans/drums).
4. Make all empty containers unusable (pierce those which are plastic and metal), and safely dispose of them by burying or burning. Keep well clear of any smoke. It is impossible to clean a pesticide container well enough to make it safe for storing food or water, or for use as a cooking vessel or for distilling alcoholic beverages.
5. Clear up any spillage as soon as possible.
6. Remove and wash protective clothing.
7. Wash thoroughly and put on clean clothing.
8. Keep an accurate record of pesticide usage, including details of workers and the number of hours each was exposed to pesticide during the operation.
9. Prevent people entering sprayed areas until it is safe to do so (see Table 9).

Key reference: Anon (1982a)

5.5 Logistics and responsibilities

5.5.1 Planning

Detailed forward planning at the beginning of each armyworm season is essential for successful control operations. As far as possible responsibilities should be clearly defined at national and regional levels. To help with this, the various responsibilities have been identified in Table 13, which gives a checklist of logistical requirements for control operations both before the outbreak season and once it begins.

Table 13. Checklist of logistic requirements for control operations and suggested areas of responsibility.

Before the armyworm season begins:

DLCO-EA (Region 1)

Supply pheromone septa to National Plant Protection Services (NPPS)

Check supplies of outbreak cards distributed

NPPS

Inspect all traps, repair and replace as necessary to ensure they are all working

Supply pheromone septa to trap operators

Supply spares for light traps as necessary

Ensure moth catch recording forms and instructions are issued to trap operators

Ensure insecticide (Vapona, Nogos, etc.) is available for the traps

Plan and undertake trap operator training courses

Provide outbreak recording cards (pre-paid postcards) to DAOs

NAC and **DAO**

Arrange finance for postage, telephone, telegrams and faxsimile (if available)

Ensure that vehicles are serviceable and that money is available for fuel and personnel

Ensure spray equipment is functional and a sufficient supply of batteries is available for ULV sprayers

Ensure suitable stocks of recommended insecticides are available nationally and locally

Warn farmers of impending outbreaks and alert commercial suppliers of insecticide in advance of outbreak occurrence

Once outbreaks are forecast or have begun or high moth numbers are caught locally:

DAO

Warn farmers of impending outbreaks

Undertake field surveys to look for eggs and young larvae

Check identification of specimens collected from all outbreaks

Assess potential severity of each outbreak both for immediate crop and for subsequent developments

Table 13 *cont.*

Report outbreaks to NAC, listing dates first seen, crops, areas and density of larvae within infestations, the amount of damage, and control measures taken
Ensure suitably trained personnel are available for marking (flagging) outbreak areas to be sprayed
NAC
Alert regional organization if control assistance is required (DLCO-EA in Region 1)
Initiate an assessment of crop losses and costs of control in collaboration with an agricultural economist or farm management specialist
Check and record effectiveness of control operations in conjunction with DAO

DAO = District Agricultural Officer, senior Crop Protection Officer or equivalent. agricultural staff at the District level.
NAC = National Armyworm Co-ordinator.
NPPS = National Plant Protection Services.

5.5.2 Strategic control – limiting the spread of outbreaks

Essential requirements for efficient strategic control are:

- efficient planning and organization of logistical requirements (finance, equipment, personnel);
- early season monitoring and sampling of armyworm moth and larval populations;
- the rapid receipt and assimilation of information as well as distribution of warnings;
- the rapid, well co-ordinated start of control operations.

This requires close collaboration between all parties concerned (DAOs, NPPS and the regional co-ordinating centre).

The methods needed for monitoring, survey, forecasting and control are similar to those needed for crop protection, however, monitoring and control preparations and related activities are intensified in the main primary outbreak areas (see Figure 22 for those known in eastern

Africa). Vigilance must be maintained for those outbreaks recognized as potentially critical with regard to the spread of armyworm during the rest of the season. For eastern Africa it is the primary outbreak areas in Kenya and Tanzania that are the most important centres for the spread of outbreaks throughout the northern migration route, following the dominant winds.

Monitoring. In eastern Africa, in September or early October, before the outbreak season starts, a network of large numbers of pheromone traps is placed in the primary outbreak areas. The traps should have clear plastic bases so that moth catches can be seen easily without having to open up the trap. These traps should be monitored daily throughout the season for sudden increases in moth catches (or numbers of 30 or more moths in a night). Such increases in trap catches often coincide with the first rainstorms of the season in these areas. High catches should be reported immediately to the local agricultural officer who will then inform the DAO (or their equivalent) and the NAC. The trap operator (who is usually an agricultural officer) and the district agricultural office must instigate larval searches **5–7 days** after the first high moth numbers in any local trap, whether or not a warning has been received from the armyworm forecasting office.

Exchange and collection of information. The DAOs offices and NPPS will receive reports from all the traps covered by them. The national co-ordinators are, therefore, able to pin-point areas which will be priorities for surveys and if necessary control of outbreaks.

Information from the Meteosat satellite is particularly valuable for detecting the first storms of the season likely to concentrate moths. This information is passed immediately by DLCO-EA to the national armyworm co-ordinators for use in preparing forecasts.

Once prepared, forecasts are passed to the plant protection services by radio through DLCO-EA base liaison staff, or directly by fax, telex, telephone or other available route. This is **urgent**, as the information is needed quickly to initiate logistical requirements of control operations. Information is also passed weekly as a routine service to all operational bases of the regional centres (DLCO-EA, IRLCO-CSA).

Control. Once outbreaks have been identified and the areas estimated, control is initially effected locally and then with support from the DAOs, NPPS and even the regional organizations, if requested.

Priority for control must be given to the largest, densest and oldest outbreaks as the purpose of control is to reduce as much as possible the numbers of moths emerging. This will reduce the chances of large numbers of moths escaping to become reconcentrated, thus reducing the spread of outbreaks downwind.

Strategic control must be carried out on rangeland, as well as on crops.

Key references: Cheke and Tucker (1995)
Latigo *et al*. (1990)
Page and Dewhurst (1987, 1992)
Rose (1978a)

6

WORKSHOPS AND TRAINING

Regular regional exchange of information, to encourage discussion between scientists, field personnel and administrators, is very important:

(i) at the decision-making level, at workshops discussing responsibilities, recommendations and developments;

(ii) with national armyworm co-ordinators from all countries in the region in order to bring them up to date with current concepts and generally to foster closer co-operation.

Training control personnel and trap operators is required every year in District Offices, at formal courses, or on site. Additional training courses, or workshops, should be arranged as the needs are identified (e.g. management, forecast methods).

The continual updating of training aids, such as booklets, leaflets, posters, life cycle charts (produced in national languages), colour slides and if possible the video series, is strongly recommended. The DLCO-EA is able to supply examples of training booklets, slides and videos which may need to be modified for local conditions. The armyworm booklet, giving detailed information on biology, ecology and control of armyworm is also available, free of charge, from DLCO-EA.

Key references: Anon (1982a)
Page and Dewhurst (1987, 1992)

7

COMPREHENSIVE LIST OF REFERENCES TO *SPODOPTERA EXEMPTA*

The literature on armyworm has been published in various ways and in many scattered sources. There are two existing bibliographies on African armyworm, one by E. S. Brown (1962) and the other by O. Mochida and T. Okada (1974). The list that follows includes literature additional to that in these sources. Some references are unseen by the compilers and are regrettably incomplete as a consequence. We have also included titles of papers currently in preparation for publication and currently unpublished in order to be as up to date as possible.

Key works are indicated in **bold typeface**. Also included are some useful general references.

Information may be obtained from The Director, Desert Locust Control Organization for Eastern Africa, PO Box 4255, Addis Ababa, Ethiopia, (dlc@telecom.net.et) or the Librarian, University of Greenwich, Central Avenue, Chatham Maritime, Kent ME4 4TB, UK (web: http://www.gre.ac.uk).

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8

APPENDICES

Appendix 1 Identification of the moths of *Spodoptera* species occurring in Africa and Madagascar

I **External characters** (See Figures 1e and 5).

1 Frenulum a single bristle (males): ...2

\- Frenulum normally two or three bristles (females): ...8

2 Antennae dentate:

Forewing pale greyish-brown, with orbicular spot circular; reniform spot more pronounced than general wing colour; hindwings without infuscation or dark venation; fore-tarsal segments black with white tips; a small rather rounded species: ...*Spodoptera cilium*

\- Antennae simple: ...3 or 4

3 Forewing dark, orbicular spot elongated at base and nearly touching reniform spot, which is usually paler than general wing colour. Hindwings white with veins not or scarcely infuscate. Fore-tarsal segments whitish. Wing span 28–35 mm. Seldom caught, except in Ethiopia: ...*Spodoptera apertura*

\- [Forewing similar to above]; orbicular and reniform spots not so close basally; hindwings more or less entirely infuscate. Fore-tarsal segments not white. Wing span 28–32 mm. Only recorded from the Madagascar: ...*Spodoptera malagasy*

4 Forewing with reniform spot black and pronounced; orbicular spot circular. Fore-tibia with a large mass of hair-scales of which part reaches or extends beyond the second tarsal segment; middle tibia with outer spur subequal to scale-free basal part of tibia: ...5

Forewing markings not as above. Fore-tibiae with a tuft of hair-scales not, or scarcely extending beyond the base of the first tarsal segment; middle tibia with outer spur less than half as long as the inner spur; outer spur with scale-free area extending beyond the middle: ...6

5 Forewing pointed apically, typically with contrasting light and dark markings; postmedial and subterminal fasciae widely separated; hindwings with some infuscation on the veins, and the wing membrane often infuscate at the edges. Fore-tibia with tuft of hair-scales extending to the tip of the tarsus; a robust species; wing span 28–43 mm, more robust than the following species. Typically recorded from coastal areas, especially in Tanzania: ...*Spodoptera mauritia*

\- Forewing rounded apically; more unicolorous; post-medial and sub-terminal fasciae not widely spaced. Hindwing veins white, not or only scarcely infuscate. Fore-tibia with tuft of hair-scales extending only to tip of second tarsal segment. Wing span 27–40 mm. A widespread, common species (sometimes attracted to *S. exempta* pheromone-baited traps): ...*Spodoptera triturata*

6 Slight, slender-bodied species; forewing unicolorous, pale greyish-brown; orbicular spot circular, pronounced, reniform spot also pale. Hindwing with veins strongly infuscate throughout; wing span 24–31 mm. Common and widespread, sometimes a pest: ...*Spodoptera exigua*

\- Larger, more robust species; forewing variegated; orbicular spot diagonally elongate, almost arrow-shaped; hindwing veins usually not infuscate: ...7

7 Forewing strongly variegated, with characteristic pattern of light and dark markings; orbicular spot open distally and continuing as a pale stripe as far as the postmedial fascia; reniform spot also elongated, and produced apically to a sharp point. Hindwing white, veins typically not infuscate. Fore-tarsal segments pale. Wing span 30–41mm. Common: ...*Spodoptera littoralis*

\- Forewing less strongly variegated; orbicular spot closed, often markedly pale yellow; reniform spot kidney or arrow-shaped; hindwing veins usually infuscate, especially distally. Fore-tarsal segments black with white tips. Wing span 29–32 mm. Seasonally very common, widespread and important pest of cereals and grasses. Larvae rarely found on dicotylendonous plants, never on trees: ...*Spodoptera exempta*

8 Orbicular spot diagonally elongate, or if not, then hair-scales inside the tip of the abdomen black: ...9

\- Orbicular spot round, or nearly so. Hair-scales inside the tip of the abdomen brown or whitish. (Fore-tarsal segments black with white tips): ...12

9 Forewing homogeneous dark brown/black; orbicular spot closed and rounded distally; hind-wings more or less infuscate distad; reniform spot indistinct. Fore-tarsal segments black with white tips. Hair-scales inside the tip of the abdomen black (only found in this species). Wing span 27–38 mm. Often caught in large numbers: ...*Spodoptera exempta*

\- Forewing somewhat variegated, with pale markings; orbicular spot expanded distally. Hindwing veins white, scarcely any infuscation. Fore-tarsal segments whitish. Inner hair-scales at the tip of the abdomen not black: ...10

10 A large species; forewing paler with characteristic pattern, all veins more or less whitish; white line along posterior margin of apical cell continued to base of wing; reniform spot with apex pointed, often with a white line running through it. Inner hair-

scales at the tip of the abdomen orange-brown. Wing span 31–42 mm (cf. couplet 8): ...*Spodoptera littoralis*

- Forewing dark, other characters not as above: ...11

11 Forewing dark brown or black; white markings confined to the central area; reniform spot more rounded; posterior margin of wing without diagonal white line. Hindwing veins not, or scarcely infuscate. Fore-tarsal segments whitish; inner hair-scales at the tip of the abdomen brown. Wing span 28–35 mm. (Seldom found in trap catches): ...*Spodoptera apertura*

- Forewing dark (not as dark as 12); hindwings more or less entirely infuscate. Basal antennal teeth conical. Fore-tarsal segments not white. Inner hair-scales at the tip of the abdomen orange-brown, sometimes forming dark clumps. Wing span 28–32 mm. Only reported from Malagasy Republic:
...*Spodoptera malagasy*

12 Small, slight slender-bodied species. Forewing with reniform spot concolorous with and never darker than general wing colour. Hindwings with veins strongly infuscate. Inner hair-scales at the tip of the abdomen white. Wing span 24–31 mm:
...*Spodoptera exigua*

- Larger, more robust species, or, if smaller, then hindwing veins not infuscate. Reniform spot sometimes concolorous but typically darker (square and black) than the general wing colour. Abdominal hair-scales not white. Wing span with larger range than above: ...13

13 Small species; antennae dentate; forewings unicolorous; hind-wing veins not infuscate. Fore-tarsal segments black with white tip. Inner hair-scales at the tip of the abdomen pale, but not white. Wing span 25–35 mm: ...*Spodoptera cilium*

- Larger species, very variable in size. Fore-tarsal segments not bicoloured. Inner hair-scales at the tip of the abdomen brown. Wing span 28–51 mm: ...14

14 Forewing tip pointed slightly; postmedial and sub-terminal fasciae widely spaced. Hindwing often infuscate at margin, veins more or less infuscate. Wing span 28–43 mm. A coastal species, commonly recorded from Tanzania coastal traps (may be confused with *S. triturata*): ...*Spodoptera mauritia*

\- Forewing tip more rounded; postmedial and sub-terminal fasciae closer together. Hind-wings white, with little or no infuscation. Wing span 27–40 mm. A common species though rarely found in large numbers: .... *Spodoptera triturata*

II Genitalia (See Figure 1b)

A Males (after Brown and Dewhurst (1975), with modifications)

1 Uncus short, peg-like; (valve long and thin): ...*Spodoptera malagasy*

\- Uncus medium or long, straight or slightly curved: ...2

2 Uncus more or less straight and short, evenly tapered to a point. (Valve indented near apex, aedeagus with ill-defined blunt cornutus): ...*Spodoptera apertura*

\- Uncus evenly tapered to tip, and variably thickened: ...3

3 Valve broad and excised ventrally (laterally on a mounted preparation); cornutus of aedeagus obtuse, rounded; juxta broadly ‘H’-shaped with convex apex (cf. oriental species *S. litura*, where juxta is narrowly triangular): ...*Spodoptera littoralis*

\- Valve not excised, or if excised then valve narrow: ...4

4 Clasper small, sharply curved in the middle; aedeagus with cornutus of numerous small spines: ...*Spodoptera exempta*

\- Clasper elongated, sharply curved near its tip: ...5

5 Aedeagus with cornutus single and pointed; (valve broad): ...*Spodoptera exigua*

- Valve broad, deeply incised near apex: ...6

6 Clasper with two sharp spines. Aedeagus with needle-like downward-pointing cornutus: ...*Spodoptera cilium*

- Valve narrow, uncus tapering abruptly at its tip. Aedeagus not as above: ...7

7 Valve with ventral margin excised near apex. Aedeagus with cornutus asymmetrically tapered to a sharp point: ...*Spodoptera triturata*

- Valve with ventral margin excised such that the part beyond the excision is almost as broad as long. Aedeagus with cornutus obtuse and armed with several teeth: ...*Spodoptera mauritia*

B Females (See Figure 1c)

1 Bursa copulatrix with long vertical or transverse signum: ...2

- Bursa copulatrix with short or medium transverse signum: ...6

2 Bursa copulatrix with signum transverse to the axis and of medium length: ...*Spodoptera cilium*

- Bursa copulatrix with signum peripheral or on its face: ...3

3 Signum long and following periphery of convex side of bursa copulatrix: ...*Spodoptera exigua*

- Signum not as above: ...4

4. Diverticulum rounded, ostium bursae much shorter than ductus bursae: ...*Spodoptera exempta*

- Diverticulum angled distad. Ostium bursae about the same length as ductus bursae: ...5

5. Ductus bursae tending to be bulbous, basal part of diverticulum, with weak striations, distal part of ductus bursae barely constricted at junction with ostium bursae: ...*Spodoptera malagasy*

- Ductus bursae with concavity above diverticulum, the latter lacking striations and noticeably constricted distally at junction of ostium bursae. (Signum vertical and at edge of the bursa copulatrix): ...*Spodoptera apertura*

6 Bursa copulatrix strongly dilated, balloon-like. Signum as broad as long, often with other thickenings. Ostium bursae significantly broader than ductus bursae at their junction. Diverticulum very much reduced: ...*Spodoptera mauritia*

- Bursa copulatrix with strong striations on signum and diverticulum: ...7

7 Diverticulum pointed on one side only: ...8

- Diverticulum with small lobe on opposite side. Ductus bursae and ostium bursae tapering gently towards their junction: ...*Spodoptera littoralis*

8. Diverticulum with 'A' shaped thickenings and folds. Bursa copulatrix tapering evenly. Ductus bursae elongated: ...*Spodoptera triturata*

- Diverticulum very acute, ductus bursae very long and thin, wider than ostium bursae at their junction. [NB: this species, does not occur in Africa.] ...*Spodoptera litura*

Key reference: Brown and Dewhurst (1975)

Appendix 2 Grasses, sedges and other plants recorded as eaten by *Spodoptera exempta* larvae

FTEA = Flora of Tropical East Africa, published under the authority of the Minister for Overseas Development by the Crown Agents for Overseas Governments and Administrations. The '**G**' refers to the volumes on the Gramineae, and the postfix numbers refer to the particular part and page, e.g.

FTEA G3: 316 = **F**lora of **T**ropical **E**ast **A**frica: **G**ramineae, Part **3**, page **316**.

Synonyms have not been included.

I Wild Gramineae

Acroceras macrum Stapf	FTEA G3: 565
Alloteropsis semialata (R.Br.) Hitchc.	FTEA G3: 616
Andropogon abyssinicus Fresen.	FTEA G3: 772
A. amethystinus Steud.	FTEA G3: 772
A. amplectens Nees.	FTEA G3: 784
A. canaliculatus Schumach.	FTEA G3: 782
A. chinensis (Nees) Merr.	FTEA G3: 779
A. chrysostachyus Steud.	FTEA G3: 773
A. distachyos L.	FTEA G3: 770
Anthephora pubescens Nees	FTEA G3: 664
Aristida adoënsis Hochst.	FTEA G1: 144
Arthraxon prionodes (Steud.) Dandy	FTEA G3: 741
Arundinella nepalensis Trin.	FTEA G2: 409
Bothriochloa bladhii (Retz.) S.T. Blake	FTEA G3: 719
Brachiaria brizantha (A. Rich.) Stapf	FTEA G3: 587
B. decumbens Stapf	FTEA G3: 586
B. dictyoneura (Fig. & De Not.) Stapf	FTEA G3: 582
B. humidicola (Rendle) Schweik.	FTEA G3: 583
B. insculpta (A. Rich.) A. Camus	FTEA G3: 720
B. jubata (Fig. & De Not.) Stapf	FTEA G3: 580
B. lachnantha (Hochst.) Stapf	FTEA G3: 589
B. platynota (K. Schum.) Robyns	FTEA G3: 579
B. serrata (Thunb.) Stapf	FTEA G3: 579
B. radicans (Lehm.) A. Camus	FTEA G3: 721
Bromus leptoclados Nees	FTEA G1: 68

Capillipedium parviflorum (R.Br.) Stapf	FTEA G3: 718
Cenchrus ciliaris L.	FTEA G3: 691
C. echinatus L.	FTEA G3: 695
C. setigerus Vahl	FTEA G3: 694
Chloris amethystea Hochst.	FTEA G2: 343
Chloris gayana Kunth.	FTEA G2: 346
C. mossambicensis K. Schum.	FTEA G2: 341
C. pycnothrix Trin.	FTEA G2: 340
C. roxburghiana Schult.	FTEA G2: 339
Chrysochloa orientalis (C.E.Hubbard) Swallen	FTEA G3: 329
Chrysopogon serrulatus Trin.	FTEA G3: 736
Cymbopogon caesius (Hook & Arn.) Stapf	FTEA G3: 761
C. giganteus Chiov.	FTEA G3: 763
C. nardus (L.)	FTEA G3: 764
C. pospischilii (K. Schum. Rendle) C.E. Hubbard	FTEA G3: 765
Cynodon dactylon (L.) Pers.	FTEA G2: 318
C. plectostachyus (K. Schum.) Pilg.	FTEA G2: 318
Cypholepis yemenica (Schweinf.) Chiov.	FTEA G2: 248
Dichanthium annulatum (Forssk.) Stapf	FTEA G3: 725
Digitaria abyssinica (A.Rich.) Stapf	FTEA G3: 641
D. diagonalis (Nees) Stapf	FTEA G3: 624
D. gazensis Rendle	FTEA G3: 643
D. maitlandii Stapf & Hubbard	FTEA G3: 629
D. milanjiana (Rendle) Stapf	FTEA G3: 647
Diplachne caudata K. Schum.	FTEA G2: 284
Echinochloa haploclada (Stapf) Stapf	FTEA G3: 560
E. pyramidalis (Lam.) Hitchc. & Chase	FTEA G3: 561
Eleusine floccifolia (Forssk.) Spreng.	FTEA G2: 267
E. jaegeri Pilger	FTEA G2: 264
Elionurus muticus (Spreng.) Kuntze	FTEA G3: 837
Enteropogon macrostachyus (A. Rich.) Benth.	FTEA G2: 332
E. rupestris (J.A. Schmidt) A. Chev.	FTEA G2: 332
Entolasia imbricata Stapf	FTEA G3: 573
Eragrostis sp.	FTEA G2: 188
E. braunii Schweinf.	FTEA G2: 227
E. caespitosa Chiov.	FTEA G2: 203
E. capensis (Thunb.) Trin.	FTEA G2: 221
E. chapelieri (Kunth) Nees	FTEA G2: 225
E. exasperata Peter	FTEA G2: 220
E. olivacea K. Schum.	FTEA G2: 201
E. paniciformis (A. Br.) Steud.	FTEA G2: 219
E. racemosa (Thunb.) Steud.	FTEA G2 230
E. superba Peyr.	FTEA G2: 211
E. tenuifolia (A.Rich.) Steud.	FTEA G2: 238
Eriochloa meyerana (Nees) Pilg.	FTEA G3: 569
Eulalia aurea (Bory) Kunth	FTEA G3: 713

E. polyneura (Pilger) Stapf	FTEA G3: 715
Eustachys paspaloides (Vahl) Lanza & Mattei	FTEA G2: 335
Exotheca abyssinica (A. Rich.) Anderss.	FTEA G3: 821
Harpachne bogdanii Kennedy-O'Byrne	FTEA G2: 272
H. schimperi A. Rich.	FTEA G2: 270
Heteropogon contortus (L.) Roem. & Schult.	FTEA G3: 827
Hyparrhenia diplandra (Hack.) Stapf	FTEA G3: 818
H. filipendula (Hochst.) Stapf	FTEA G3: 803
H. hirta (L.) Stapf	FTEA G3: 798
H. papillipes (A.Rich.) Stapf	FTEA G3: 808
H. pilgerana C.E. Hubbard	FTEA G3: 807
H. rufa (Nees) Stapf	FTEA G3: 794
Hypertheta dissoluta (Steud.) W.D. Clayton	FTEA G3: 786
Ischaemum afrum (J.F. Gmel.) Dandy	FTEA G3: 747
Leptochloa obtusiflora Hochst.	FTEA G2: 278
L. rupestris C.E. Hubbard	FTEA G2: 277
Leptothrium senegalense (Kunth) W.D. Clayton	FTEA G2: 402
Lintonia nutans Stapf	FTEA G2: 302
Loudetia arundinacea (A. Rich.) Steud.	FTEA G2: 417
L. flavida (Stapf) C.E. Hubbard	FTEA G2: 416
L. kagerensis (K. Schum.) Hutch.	FTEA G2: 420
L. phragmitoides (Peter) C.E. Hubbard	FTEA G2: 415
L. simplex (Nees) C.E. Hubbard	FTEA G2: 418
Melinis minutiflora P. Beauv.	FTEA G3: 506
Miscanthus violaceus (K. Schum.) Pilg.	FTEA G3: 704
Panicum coloratum L.	FTEA G3: 485
P. deustum Thunb.	FTEA G3: 468
P. dregeanum Nees	FTEA G3: 478
P. infestum Peters	FTEA G3: 472
P. massaiense Mez	FTEA G3: 480
P. maximum Jacq.	FTEA G3: 471
P. trichlocadum K. Schum.	FTEA G3: 473
Paspalum auriculatum Presl.	FTEA G3: 609
P. dilatatum Poir.	FTEA G3: 608
P. scrobiculatum L.	FTEA G3: 610
Pennisetum clandestinum Chiov.	FTEA G3: 675
P. dowsonii Stapf & Hubbard	FTEA G3: 686
P. hohenackeri Steud.	FTEA G3: 685
P. macrourum Trin.	FTEA G3: 689
P. massaicum Stapf	FTEA G3: 687
P. mezianum Leeke	FTEA G3: 686
P. polystachion (L.) Schult.	FTEA G3: 679
P. procerum (Stapf) W. D. Clayton	FTEA G3: 682

P. purpureum Schumach.	FTEA G3: 677
P. setaceum (Forssk.) Chiov.	FTEA G3: 675
P. sphacelatum (Nees) Th. Dur. & Schinz	FTEA G3: 689
P. squamulatum Fresen.	FTEA G3: 676
P. thunbergii Kunth	FTEA G3: 687
P. trachyphyllum Pilg.	FTEA G3: 682
P. trisetum Leeke	FTEA G3: 682
P. unisetum (Nees) Benth.	FTEA G3: 681
Perotis patens Gand.	FTEA G2: 394
Pogonarthria squarrosa Roem. & Schult.) Pilg.	FTEA G2: 267
Rhynchelytrum nerviglume (Franch.) Chiov.	FTEA G3: 514
R. repens (Willd.) C.E. Hubbard	FTEA G3: 515
R. scabridum (K.Schum.) Chiov.	FTEA G3: 511
R. subglabrum (Mes) Stapf & Hubbard	FTEA G3: 513
Rottboellia cochinchinensis (Lour.) W.D. Clayton	FTEA G3: 853
Saccharum spontaneum L.	FTEA G3: 704
Schmidtia pappophoroides J.A. Schmidt	FTEA G1: 165
Sehimia nervosum (Rottler) Stapf	FTEA G3: 750
Setaria atrata Hack.	FTEA G3: 524
S. megaphylla (Steud.) Th. Dus.& Schinz.	FTEA G3: 539
S. sphacelata (Schumach.) Moss	FTEA G3: 527
Sorghastrum stipoides (Kunth) Nash	FTEA G3: 732
Sporobolus africanus (Poir.) Robyns & Tournay	FTEA G2: 375
S. agrostoides Chiov.	FTEA G2: 378
S. angustifolius A.Rich.	FTEA G2: 377
S. confinis (Steud). Chiov.	FTEA G2: 382
S. fimbriatus (Trin.) Nees	FTEA G2: 377
S. helvolus (Trin.) Th. Dur. & Schinz.	FTEA G2: 371
S. ioclados (Trin.) Nees	FTEA G2: 367
S. spicatus (Vahl) Kunth	FTEA G2: 369
Stenotaphrum dimidiatum (L.) Brongn.	FTEA G3: 549
Themeda triandra Forssk.	FTEA G3: 829
Trachypogon spicatus (L.f.) Kuntze	FTEA G3: 709
Tricholaena teneriffae (L.f.) Link	FTEA G3: 504
Urochloa oligotricha (Fig. & De Not.) Henr.	FTEA G3: 606
U. panicoides P. Beauv.	FTEA G3: 602

II Cultivated Gramineae

Avena sativa L.	FTEA G1: 82
Eleusine coracana (L.) Gaertn.	FTEA G2: 260
Eragrostis tef (Zucc.) Trotter	FTEA G2: 213

Hordeum vulgare L.	FTEA G1: 73
Oryza sativa L.	FTEA G1: 28
Panicum miliaceum L.	FTEA G3: 489
Saccharum officinarum L.	FTEA G3: 70
Secale cereale L.	FTEA G1: 73
Setaria italica (L.) P. Beauv.	FTEA G3: 520
Sorghum vulgare L.	FTEA G3: 726
Triticum vulgare L.	
Zea mays L.	FTEA G3: 857

III Plants including crops, other than Gramineae, recorded as eaten by *Spodoptera exempta* larvae

Compositae

Helianthus sp.
Xanthium sp.

Convolvulaceae

Ipomoea batatas (L.) Lam

Cyperaceae

Cyperus esculentus L.
C. margaritaceus Vahl.
C. rotundus L.

Iridaceae

Tritonia crocosmiflora (Lemoine)

Leguminosae

Arachis hypogaea L.
Phaseolus sp.
Pisum sativum L.

Liliaceae

Xerophyllum sp.

Malvaceae

Gossypium sp.
Sida sp.

Musaceae
Musa sp.

Palmae
Cocos nucifera L.

Rubiaceae
Coffea sp.

Solanaceae
Capsicum sp.
Lycopersicon esculentum Miller
Nicotiana tabacum L.
S. tuberosum L.

Zygophyllaceae
Tribulus terrestris L.

Key references: Clayton (1970)
Clayton *et al.* (1974)

Appendix 3 Natural enemies recorded for *Spodoptera exempta*

Imago	Parasitoids	Nematoda: Gordiidae Mermithidae
	Predators	Arachnida (spiders, scropions): Solifugidae Aves (birds, especially crepuscular species) Amphibia (frogs and toads) Chiroptera (bats)
Ovum (egg)	Parasitoids	Hymenoptera: Scelionidae (*Teleonomus* sp.)
	Predators	Formicidae: Various ant genera Heteroptera: Anthocoridae Phlaeothripidae
Larva (caterpillar)	Parasitoids	Diptera: Bombyliidae *Geron exemptus* Bowden *Villa sexfasciata* (Wiedemann) *V. paniscoides* Bezzi *V. (Exhyalanthrax) abruptus* (Loew) *V. (E.) lugens* (Loew) *V. (E.) viduatus* (Loew) Tachinidae *Blepharella analis* (Curran) *Bracheliopsis geniseta* Emden *Campylocheta risbeci* (Mesnil) *Ceromya* cf. *buccalis* (Curran) *Chetogena* cf. *setosina* (Curran) *Dejeania bombylans* (Fabricius) *Exorista xanthaspis* (Wiedemann) *Goniopthalmus halli* Mesnil *Linnaemya longirostris* (Macquart) *L. alopecina* (Speiser) *Nemoraea rubellana* Villeneuve

Larva *cont.* Parasitoids *cont.*

N. capensis (Robineau-Desvoidy)
Pales sarcophagaeformis (Jaennicke)
Palexorista zonata (Curran)
Peleteria iavana (Wiedemann)
Peribaea orbata (Wiedemann)
P. mitis (Curran)
P. suspecta (Malloch)
Periscepsia decolor (Emden)
Pseudogonia rufifrons (Wiedemann)
Winthemia quadrata (Wiedemann)

Hymenoptera:

Braconidae

Chelonus bifoveolatus Szepligeti
Disophrys luteum (Brulle)
Dolichogenidea aethiopicus (Wilkinson)
Homolobus sp.
Protomicroplitis sp. nr. *Xanthaspis* (Ashmead)

Chalcididae

Brachymeria excarinata Gahan
B. marmonti (Girault)
Invreia sp.

Eulophidae

Euplectrus laphygmae Ferriere
Pediobius bruchicida (Rondani)
Tetrastichus '(miser group)'

Ichneumonidae

Barylypa bipartita (Morley)
Campoletis sp. prob. *pedunculata* (Enderlein)
Charops sp.
Diatora sp.
Itamoplex nigropictus Cameron
Mesochorus sp.
Metopius discolor Tosquinet
Netelia sp. prob. *luteolata* (Tosquinet)

Larva *cont.*	Parasitoids *cont.*	*Parania prima* Gauld *Vernamalon spilopterum* (Morley) Pteromalidae *Mesopolobus* sp. *Pteromalus* sp. *Systasis* sp.
	Pathogens	Bacteria – primary and/or secondary infections Cytoplasmic virus (CPV) Fungus: *Nomuraea rileyi* (Farlow) Sampson Virus: *S. exempta* nucleopolyhedrovirus (SpexNPV) Protozoa: Microsporidia *Nosema necatrix* Kramer
	Predators	Mammalia: Cercopithecidae *Papio anubis* Muridae, especially Soricidae Aves: *Ciconia abdimii* (Abdim's Stork) *Ciconia ciconia* (European (White) Stork) *Corvus albus* (Pied Crow) *Falco naumanni* (Lesser Kestrel) *Leptoptilus crumeniferus* (Marabou Stork) *Milvus migrans* (Black Kite – both races) Other bird species (such as bustards, flycatchers, shrikes, bulbuls and other passerines) have a negligible effect on outbreak populations

Larva *cont.*	Predators *cont.*	Birds, such as ostriches, will eat larvae inadvertently
		Amphibia: Frogs Toads
		Reptilia: Various lizards and skinks
		Insecta: Coleoptera: Carabidae *Calossoma* sp.
		Hymenoptera: *Ammophila* sp. *Sphex* sp. Arachnida Solifugidae
Pupa (chrysalis)	Pathogens	Bacteria – possibly as secondary infections Fungus – as secondary infection, following damage to the pupa
	Predators	Rodentia: Various mice and shrews
		Insecta: Coleoptera: Staphylinidae
	Parasitoids	Diptera: Bombyliidae *Exhyalanthrax abruptus* (Loew) *E. lugens* (Loew) *E. viduatus* (Loew) *Geron exemptus* (Bowdan)

Pupa *cont.* Parasitoids *cont.* *Villa paniscoides* Bezzi
V. sexfasciata Bezzi

Hymenoptera:
Braconidae
Disophrys luteum (Brulle)
Ichneumonidae:
Barylaypa bipartita Morley
Netelia sp. prob. *luteolata* (Tosquinet)
Parania prima Gauld
Vernamalon spilopterum (Morley)

Key reference: Merrett (1986b)

Appendix 4 Conversion tables

(after Amiran and Schick, 1961)

Lengths

1 micron (1 mm)	= 0.001 mm
1 mm	= 0.1 cm
	= 0.039 in
1 cm	= 10 mm
	= 0.01 m
	= 0.394 in
	= 0.033 ft
1 metre (m)	= 100 cm
	= 39.37 in
	= 3.28 ft
1 km	= 100 000 cm
	= 1 000 m
	= 1 093.6 yd
	= 0.62 mi
	= 0.54 nmi
1 inch (in)	= 25.4 mm
	= 2.54 cm
	= 0.025 m
1 foot (ft)	= 12 in
	= 30.48 cm
	= 0.3048 m
1 yard (yd)	= 36 in
	= 3 ft
	= 91.44 cm
	= 0.9144 m
1 mile (mi)	= 5280 ft
	= 1760 yd
	= 1609.344 m
	= 1.609 km
	= 0.870 nmi
1 nautical mile	= 1 852 m
(International)	= 1.85 km
(nmi)	= 6 076 ft
	= 204.5 yd
	= 1.15 mi

Area

International units

1 cm^2	= 100 mm^2
	= 0.000 1 m^2
	= 0.155 in^2
1 m^2	= 10 000 cm^2
	= 1 550 in^2
	= 10.764 ft^2
	= 1.196 yd^2
1 hectare (ha)	= 10 000m^2
	= 0.02 km^2
	= 11960 yd^2
	= 2.471 acre
1 km^2 (kilometre)	= 10 000 000 000 cm^2
	= 1 000 000 m^2
	= 100 ha
	= 247.1 ac
	= 0.386 mi^2
1 in^2	= 6.452 cm^2
1 ft^2	= 144 in^2
	= 929.0 cm^2
	= 0.092 9 m^2
1 yd^2	= 9 ft^2
	= 8 361.3 cm^2
	= 0.836 m^2

Area *cont.*

1 acre	= 43 560 ft^2
	= 4 840 yd^2
	= 4 046.86 m^2
	= 0.405 ha
	= 0.004 km^2
1 mi^2 (mile)	= 640 acre
	= 259.0 ha
	= 2.59 km^2

Eritrea, Ethiopia

1 gasha	= 99 acre
	= 40 ha

Egypt, Sudan

1 feddan	= 1.038 acre
	= 0.42 ha

Weight

International units

1 gramme (g)	= 0.001 kg
	= 0.035 oz
	= 0.002 lb
1 kilogramme (kg)	= 1 000 g
	= 0.001 t
	= 35.27 oz
	= 2.2 lb
1 quintal (q)	= 100 kg
	= 220.5 lb
1 metric tonne (t)	= 10 q
	= 1000 kg
	= 2 204.6 lb
	= 19.7 cwt
	= 0.98 ton (lgt)
1 ounce (oz)	= 28.35 g
1 pound (lb)	= 16 oz
	= 453.6 g
	= 0.45 kg
1 hundredweight (cwt)	= 112 lb
	= 50.8 kg
1 short ton (sht)	= 2000 lb
	= 0.893 lgt
	= 907.18 kg
	= 0.907 t
1 ton (lgt)	= 2240 lb
	= 20 cwt
	= 1 016 kg
	= 1.016 t

Standard (Pakistan)

1 seer	= 2.06 lb
	= 0.93 kg

Standard (Pakistan)

1 maund	= 40 seer
	= 82.28 lb
	= 37.32 kg

Egypt, Sudan

1 rotl	= 0.99 lb
	= 0.45 kg

Egypt, Sudan

1 kantar	= 100 rotl
	= 99 lb
	= 44.9 kg

Velocity

1 m/s	= 3.28 ft/s
	= 3.6 km/h
	= 2.24 m/h
1 m/min	= 0.06 km/h
	= 0.037 mi/h
(mph)	
	= 0.033 kn
1 km/h	= 0.278 m/s
	= 0.91 ft/s
	= 0.62 m/h
	= 16.7 m/min
1 ft/s	= 0.3 m/s
	= 1.1 km/h
	= 0.68 mi/h

Velocity *cont.*

1 mi/h	= 1.47 ft/s
	= 0.45 m/s
	= 26.8 m/min
	= 1.61 k/h
	= 0.87 kn
1 knot (kn)	= 0.514 m/s
(International)	= 30.8 m/min
	= 1.852 km/h
	= 1.688 ft/s
	= 1.151 mi/h
	= 1.0. nmi/h

Volume

1 litre (l)	= 1000 ml
	= 1.76 pint
	= 0.22 gal (UK or Imperial)
	= 0.264 gal (US)
1 pint	= 0.57 l
1 gallon (gal) (UK)	= 8 pint
	= 4.55 l
	= 1.2 gal (US)
1 gal (US)	= 3.785 l
	= 0.83 gal (UK)

Fluids

1 ml/ha	= 0.136 fl oz/acre
1 l/ha	= 0.10 gal (US)/acre
	= 0.083 gal (UK)/acre
1 fluid oz/acre	= 73 ml/ha
1 gal (US)/acre	= 9.35 l/ha

Solids

1 g/ha	= 0.014 oz/acre
1 kg/ha	= 14.27 oz/acre
	= 0.89 lbs/acre
1 oz/acre	= 70.1 g/ha
	= 0.07 kg/ha
1 lb/acre	= 1.121 kg/ha

Beaufort wind scale (after McIntosh, 1972)

Force	Description	Specification for use on land	Speed m/s
0	Calm	Calm, smoke rises vertically	0.0–0.2
1	Light air	Direction of wind shown by smoke drift but not by wind vanes	0.3–1.5
2	Light breeze	Wind felt on face, leaves rustle, ordinary vane moved by wind	1.6–3.3
3	Gentle breeze	Leaves and small twigs in constant motion, wind extends light flag	3.4–5.4
4	Moderate breeze	Raises dust and loose paper, small branches moved	5.5–7.9
5	Fresh breeze	Small trees in leaf begin to sway, crested wavelets form on inland waters	8.0–10.7
6	Strong breeze	Large branches in motion, whistling heard in telegraph wires, umbrellas used with difficulty	10.8–13.8
7	Near gale	Whole trees in motion, inconvenience felt when walking against wind	13.9–17.1
8	Gale	Breaks twigs off trees, generally impedes progress	17.2–20.7
9	Strong gale	Slight stuctural damage occurs (chimney pots and roofing material removed)	20.8–24.4
10	Storm	Seldom experienced inland; trees uprooted, considerable structural damage occurs	24.5–28.4
11	Violent storm	Very rarely experienced; widespread damage	28.5–32.6

Appendix 5 Useful addresses

I DLCO-EA: headquarters and bases in member countries

Desert Locust Control Organization for Eastern Africa (DLCO-EA)
P O Box 4255
ADDIS ABABA, **Ethiopia**
e-mail: dlc@telecom.net.et

DLCO-EA
P O Box 1987
DJIBOUTI, **Djibouti**

DLCO-EA
P O Box 231
ASMARA, **Eritrea**

DLCO-EA
P O Box 30023
NAIROBI, **Kenya**

DLCO-EA
P O Box 36
HARGEISA, **Somalia**

DLCO-EA
P O Box 412
MOGADISCIO, **Somalia**

DLCO-EA
P O Box 328
KHARTOUM NORTH, **Sudan**

DLCO-EA
P O Box 593
ARUSHA, **Tanzania**

DLCO-EA
P O Box 9134
KAMPALA, **Uganda**

II Other regional organizations

Food and Agricultural Organization
of the United Nations (FAO)
Regional Office for Africa (RAFR)
P O Box 1628
ACCRA, **Ghana**

International Institute of
Tropical Agriculture (IITA)
PMB 5320
IBADAN, **Nigeria**

International Red Locust Control
Organisation for Central and
Southern Africa
(IRLCO-CSA)
P O Box 240252
NDOLA, **Zambia**
email: locust@zamnet.zm

Organisation Commune de Lutte
Antiacridienne et de Lutte
Antiaviare
(OCLA-LAV)
BP 1066
DAKAR, **Senegal**

III Research organizations

Faculty of Agricultural Sciences
State University of Gent
Coupure Links 653
B-9000 GENT, **Belgium**

Institut National pour Recherches
Agronomiques (INRA)
Station de Zoologie et d'Apidologie
84140 MONTFAVET, **France**

Ruhr-Universität Bochum
Facultät für Biologie
Universitätstrasse 150
4630 B'OCHUM, **Germany**

Wageningen Agricultural University
Postbus 8031
6700 EH WAGENINGEN
The Netherlands

International Centre for Insect
Physiology and Ecology (ICIPE)
P O Box 30772
NAIROBI, **Kenya**

Kenya Agricultural Research
Institute (KARI)
P O Box 30148
NAIROBI, **Kenya**

Kenya Meteorological Research
Institute
P O Box 30259
NAIROBI, **Kenya**

University of Nairobi
P O Box 30197
NAIROBI, **Kenya**

Sokoine University of Agriculture
P O Box 3000
MOROGORO, **Tanzania**

University of Dar es Salaam
P O Box 35091
DAR ES SALAAM, **Tanzania**

Imperial College at Silwood Park
ASCOT, Berkshire
SL5 7PY, **UK**

Department of Geography
University of Reading
Whiteknights, READING
RG6 2AB, **UK**

Natural Resources Insititute (NRI)
University of Greenwich,
Central Avenue
Chatham Maritime,
CHATHAM, Kent
ME4 4TB, **UK**
(web page: http://www.nri.org)

School of Biological Sciences
University College of North Wales
(UCNW)
BANGOR, Gwynedd
LL57 2UW, **UK**

University of Bradford
BRADFORD, West Yorkshire
BD7 1DP, **UK**

IV Equipment suppliers

Pheromones and Uni-traps

International Pheromone Systems
Units 12 and 13 Meadow Lane
Meadow Lane Industrial Estate
Ellesmere Port
South Wirral
L65 4EH, **UK**
(tel: 0151 3572655)

Agrisense-BCS
Treforest Industrial Estate
Treforest
Pontypridd
Mid-Glamorgan
CF37 5SU, **UK**
(tel: 01443 841155,
web:
http://www.agrisense.demon.co.uk)

Sprayers

Micron ULVA hand-sprayers, also vehicle- and aircraft-mounted sprayers

Micron Sprayers Ltd
Three Mills
Bromyard
Herefordshire
HR7 4HU, **UK**
(tel: +44 (0) 1885 482397,
e-mail: micron@micron.co.uk)

Hand sprayers, also vehicle- and aircraft-mounted sprayers

Micronair Ltd (now part of Micron Sprayers Ltd)
Bembridge Fort
Sandown
Isle of Wight
PO36 8QS, **UK**

Abbreviations

ARCZ	African Rift Convergence Zone
BHT	butylated hydroxytoluene
CAB	Congo Air Boundary
CCD	cold cloud duration
CPV	cytoplasmic virus
DAO	District Agricultural Officer
DFID	Department for International Development
DGPS	Differential Global Positioning System
DLCO-EA	Desert Locust Control Organization for Eastern Africa
EAAFRO	East African Agriculture and Forestry Research Organization
ENS	exhaust nozzle sprayer
ENSO	El Niño Southern Oscillation
FAO	Food and Agriculture Organization of the United Nations
ICIPE	International Centre for Insect Physiology and Ecology
IGRs	insect growth regulators
IPM	integrated pest management
IRLCO-CSA	International Red Locust Control Organisation for Central and Southern Africa
ITCZ	Inter-Tropical Convergence Zone
KARI	Kenya Agricultural Research Institute
NAC	National Armyworm Co-ordinator
NARC	National Agricultural Research Centre
NARL	National Agricultural Research Laboratories
NPPS	National Plant Protection Services
NRI	Natural Resources Institute
OCLA-LAV	Organisation Commune de Lutte Antiacridienne et de Lutte Antiaviare
PCS	Pest Control Services, Arusha, Tanzania
PDUS	Primary Data User System
SADC	Southern African Development Community
SpexNPV	*S. exempta* nucleopolyhedrovirus
TARO	Tanzania Agricultural Research Organization
ULV	ultra-low-volume
USAID	United States Agency for International Development
VAR	volume application rates
VMD	volume median diameter

Plate 1 *Spodoptera exempta* moths: male above, female below.

Plate 2 Male (left) and female (right) moths of other African species in the genus *Spodoptera*.

Plate 3a Entire batch of *Spodoptera exempta* eggs.

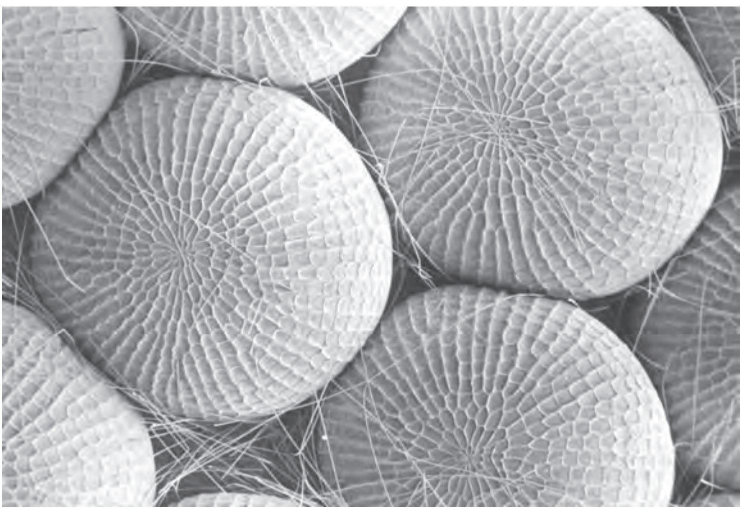

Plate 3b Individual *Spodoptera exempta* eggs.

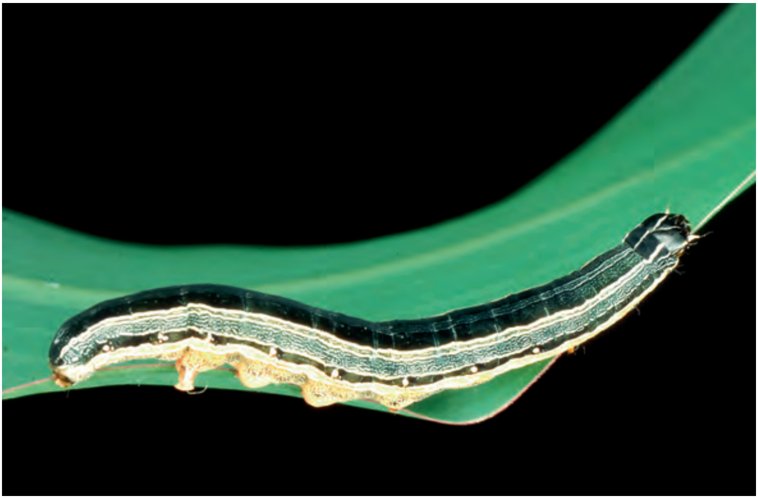

Plate 4a Gregarious phase larvae of *Spodoptera exempta* – lateral view.

Plate 4b Gregarious phase larvae of *Spodoptera exempta* – dorsal view.

Plate 5 Gregarious phase larva of *Spodoptera exigua* for comparison with Plate 4.

Plate 6 A sawfly larva (Hymenoptera) for comparison with Plate 4.

Plate 7 Solitary *Spodoptera exempta* larva.

Plate 8 Gregarious phase *Spodoptera exempta* larvae marching.

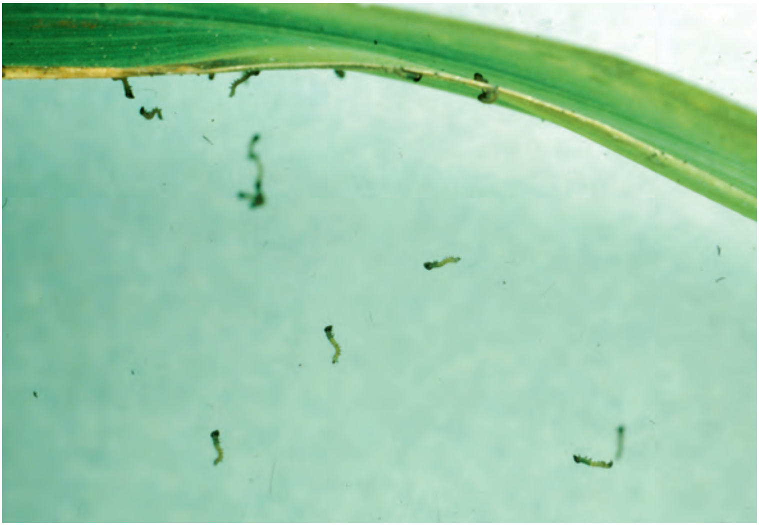

Plate 9 *Spodoptera exempta* first instar larvae suspended on silken threads.

Plate 10 'Windowing' effect on host plant caused by the feeding of young *Spodoptera exempta* larvae.

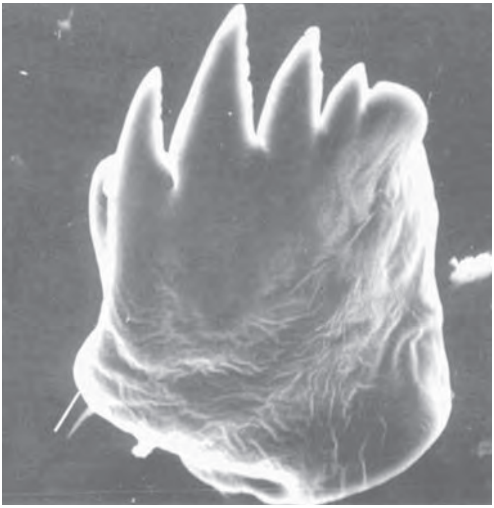

Plate 11a Mandibles of *Spodoptera exempta* larvae at first insta r.

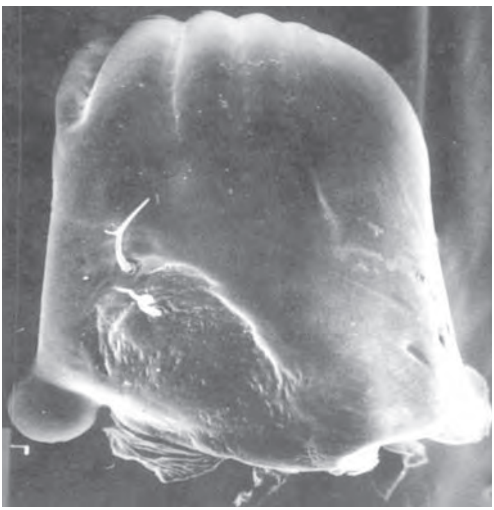

Plate 11b Mandibles of *Spodoptera exempta* larvae at sixth insta r.

Plate 12 Last instar larva of *Spodoptera exempta* burrowing into the ground to pupate.

Plate 13 Small mounds of earth pushed up from where *Spodoptera exempta* larvae had burrowed into the ground to pupate.

Plate 14 Pre-pupae and pupae of *Spodoptera exempta*.

Plate 15 Newly emerged *Spodoptera exempta* moths before and after expanding their wings.

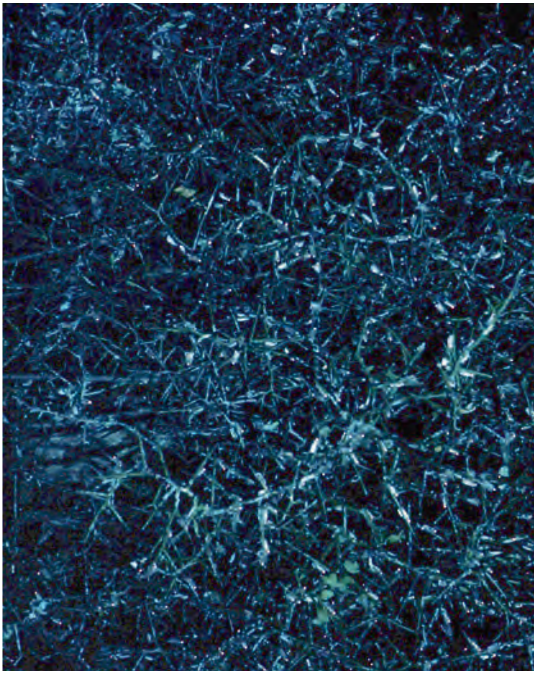

Plate 16 *Spodoptera exempta* moths congregated in trees at night.

Plate 17 Male *Spodoptera exempta* moth hiding under a cow pat (day shelter).

Plate 18 *Spodoptera exempta* moths hiding under a stone (day shelter).

Plate 19 Female *Spodoptera exempta* moth ovipositing.

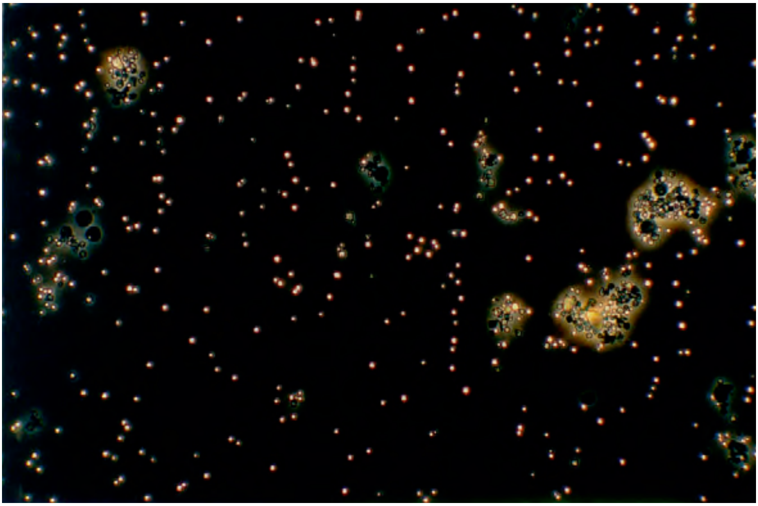

Plate 20a A suspension of nucleopolyhedrovirus (SpexNPV), x400 under a light microscope, showing the bright refractile infectious particles diagnostic of infection.

Plate 20b Larva of *Spodoptera exempta* killed by nucleopolyhedrovirus (SpexNPV).

Plate 21 Larva of *Spodoptera exempta* killed by the fungus *Nomuraea rileyi*.

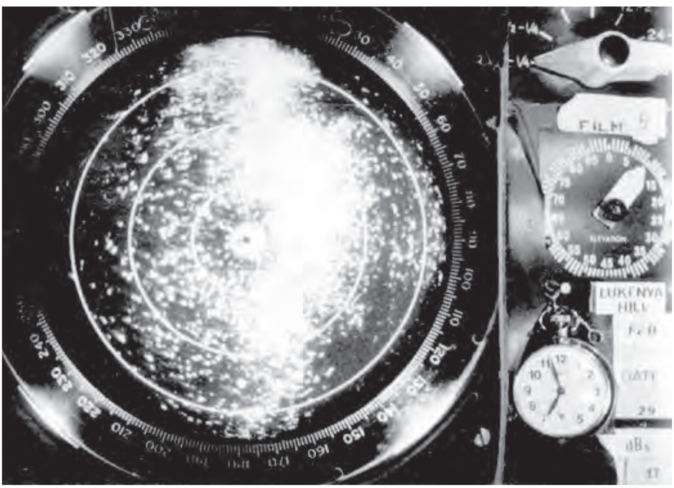

Plate 22 Radar image of armyworm moth concentration at a storm outflow gust front.

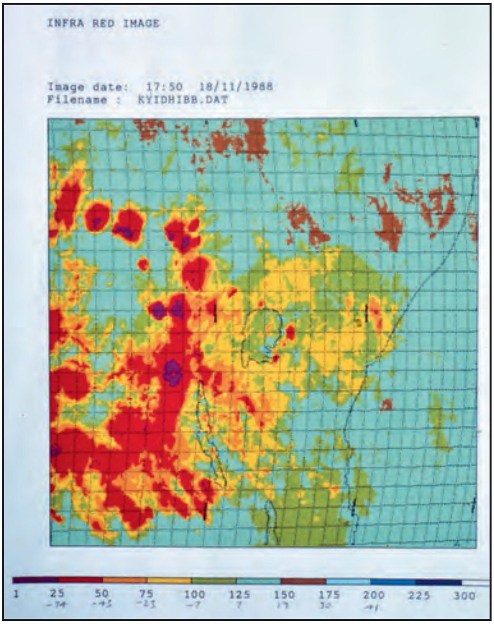

Plate 23 Meteosat image in infra-red showing cloud build up on the Inter-tropical Convergence Zone (ITCZ) over Tanzania and Zaire.

Plate 24 Model relief map showing topography in primary outbreak areas of Kenya.

Plate 25 Model relief map showing topography in primary outbreak areas of Tanzania.

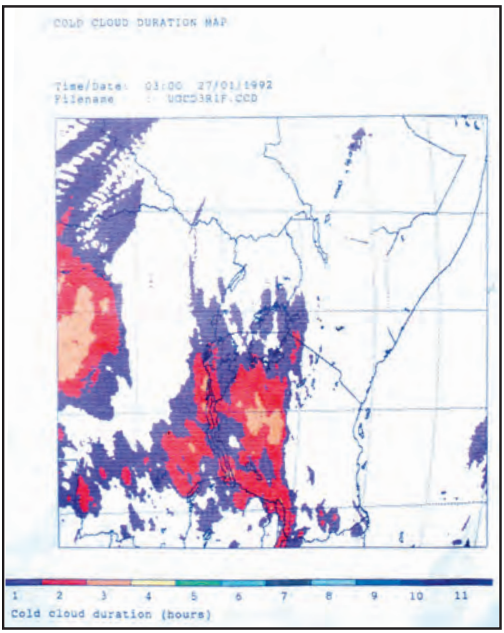

Plate 26 Example of a cold cloud duration map (CCD) of eastern Africa for one night, showing storm clouds with temperatures below -50 °C, and their duration.





The African Armyworm Handbook

The African armyworm, *Spodoptera exempta* (Walker) (Lepidoptera: Noctuidae) is a serious migratory pest of cereal crops and grasslands recognized as such since early in the seventeenth century. It can have a major impact on the livelihoods of farmers in many countries of sub-Saharan Africa, as well as Yemen, some Pacific islands and parts of Australia.

The African Armyworm Handbook includes as much up to date information as possible on the biology, ecology, epidemiology and management of this pest as well as containing an extensive list of useful references. The handbook will be of interest to those working in the agricultural sector, particularly those responsible for the control of migrant pests.

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