Tasmanian
Pasture and Forage Pests
Identification, biology and control

Peter McQuillan, University of Tasmania
John Ireson, Tasmanian Institute of Agricultural Research (TIAR)
Lionel Hill, Department of Primary Industries and Water (DPIW)
Catherine Young, Department of Primary Industries and Water (DPIW)
Authors
Peter McQuillan, University of Tasmania
John Ireson, Tasmanian Institute of Agricultural Research (TIAR)
Lionel Hill, Department of Primary Industries and Water (DPIW)
Catherine Young, Department of Primary Industries and Water (DPIW)

This book is an update of Tasmanian Pasture Pests by McQuillan and Ireson that was published by the Department of Agriculture, Tasmania (Hobart) in 1987.

Technical expertise
This book is based on the latest knowledge and experience of many DPIW and TIAR officers. This knowledge has been used and incorporated into the text, creating a publication focused specifically on pastures and forage pests in the Tasmanian environment.

Acknowledgments
Thank you to all the people who have generously contributed their time and provided valuable feedback on this publication. Special thanks to Stuart Smith (DPIW) for his contribution to the section 'Pest control when renewing damaged pasture'.
Layout by Jenny Baulis and Tracey Taylor (DPIW)
Photo enhancement by Graeme Harrington (DPIW)

Financial assistance
We would like to thank Australian Wool Innovation via the 8x5 Wool Profit Program for their generous contribution.

Disclaimer
The information in this book is collated from project experience, expert advice and reference sources and is offered in good faith as a useful guide to pasture and forage pests in Tasmania.

Copyright
© State of Tasmania 2007
Photos courtesy of the State of Tasmania unless otherwise credited.

Copies
For copies of this book contact:
Department of Primary Industries and Water
PO Box 46
Kings Meadows Tas 7249
Mt Pleasant Laboratories
165 Westbury Road, Prospect
Phone: 1300 368 550
Email: ExtensiveAgEnquiries@dpiw.tas.gov.au

ISBN
ISBN: 0–7246–6761 X
Table of Contents

Introduction 1

QUICK REFERENCE GUIDES
Diagnosis of pasture damage by symptoms 4
Diagnosis of forage damage by symptoms 5
Main periods of pest damage 6
Summary of insecticidal options for major pasture pests 7
Insect development stages—January to June 8
Insect development stages—July to December 9
Pest control when renewing damaged pasture 10
Methods of insect control 12

INDIVIDUAL PESTS
Moths - caterpillars, corbies, cutworms and armyworms
Corbie, winter corbie 14
Oxycanus grass grub 20
Diamondback moth 22
Cabbage white butterfly 26
Armyworms 29
True cutworms 34
Chevron cutworm 38
Green cutworms 40
Grass anthelid caterpillar 42

Beetles—cockchafers and weevils
Blackheaded pasture cockchafers 44
Redheaded pasture cockchafer 48
Yellowheaded pasture cockchafers 52
Argentine stem weevil 54
Sitona weevil 56
Whitefringed weevil 57
Grasshoppers and crickets
Wingless grasshopper 59
Black field cricket 63

Fleas, mites and aphids
Lucerne flea 66
Earth mites 71
Aphids 75

Snails and slugs
Small pointed snail 76
Slugs 77

Biological diversity & pasture pest control 80
Dung Beetles 84
Earthworms 85

Glossary 86

Names of pest and beneficial species 88
Introduction

This book provides fundamental biological information about most pasture and forage pests in Tasmania. It can be used directly by farmers or by those who supply advice or training to farmers. It also provides some suggestions for control, pasture selection and pasture management, but does not detail pesticide treatments.

A productive pasture is a valuable capital asset and an essential foundation for a profitable grazing enterprise. Indeed sown pastures are probably the most valuable renewable primary resource in Tasmania and should be well cared for.

Pastoral agriculture developed rapidly after the arrival of Europeans and sown pasture now accounts for approximately 860 000 of the 1.34 million hectares of pasture in the State. Its evolution has been characterised by the introduction, from temperate regions of the world, of high–yielding pasture plants (especially perennial ryegrass), into land cleared from the indigenous vegetation, the routine application of phosphatic fertiliser, the use of clovers as fixers of nitrogen and efficient grazing techniques. However, sown pastures in Tasmania still do not contain the diversity of plants found in the native pastures of Europe and Central Asia which have evolved over centuries of grazing and pest pressure.

A range of native insects have adapted to this new ecosystem. Some of these insects have been able to feed in sown pasture and sustain populations on such a vast scale that farmer intervention is necessary to limit economic losses. Indeed, the biomass of insects feeding in a pasture sometimes exceeds that of the livestock.

Forage crops are also attacked by a mixture of native and introduced insects. Some pests are shared with related cash crops (canola, broccoli, cauliflower and cereals).

Potential pests are present in all pastures at all times but in most years they remain in low numbers. They are held at these low densities by factors such as competition with other insects, predators, diseases and weather conditions. Under such circumstances insect feeding has little effect on pasture productivity. Low populations benefit pasture by aerating soil, enhancing organic cycles and stimulating new growth. They also maintain viable populations of the pests’ natural enemies (predators, parasites and diseases) which help suppress them most of the time.

However problems arise when the insect population reaches a level at which the feeding rate exceeds the capacity of the pasture plants to compensate and the resulting monetary loss of production exceeds the cost of treatment. At this stage control measures may be necessary.
For effective control of any pests three factors are important:

- correct identification of the pest causing the problem
- an assessment of the population trend of the pest (Is it present in sufficient numbers to warrant treatment? Is it unrestrained by predators, parasites, diseases and weather?)
- correct timing and application of any treatment.

This book provides a convenient means of identifying pests of pasture and forage crops in Tasmania, provides basic information on their distribution and life cycle and outlines appropriate control measures.

Current details of pesticides are available from resellers, commercial agronomists or by purchase of commercial databases such as Infopest® (Queensland Department of Primary Industries, phone 07 3239 3967, email infopest@dpi.qld.gov.au, GPO Box 46, Brisbane 4001). The official database of the Australian Pesticides and Veterinary Medicines Authority is called PUBCRIS and can be freely accessed at http://www.apvma.gov.au.

The official Australian list of scientific and common names of common or pest insects, mites and snails can be found at the CSIRO web site http://www.ento.csiro.au/aicn/.

**How to get the most from this book**

The book begins with tables summarising symptoms of damage, times of damage, insecticidal options and pest life stages. This is followed by two sections on methods of pest control.

The major section of the book deals with pests individually and they are arranged in the following broad groupings: caterpillars of moths, grubs of beetles, grasshoppers and crickets, microscopic pests and slugs and snails.

This book concludes with sections on beneficial insects and glossaries of scientific terms and names.

**Biosecurity**

Commercial agronomists provide daily advice to farmers on the management of pasture pests. Entomologists of the Department of Primary Industries and Water support that service by identifying, when necessary, insects and allied animals on a fee for service basis. However, the fee is waived if the identification request arises from a concern that a pest new to Tasmania has been detected. Phone 03 6233 3352 (Quarantine Tasmania, Hobart) if you are concerned that you have encountered a new pest.
Quick Reference Guides
## Diagnosis of pasture damage by symptoms

<table>
<thead>
<tr>
<th>Symptom</th>
<th>Time of year</th>
<th>Possible cause</th>
</tr>
</thead>
<tbody>
<tr>
<td>General thinning of sward often leading to bare patches which become weed dominant; holes present in soil under loose capping of silk-bound soil and debris.</td>
<td>May–September</td>
<td>Winter corbie (esp. NW Tas.)</td>
</tr>
<tr>
<td></td>
<td>August–November</td>
<td>Corbie</td>
</tr>
<tr>
<td>General thinning, especially of clover, often leading to bare patches; numerous low heaps of soil thrown up on surface.</td>
<td>April–October but especially May–June</td>
<td>Blackheaded pasture cockchafers</td>
</tr>
<tr>
<td>General unthriftiness of pasture, sometimes with sward uprooted by birds and stock.</td>
<td>April–October but especially April–June</td>
<td>Redheaded pasture cockchafer and other root-feeding cockchafer.</td>
</tr>
<tr>
<td>Clover leaves showing speckled appearance of green tissue removed from both surfaces leaving window–like holes (leaf skeletonising).</td>
<td>April–June September–November (Summer if irrigated pasture).</td>
<td>Lucerne flea (esp. NW Tas.)</td>
</tr>
<tr>
<td>Clover leaves showing a blotchy silvery discolouration.</td>
<td>April–June September–November</td>
<td>Redlegged earth mite (esp. light soils in coastal areas)</td>
</tr>
<tr>
<td>Leaf chewing of clover and grasses; stems of grasses severed below seed head; numerous caterpillars in sward may cause fouling.</td>
<td>December–January (occasionally September–February)</td>
<td>Southern armyworm</td>
</tr>
<tr>
<td>Selective loss of clovers and flatweeds, often followed by a loss of grass in autumn; grasshoppers abundant mainly on light soils.</td>
<td>December–April</td>
<td>Wingless grasshopper</td>
</tr>
<tr>
<td>Poor germination of annual–type pastures or new pastures in autumn; loss of pasture adjacent to cracks in heavier soils.</td>
<td>March–May</td>
<td>Black field cricket</td>
</tr>
</tbody>
</table>
## Diagnosis of forage damage by symptoms

<table>
<thead>
<tr>
<th>Symptom</th>
<th>Time of year</th>
<th>Possible cause</th>
</tr>
</thead>
<tbody>
<tr>
<td>Seedlings lopped or foliage chewed, dark grey caterpillars in soil</td>
<td>Spring</td>
<td>Brown &amp; common cutworms</td>
</tr>
<tr>
<td>adjacent to seedlings</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Seedlings not establishing, slime trails present on soil</td>
<td>Spring</td>
<td>slugs</td>
</tr>
<tr>
<td>Swede foliage and tubers chewed; grey caterpillars with black chevron</td>
<td>Winter</td>
<td>Chevron cutworm</td>
</tr>
<tr>
<td>markings along back present on foliage</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Swede foliage chewed; green or brown caterpillars with pair of</td>
<td>Winter</td>
<td>Green cutworm</td>
</tr>
<tr>
<td>white spots on rear present</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Brassica foliage chewed; small wrinkly green to brown</td>
<td>Spring</td>
<td>Diamondback moth</td>
</tr>
<tr>
<td>caterpillars present</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Maize silks chewed; tip kernels chewed, husks with holes, green or</td>
<td>Summer</td>
<td>Native budworm &amp; corn earworm</td>
</tr>
<tr>
<td>brown to orange caterpillars present</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Brassica foliage chewed; large docile green caterpillars present</td>
<td>Autumn</td>
<td>Cabbage white butterfly</td>
</tr>
<tr>
<td>Maize foliage chewed; silks chewed, caterpillar frass present</td>
<td>Summer</td>
<td>Common &amp; southern armyworms</td>
</tr>
<tr>
<td>Lucerne unthrifty; roots absent or chewed; fat, white, legless, grubs</td>
<td>Any season</td>
<td>Whitefringed weevil grubs</td>
</tr>
<tr>
<td>in soil.</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lucerne and medics unthrifty (nitrogen starved); root nodules damaged,</td>
<td>Any season</td>
<td>Sitona weevil grubs</td>
</tr>
<tr>
<td>containing or bearing white, legless grubs</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lucerne and medic foliage scalloped or defoliated</td>
<td>Spring and autumn</td>
<td>Sitona weevil adults</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>
## Main periods of pest damage

<table>
<thead>
<tr>
<th>Pest</th>
<th>January</th>
<th>February</th>
<th>March</th>
<th>April</th>
<th>May</th>
<th>June</th>
<th>July</th>
<th>August</th>
<th>September</th>
<th>October</th>
<th>November</th>
<th>December</th>
</tr>
</thead>
<tbody>
<tr>
<td>Corbie</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Winter corbie</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Oxycanus grass grub</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Diamondback moth</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Cabbage white butterfly</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Southern armyworm</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>True cutworms</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Chevron cutworm</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Green cutworms</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Grass anthelid caterpillar</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Blackheaded cockchafers</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Redheaded cockchafer</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Yellowheaded cockchafer</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Wingless grasshopper</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Black field cricket</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lucerne flea</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Earth mites</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Small pointed snail</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Slugs</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

Forage crops

Pasture
## Summary of insecticidal options for major pasture pests

<table>
<thead>
<tr>
<th>Month</th>
<th>redheaded cockchafer</th>
<th>black–headed cockchafer</th>
<th>winter corbie</th>
<th>corbie</th>
<th>earth mites</th>
<th>lucerne flea</th>
</tr>
</thead>
<tbody>
<tr>
<td>January</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>February</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>March</td>
<td>Inspect roots</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>April</td>
<td>Spell grass if roots damaged, drill Chafer Guard*</td>
<td>First chance to spray</td>
<td>First chance to spray</td>
<td></td>
<td>Spray</td>
<td>Spray</td>
</tr>
<tr>
<td>May</td>
<td>Spell grass or block graze to trample</td>
<td>Optimum time to spray</td>
<td>Optimum time to spray</td>
<td></td>
<td></td>
<td>Spray</td>
</tr>
<tr>
<td>June</td>
<td>Spell</td>
<td>Last chance to spray</td>
<td>Spray when mild</td>
<td>Spray when mild</td>
<td></td>
<td></td>
</tr>
<tr>
<td>July</td>
<td>Spell</td>
<td></td>
<td>Spray when mild</td>
<td>Spray when mild</td>
<td></td>
<td></td>
</tr>
<tr>
<td>August</td>
<td>Spell grass if roots damaged, drill Chafer Guard*</td>
<td>Last chance to spray</td>
<td>Spray when mild</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>September</td>
<td></td>
<td></td>
<td>Spray when mild</td>
<td></td>
<td></td>
<td>Spray</td>
</tr>
<tr>
<td>October</td>
<td></td>
<td></td>
<td>Last chance to spray</td>
<td>Spray, middle of month option</td>
<td></td>
<td>Spray</td>
</tr>
<tr>
<td>November</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>December</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

* Chafer Guard™ is a granular insecticide containing a fungus that is toxic to redheaded cockchafer grubs. It is best used when resowing pasture.
## Insect development stages
### January–June

<table>
<thead>
<tr>
<th>Insect Type</th>
<th>January</th>
<th>February</th>
<th>March</th>
<th>April</th>
<th>May</th>
<th>June</th>
</tr>
</thead>
<tbody>
<tr>
<td>Corbie</td>
<td>pupa</td>
<td>moth</td>
<td>egg</td>
<td>egg</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Winter corbie</td>
<td>egg</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Blackheaded pasture cockchafer</td>
<td>beetle</td>
<td>egg</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Redheaded pasture cockchafer</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>two year cycle</td>
<td></td>
</tr>
<tr>
<td>Yellowheaded pasture cockchafer</td>
<td></td>
<td></td>
<td></td>
<td>several species with one or two year cycles</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Lucerne flea</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>juvenile</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Redlegged earth mite</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Southern armyworm*</td>
<td>larva</td>
<td>pupa</td>
<td>adult</td>
<td>egg</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Wingless grasshopper</td>
<td>mixed</td>
<td>adult</td>
<td>adult</td>
<td>adult</td>
<td>egg</td>
<td>egg</td>
</tr>
<tr>
<td>Black field cricket</td>
<td>mixed</td>
<td>adult</td>
<td>adult</td>
<td>adult</td>
<td>egg</td>
<td>egg</td>
</tr>
<tr>
<td>Oxycanus grass grub</td>
<td>pupa</td>
<td>pupa</td>
<td>adult</td>
<td>adult</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Grass anthelid caterpillar</td>
<td>pupa</td>
<td>pupa</td>
<td>adult</td>
<td>adult</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>True cutworms</td>
<td>larva</td>
<td>pupa</td>
<td>adult</td>
<td>adult</td>
<td>absent</td>
<td>absent</td>
</tr>
<tr>
<td>Chevron cutworm</td>
<td>adult</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Green cutworm</td>
<td>mixed</td>
<td>mixed</td>
<td>adult</td>
<td>adult</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Diamondback moth</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Cabbage white butterfly</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>pupa</td>
<td>pupa</td>
<td>pupa</td>
</tr>
</tbody>
</table>

* Southern armyworm—local populations of these species are usually insignificant in winter. Moths immigratory from the mainland in spring usually initiate the major infestations.
# Insect development stages

## July–December

<table>
<thead>
<tr>
<th></th>
<th>July</th>
<th>August</th>
<th>September</th>
<th>October</th>
<th>November</th>
<th>December</th>
</tr>
</thead>
<tbody>
<tr>
<td>Corbie</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>prepupa</td>
<td>pupa</td>
</tr>
<tr>
<td>Winter corbie</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>prepupa</td>
<td>pupa</td>
<td>adult</td>
</tr>
<tr>
<td>Blackheaded pasture cockchafers</td>
<td>larva</td>
<td>larva</td>
<td>prepupa</td>
<td>prepupa</td>
<td>pupa</td>
<td>pupa</td>
</tr>
<tr>
<td>Redheaded pasture cockchafer</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>two year cycle</td>
</tr>
<tr>
<td>Yellowheaded pasture cockchafers</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>several species with one or two year cycles</td>
</tr>
<tr>
<td>Lucerne flea</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>adult</td>
<td>egg</td>
</tr>
<tr>
<td>Redlegged earth mite</td>
<td>mixed</td>
<td>mixed</td>
<td>mixed</td>
<td>adult</td>
<td>mixed</td>
<td>egg</td>
</tr>
<tr>
<td>Southern armyworm</td>
<td>larva</td>
<td>larva</td>
<td>mixed</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Wingless grasshopper</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>juvenile</td>
<td>juvenile</td>
</tr>
<tr>
<td>Black field cricket</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>egg</td>
<td>juvenile</td>
<td>juvenile</td>
</tr>
<tr>
<td>Oxycanus grass grub</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>two year cycle</td>
</tr>
<tr>
<td>Grass anthelid caterpillar</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>True cutworms</td>
<td>absent</td>
<td>absent</td>
<td>adult</td>
<td>mixed</td>
<td>larva</td>
<td>larva</td>
</tr>
<tr>
<td>Chevron cutworm</td>
<td>larva</td>
<td>mixed</td>
<td>mixed</td>
<td>adult</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Green cutworm</td>
<td>larva</td>
<td>larva</td>
<td>larva</td>
<td>adult</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Diamondback moth</td>
<td>larva</td>
<td>larva</td>
<td>adult</td>
<td>adult</td>
<td>mixed</td>
<td>mixed</td>
</tr>
<tr>
<td>Cabbage white butterfly</td>
<td>pupa</td>
<td>pupa</td>
<td>adult</td>
<td>adult</td>
<td>mixed</td>
<td>mixed</td>
</tr>
</tbody>
</table>

Note: Larva can mean either the grub stage of a beetle or the caterpillar stage of a moth (refer to the Glossary for a more detailed explanation).
Pest control when renewing damaged pasture

The department’s ‘Species for Profit, a Guide for Tasmanian Pastures and Field Crops’ is a useful book that also contains some notes on susceptibility of pasture plants to pests. In general, resistance to pests differs much less between cultivars within species (for example Australian phalaris versus Uneta phalaris) than between species (for example any phalaris versus perennial ryegrass).

Resowing pastures

Resowing pastures is an expensive undertaking, and prior to resowing, every attempt should be made to minimise damaging insect pest effects particularly during the year of establishment.

A well managed perennial pasture should remain productive for 20 years but continued insect attack can reduce the period of high yields to 4–5 years.

Resowing with conventional cultivation and seedbed preparation

Cultivation and planting a break crop prior to re-establishing pasture will reduce organic matter debris from the old pasture and reduce the presence of perennial weed species. As well, it helps to disrupt the life cycle of many insect pests.

Generally the soil will be cultivated and prepared in spring so that soil moisture is conserved in the fallow over summer. If this is done early in the spring many grubs of cockchafers and caterpillars of corbies will still be high in the soil profile and therefore killed. Annual grasses and other weeds will not have had a chance to set seed. A fallow paddock should be unattractive for egg-laying by corbies and cockchafers in the summer. Therefore, pasture sown in the autumn should be assured of a pest-free start. Check that cutworms have not established in weedy fallow.

Conventional seedbeds may also be prepared in late summer/autumn but these often do not disrupt insect pests as effectively as spring prepared seedbeds. This is particularly the case with root feeding cockchafers which will be deep in the soil and which will later feed on the new pasture plants during their early growth stages.

Attention needs to be paid to good seedbed preparation, timing, mixtures and sowing procedure to ensure that a good, vigorous pasture results. A dense, vigorous pasture based on perennial species is, on its own, the best defence against insect attack. A poorly established pasture will usually be more prone to damage later.

The major insect hazard to newly established pasture in the summer after autumn sowing is armyworm.

Sowing into an existing pasture (pasture renovation)

This usually involves ‘spraying–off’ which reduces the vigour of remaining pasture plants and helps to control weedy species with herbicide preferably after three months of hard grazing. Grazing also helps to reduce the vigour of all remaining plants through reducing root system vigour. Sowing is most commonly performed in autumn to ensure good establishment. Various procedures are used in this process and none involve full cultivation and seedbed preparation.
There are two options: direct drilling, or minimum cultivation (minimum tillage).

**Direct Drilling**

Direct drilling does not involve any soil disturbance at all, other than that caused by the seed drill at the time of sowing. The degree of soil disturbance depends on the type of seed drill. This ranges from practically zero disturbance, in the case of a disc drill with press wheels, to full surface soil disturbance with tyned drills. For the latter, disturbance would only be 20–30 mm deep. There is no disturbance of deeper soil layers with direct drilling as there is with conventional cultivation.

Direct drilling is vulnerable to failure due to insect pests. Before suppressing or killing an old pasture with herbicide, pest control is necessary if corbies, winter corbies, pasture cockchafers or cutworms are present. It is tempting to mix the herbicide and insecticide together (assuming they are chemically compatible), to minimise application costs. However, failures have been reported under such circumstances. It is possible that the herbicide treated herbage is unpalatable to the insects, which respond by not feeding until the new seeds germinate. Consequently it is wise to spray the insecticide a week or so before using herbicide to give the pests a chance to feed on the insecticide treated herbage.

Direct drilling in autumn, especially in March or early April, can be risky if pests are present at the site. At this time of year it is difficult to assess population levels of corbies, winter corbies and cockchafers. Insect feeding during the establishment phase of a pasture may seriously compromise the vigour and density of the resulting sward. If direct drilling in early autumn is planned, vigilance is needed from late autumn to early spring to ensure detection of damaging levels of cockchafers or corbie/winter corbie which will require immediate treatment if present.

**Minimum cultivation (minimum tillage)**

Minimum cultivation or minimum tillage involves one or more shallow cultivations to level a rough soil surface and/or to create a shallow seedbed. The depth of cultivation is rarely greater than 75 mm. It is done over a short period of time compared to conventional seedbed preparation. It does not greatly help in controlling insect pests that are living in the deeper soil layers. Therefore, it is important to determine whether any insect pests are present and to control these before undertaking other operations such as herbicide applications and the commencement of shallow cultivation operations. Achieving good pasture renewal from these techniques is a skilled operation and unless a farmer has prior experience it will pay to seek professional advice.

**Brassica forage**

In spring, check existing vegetation for lucerne flea, redlegged earth mite and cutworm prior to herbicide application. Control of these pests is essential before sowing. To ensure maximum effectiveness of any insecticide application apply it at least three days before applying a herbicide. In spring, slugs can be a problem with direct drilling in high rainfall areas (>700 mm). Monitor for lucerne flea, redlegged earth mite and cutworm for at least the first two weeks after sowing. Spray if rate of seedling loss is economically damaging.
Methods of insect control

There are several methods of control.

**Cultural control**
Cultural control includes several methods.

- Choice of cultivars is important. For example, Victoca ryegrass resists root-feeding cockchafers better than other ryegrasses, but will succumb eventually. Phalaris is least susceptible to damage.

- Soil cultivation exposes pests and disrupts life cycles if done at the right time (winter feeding pests will be deep in the soil by spring).

- Heavy grazing tramples pests (but compacts soil).

- Heavily grazed pasture is less attractive to corbie moths laying eggs in summer or armyworm moths laying eggs in spring (but more attractive to cockchafer beetles laying eggs in summer).

- Long fallow periods deprive pests of food (but young weeds in fallow may attract cutworm moths to lay eggs in spring).

- Time of planting—autumn sown pasture is exposed to winter feeding pests like corbie and cockchafers if it does not follow a summer fallow.

- Lack of irrigation stresses forage plants and makes them more attractive to insect egg laying or feeding.

- Rain and irrigation wash the eggs of some pests from leaves while fungal infections spread in humid conditions and droplet splashes

**Biological control**
Biological control includes natural enemies such as predators, parasites and diseases. Most that are relevant to pasture and forage plants occur naturally and do not need to be introduced annually. The effectiveness of biological control can be severely reduced by using broad spectrum insecticides. Some cultural practices may also impair the efficiency of natural enemies.

**Chemical control**
Insecticides include synthetic chemicals, chemicals of botanical origin and products derived from viruses, bacteria and fungi. The latter products tend to be very specific and less disruptive of biological control but have short shelf lives, indicating the need to use fresh product. Chafer Guard™ is one of these.

Pesticides are generally a mixture of an active constituent and various additives. These other materials are needed to ensure the correct distribution of the active material on the crop or pest. The total mixture is called the formulation. Formulations suitable for spraying on pastures may be water-soluble materials, emulsifiable concentrates, wettable granules or wettable powders. Other formulations not used as sprays include granules, dusts and baits. These may be applied by a variety of methods.
Tasmanian Pasture & Forage Pests

Identification, biology & control
These two closely related insects differ mainly in the rainfall they tolerate and the time of year when they cause damage.

**Description**
The adult corbie and winter corbie are brownish–grey moths about 30 mm long. The front pair of wings bear an intricate pale pattern and the span across the outstretched wings is about 40 mm. Compared to corbie, the winter corbie (Fig. 1) has a more reddish–brown tinge to the wings. The female moth is larger than the male and has a noticeably larger abdomen due to her egg load. Eggs are ovoid in shape, less than 1 mm in diameter and creamy–white when first laid. Fertile eggs turn black within 24 hours (Fig. 2). Newly hatched caterpillars are about 3 mm long and pass through seven stages as they grow, reaching 60 mm in length by late spring. The body is smooth skinned and dark greyish or bluish–grey in colour (Fig. 3). The smooth, shiny head is dark reddish brown. Three pairs of legs are situated behind the head while four pairs of sucker–like false legs are present on the underside of the middle third of the body. The non–feeding prepupal stage, which occurs next, resembles the caterpillar but it is whitish in colour. The pupa is a shiny brown, relatively rigid capsule about 30 mm in length.

**Distribution**
The corbie is found only in Tasmania where it ranges widely over all the pastoral areas, except the Bass Strait islands. Outbreaks of damage may occur almost anywhere but are more common in the eastern and southern parts of the State, especially the lower Midlands and Derwent Valley. Caterpillars can survive in a wide range of soils and pasture types, including forage crops.

The winter corbie occurs in south–eastern mainland Australia but is less commonly a problem in Tasmania than the corbie. It is more prevalent in the higher rainfall pastures of the north–east and north–west, but also occurs in lawns in drier areas where watering supplements the natural rainfall.
Damage—pasture

Damage to pastures from both pests may occur by a direct loss in production from caterpillar feeding (Fig. 4), and/or by a shift in composition away from sown perennial species towards weeds and annual grasses caused by selective feeding and weed invasion.

A shift in composition is usually dramatic because large areas of bare ground are created. Weed seeds present in the bare areas germinate and grow without competition from the desired plants (Fig. 5).

Both pests feed on all sown perennial grasses and clovers, but generally they avoid flat weeds such as docks, dandelions and thistles. In order of tolerance, phalaris is the most tolerant to the activities of both corbies followed by cocksfoot, tall fescue and perennial ryegrass.

Pasture thinning occurs when the caterpillars reach about 30 mm in length and is first apparent in May–June for winter corbie and August–September for corbie. Because the larvae chew off pasture at ground level severe damage can result in a short time. The small bare patches that first appear rapidly increase in area and may coalesce to form extensive areas of denuded pasture by August (winter corbie), or October–November (corbie).

In rank pasture, corbie infestations may cause ‘haying off’ of the sward in late spring.

Care must be taken when direct drilling new pasture or fodder crops into old pasture in late autumn. Many failures have occurred due to the unrecognised presence of large numbers of corbie and winter corbie.

Assessing corbie numbers—pasture

Initially it is worth walking around paddocks at dusk several times during the expected flight times of the adult moths to observe locations where egg-laying may be taking place. From experience most farmers will know which paddocks or areas have been especially prone to damage in the past and these should receive special attention.

Heavy egg-laying does not necessarily mean an outbreak will occur. It is the variation in mortality factors in the young caterpillars, such as weather conditions and diseases, which are much more influential.
Out of 750 eggs laid by a female only two need to develop to adulthood to re-establish the populations at the previous year’s level. If four survive then the pest population will be double.

Caterpillar numbers can be assessed by digging a spade square sample to a depth of 200 mm in May–June for winter corbie and August–September for corbie. At these times the caterpillars will be 30–40 mm long and in shallow tunnels. A square should be dug every 20 paces in a line across the paddock. An average of more than two caterpillars per spade square will warrant treatment.

Another method is to use thin metal plates about 300 mm square (Fig. 6). Use a sharpened spade to skim off a 300 mm flap of pasture. Lay the plate on the bare ground. It is advisable to mark its position with a stick to easily relocate it. On the next day the plate is lifted and any corbies present will have re-opened neat holes which are easily counted. A minimum of about five plates per hectare should be laid. Three corbies or more per plate warrant treatment. Corbie tunnels are lined with silk (Fig. 9) while cockchafer tunnels are not.

Life cycle—pasture (Figs 7 and 8)

Adult moths emerge from the pupae between mid-January and mid-March (corbie) and late November and late December (winter corbie). The moths fly at dusk and mating occurs on the ground or on grass stems. The moths have a rapid erratic flight usually within a metre or two of the ground. Swarming occurs when numbers are high and their dusk activity may be audible as a low buzzing sound. After mating the female seeks suitable areas of sward into which to lay eggs. Rank cover is favoured since this offers some protection to the eggs which are dropped in batches on the ground or among plant litter. Each female may lay 750 eggs during her adult lifetime of one fortnight.

The eggs hatch in mid-autumn (corbie) and mid-summer (winter corbie), after a 5–8 week incubation period. The young caterpillars band together on the surface and spin communal webbing over themselves so they can feed in relative security. These webbings can be conspicuous on frosty autumn mornings. At this stage the caterpillars probably feed on fungus growing on litter at the base of plants but eventually they start to consume pasture
Corbie life cycle

- Pupae
- Moths flying
- Eggs hatch
- Caterpillars growing steadily
- Caterpillars enter soil
- Caterpillars under webbing
- Non-feeding prepupae
- Fully grown caterpillars
- Severe damage to pasture

Winter corbie life cycle

- Pupae
- Moths flying & eggs laid
- Eggs hatch
- Prepupae
- Fully grown caterpillars
- Severe damage to pasture
- Signs of damage to pastures becoming evident
at ground level. Bare patches the size of dinner plates appear in the pasture at this time. While living on the surface the caterpillars are highly vulnerable to dehydration as well as to predators such as beetles and spiders.

Not surprisingly the majority succumb to environmental hazards and this is a further reason why rank cover, by providing protection, promotes later outbreaks.

As the caterpillars grow they disperse from these congregations and in early winter establish individual silk-lined tunnels in the soil from which they emerge at night to feed on the adjacent pasture (Fig. 9). The caterpillars increase in size steadily through the winter and spring to become fully grown in early spring (winter corbie), or late spring (corbie). Their tunnels at maturity are about the diameter of a pencil. One or two silken ‘pathways’ often radiate a short distance into the pasture from the opening, which is itself protected by a silken canopy incorporating small bits of dung and straw.

When fully grown the caterpillar finishes feeding and securely seals its tunnel with a silken cap before entering the prepupal stage. The next stage is the pupa which is formed at the bottom of the tunnel in late spring (winter corbie), or mid summer (corbie). The moth then forms within the almost rigid skin of the pupa. Four to eight weeks later the pupa wriggles its way to the surface using spiny ridges on the abdomen and the moth emerges (Fig. 10). These emergences are usually well synchronised over large areas leading to swarms of moths in the late evening. The empty shell of the pupa protrudes from the tunnel after the moth leaves it (Fig. 11).

**Control—pasture**

Rank pasture is more attractive to the egg-laying moths than short pasture, whereas the reverse is the case for pasture cockchafer. Cocksfoot is less susceptible than ryegrass. Phalaris is resistant to all cockchafers and corbies. It is not yet known how effective the novel endophytes MaxP and AR1, plus others yet to be released, will be in protecting fescues and ryegrass from Tasmanian pasture pests.

Early treatment is better than late treatment but spraying as late as July–August for winter corbie or
October for corbie can still be worthwhile. If areas have been laid bare, consider closing them up beyond the pesticide's withholding period in order to encourage the desirable pasture species to recolonise these patches, thereby excluding weeds, which would otherwise invade.

When pasture cockchafer is also present, the optimum time for spraying is mid to late May when both pests can be controlled with the same insecticide.

Pasture should be grazed short prior to spraying. This will promote better coverage and penetration of the chemical. Spray only onto dry pasture, preferably late in the afternoon of a fine day. Otherwise the mixture will run off wet grass and be lost in the soil. Once dried on the sward (several hours), the mixture is not readily washed off by rain. Late afternoon spraying means the spray is at its most toxic that evening when the corbies emerge to feed. Spraying late in the day also reduces sunlight degradation of the insecticide.
Oxycanus grass grub

*Oxycanus antipoda*

This insect is related to corbie and winter corbie. It is a minor pest of pastures and in particular the north–west of Tasmania.

**Description**

The adult moth is large with a wingspan of 60–75 mm. The front pair of wings are greyish brown overlain with a distinctive pattern of irregular greyish markings. The hind wings are paler (Fig. 12). Female moths are slightly larger and paler than males.

The tiny eggs are elliptical in outline and less than 1 mm in diameter. When first laid they are creamy white but turn blackish within a few hours. The newly hatched caterpillar is about 3 mm long and has a pale grey body and a brown head capsule. The body darkens in colour as the caterpillar grows. Full size is reached in late autumn or early winter when the caterpillar is about 70 mm long and 7 mm wide and is dark greenish–brown in colour. It resembles a very large corbie (Fig. 13). The non feeding pupal stage, which rests in the soil, is a shiny brown rough capsule, 30–40 mm long and 7–10 mm wide.

**Distribution**

Oxycanus grass grub occurs in south–eastern Australia and the south–western corner of Western Australia. In Tasmania it is widespread in pastoral areas but is only considered a pest on the Bass Strait Islands and on the north–west coast. It seems favoured by high rainfall.

**Damage—pasture**

The first evidence of infestation is small mounds of soil produced by the tunnelling activity of young caterpillars in late summer (Fig. 14). During autumn, infested pastures produce noticeably less growth than those not infested, often leading to a serious shortfall in winter feed.

Because caterpillars prefer to feed on grass a trend to clover dominance is usually apparent by spring. It is rare to see complete removal of pasture due to this pest since caterpillar densities are usually much lower than those achieved by corbies. However, severe damage has been reported from King Island. Localised outbreaks
seem to occur once or twice a decade at irregular intervals.

**Life cycle—pasture**

The life cycle lasts two years. Moths fly shortly after sunset in late autumn, especially after an afternoon rain. In most years flights take place between the end of April and the first half of May. Loose swarms flying within 300 mm of the ground may be seen when the moths are abundant.

Mating occurs on grass stems near the ground. After mating the female may lay 1000–2500 eggs as she crawls or flutters over the pasture during the night. The eggs rest through winter and hatch in spring, 4–5 months after they are laid.

Young caterpillars inhabit horizontal, silk-lined runways among dead grass or litter on the surface of the soil.

By early summer the rapidly growing caterpillars construct silk-lined, vertical tunnels in the soil (Fig. 15). These are enlarged as the caterpillars grow, reaching a depth of 150 mm in autumn and up to 250 mm by August. The tunnel has a diameter of 10 mm and is usually branched towards the surface, having two concealed entrances about 70 mm apart.

Like the corbie, oxycanus grass grubs remain in their tunnels by day and emerge to feed at night. Caterpillars complete their feeding in late spring, roughly one year after hatching, and rest in tunnels as a non-feeding prepupa before entering the pupal stage in February or March of the next year.

The adult moth slowly develops within the pupa over a 2–3 month period deep in the soil. It is stimulated to emerge by rainfall late in autumn.

**Control—pasture**

Pastures showing poor autumn growth should be checked in May for the presence of this pest. Digging with a spade should reveal any caterpillars present. These can be distinguished from corbie or winter corbie by their larger size (about 70 mm) (Fig. 13), and their Y-shaped tunnel. Economic thresholds are uncertain but populations exceeding seven caterpillars in ten spade squares (equal to about seven caterpillars per square metre), should be regarded as damaging.
Diamondback moth

*Plutella xylostella*

Caterpillars of this pest only attack plants in the brassica family such as swedes, turnip, kale, vegetable brassicas and canola.

**Description**

The adult is a small, slender moth about 10 mm long (Fig. 16). It has a pale yellow stripe forming three coalescing diamonds along the back—hence the common name. It is sometimes known as cabbage moth but should not to be confused with the much larger cabbage white butterfly, *Pieris rapae*. Eggs are 0.5 mm long, flat and broadly oblong. They occur singly or in clusters of two to six (Fig. 17). The newly hatched caterpillar is roughly 1 mm long and initially feeds within a short, linear leafmine about 2–3 mm long before emerging to feed on the surface of foliage. These caterpillars have a collar of dark hairs behind the head absent in the similar sized caterpillars of the cabbage white butterfly. The mines appear as short, pale streaks on foliage and should not be confused with the much larger leaf mines that progress from lines into large blisters, which are caused by a tiny fly, *Scaptomyza flaveola*. Fully grown caterpillars are 12 mm long (Fig. 18), green and wriggle violently if disturbed, unlike young cabbage white butterfly caterpillars. They often drop on a silken thread. The pupa is 12 mm long, grey to green and rests in a silken cocoon with an open mesh attached to foliage or leaf debris (Fig. 19).

**Distribution**

This is an introduced species that has been present in Tasmania for over 100 years. It occurs across the state wherever cultivated, weedy or native brassica plants grow.

**Damage—forage**

Caterpillars of this pest feed almost exclusively on plants of the brassica family including weeds, forage, canola, broccoli and cauliflower. In forage the damage is by defoliation when caterpillars are abundant, or when infestation occurs very early in crop establishment such
as when eggs are laid on cotyledons as they emerge. Lower densities are not a problem in forage but are a problem in food brassicas because of the cosmetic effect of the presence of caterpillars and pupae. Spring sown forage is more likely to suffer damage than autumn forage as explained in the next section.

**Life cycle—forage (Fig. 20)**

Moths fly at dusk to lay eggs on the foliage of host plants. Eggs are often placed near leaf ribs. Eggs are white initially, but turn yellow and then grey as they develop. The head capsule of caterpillars that are about to hatch can be seen as a black spot at one end of the grey eggs.

Newly hatched caterpillars tunnel into the leaf to create a mine roughly 3 mm long and generally emerge after their first skin moult. One caterpillar may make several mines during this first stage.

They then feed externally to chew holes in foliage, sometimes leaving a tissue-thin layer of leaf epidermis—a window. After moulting their skin three times during growth, they spin a cocoon on sheltered foliage or debris in which they form the pupal stage. The pupa does not feed but transforms to the moth stage, which escapes from the end of the cocoon.

There are four generations annually. The fastest (summer) generation takes four weeks or more, whereas the slowest (winter) takes about three months. The pupa requires a higher temperature than the caterpillar to complete development. This may lead to synchronous emergence in spring. A spring generation may take five to seven weeks to complete.

Although this species can overwinter in Tasmania as slow growing caterpillars and pupae, much larger numbers migrate as moths from the mainland in spring and early summer. The size of these migrations varies considerably from year to year and greatly influences pest pressure in Tasmania. Progressive increases in the size of migratory flights through the 1990s perhaps reflected the increasing area of canola grown on the mainland. Record flights occurred in November 2002 (week 1) and 2003 (week 3) and coincided with rapid drying of mainland canola.
Damage is most apparent in spring when sudden influxes of migrant moths initiate a cohort of caterpillars that temporarily overpowers predators and parasites. By December, parasitism of caterpillars exceeds 80% where broad spectrum insecticides are not being used. By February, disease, parasitism, predation and perhaps a cessation of migration from the mainland, rapidly reduce the population to low levels until next spring. This is in contrast to the cabbage white butterfly which is sparse in spring and peaks in the autumn generation.

**Control—forage**

Three species of parasitic wasps, *Diadegma semiclausum*, *Diadegma rapi* and *Diadromis collaris*, that only attack diamondback moth were introduced to Tasmania around 1947 and now severely restrain the pest by late summer each year. These, and important general predators such as damsel bugs, *Nabis kinbergi*, are eliminated if broad spectrum insecticides are applied.

When supplementary control is required, it is best to use BT sprays (based on the bacterium, *Bacillus thuringiensis*). To be effective, these must be applied at high volumes, in late afternoon and when caterpillars are small. Older caterpillars are less susceptible to BT. More potent selective sprays are registered for food brassicas but not for forage. A BT strategy requires forethought and early monitoring. Broad spectrum insecticides such as synthetic pyrethroids and organophosphates will trigger an upsurge of aphids that are otherwise controlled by general predators such as brown lacewing, *Micromus tasmaniae*, and ladybird beetles, *Coccinella undecimpunctata* and *Coccinella transversalis*, as well as by parasitic wasps.

Heavy rain or irrigation limits the severity of infestations, as has been observed in irrigation trials in which infestation across plots was directly proportional to the volume of water applied. Water-stressed plants seem more attractive to moths and eggs are washed from foliage.
Diamondback moth life cycle

- Mainland moths immigrate
- Local caterpillars grow slowly
- Local moths emerge
- Caterpillars grow

Fig 20
Cabbage white butterfly

*Pieris rapae*

**Description**

The adult is a white (male) to cream (female) butterfly with 50 mm wingspan. The front wings bear one (male) or two (female) black spots and the hind wings bear one black spot each. The bases of the wings are black and this dark area is larger in cool environments. The eggs are slender, vertically ridged and sit singly and upright on foliage and are 1.5 mm high. The eggs of ladybird beetles are superficially similar but usually cluster and do not have ridges. Eggs change colour from white to yellow as they develop. Newly hatched caterpillars are roughly 2 mm long. Unlike the diamond back moth caterpillar they lack a collar of dark hairs behind the head. They grow via several moults into green caterpillars that are 30 mm long and are docile (Fig. 21), unlike the caterpillars of diamondback moth. In the final moult the pupa emerges and adheres to a substrate by fine silken strands at the hind end and a strand around the mid section. The pupa (Fig. 21) varies in colour blending with the substrate which may be a leaf, plant debris or other object. It does not feed but transforms its internal tissues into the adult form.

**Distribution**

This is not a native insect. It entered the state around 1940 having first appeared in Australia in Victoria in the previous year and not long before in New Zealand. It now occurs across the state wherever cultivated, weedy or native brassica plants grow.

**Damage—Forage**

Caterpillars of this pest feed only on plants of the brassica family including weeds, forage, canola, broccoli and cauliflower (Fig. 22). In forage the damage is by defoliation when caterpillars are abundant. Lower densities are not a problem in forage but are significant in food brassicas because of the cosmetic effect of the presence of caterpillars and pupae. Defoliation by caterpillars becomes most apparent from February to April as the third generation develops, but lesser leaf damage can be seen earlier in the season.
Life cycle—forage (Fig. 23)

Cabbage white butterflies are first seen on the wing in September. They lay eggs on foliage and die. By summer another cohort of butterflies emerges. There are perhaps three generations through the warm season. Butterflies become common in late summer. The autumn generation is often largest. This is in contrast to diamondback moth caterpillars that are common in spring but sparse by late summer.

Fully fed caterpillars of the autumn generation tend to wander metres from host plants in April and May looking for warm, sheltered sites in which to form overwintering pupae. However, in brassica food plants they may seek shelter in the heads of broccoli and cauliflower.

In other continents this species is known to migrate long distances. This may explain their sudden first appearance in large numbers in northern Tasmania in 1940. Whether migration across Bass Strait continues to be significant in determining annual levels of abundance is uncertain, but is less likely than for diamondback moth.
Control—forage

Three species of parasitic wasps, *Cotesia glomerata*, *Cotesia rubecula* and *Pteromalus puparum*, that only attack cabbage white butterfly were introduced into Tasmania around 1947. They now help restrain this pest. These, and important general predators of caterpillars such as damsel bugs, *Nabis kinbergi*, and spiny shield bugs, *Oechalia schellenbergii*, are eliminated if broad spectrum insecticides are used.

When supplementary control is required, it is best to use BT sprays (based on the bacterium, *Bacillus thuringiensis*). To be effective, these must be applied at high volumes in late afternoon and when caterpillars are small. Older caterpillars are less susceptible to BT. A BT strategy requires forethought and early monitoring. Novel selective sprays are registered for food brassicas but not for forage. Older broad spectrum insecticides such as synthetic pyrethroids and organophosphates will trigger an upsurge of aphids that are otherwise controlled by general predators such as brown lacewing, *Micromus tasmaniae*, and ladybird beetles, *Coccinella undecimpunctata* and *Coccinella transversalis*, as well as by parasitic wasps.

Do not use broad spectrum insecticides as these can eliminate the natural predators of this pest.
Armyworms

Southern armyworm, *Persectania ewingii*
Common armyworm, *Mythimna convecta*

The term 'armyworm' refers to the damaging caterpillars of two species of moths native to Tasmania and parts of mainland Australia. They are called armyworms because of their habit of mass movement of caterpillars when the insects occur in plague numbers. In Tasmania, common armyworm is far less common as a pest than southern armyworm. Moths of a third species, the inland armyworm, *Persectania dyscrita*, migrate to Tasmania occasionally but do not generate infestations.

Description

The young caterpillars are pale yellow or cream in colour. The older ones are usually marked with a variable pattern of grey, white and black stripes (Fig. 24) but may also be brown or greenish. Mature armyworm caterpillars are from 30–40 mm long. They are sometimes confused in the field with caterpillars of cutworms and native budworm, which rarely eat grass. Armyworms can be distinguished from the others by the markings on a ‘collar’ behind the head (Fig. 25). These markings consist of three parallel white stripes running back from the head. One is in the centre of the collar and the others are close on either side.

The distinction between the two armyworm species in the caterpillar stage is microscopic and subtle. The hairs above the spiracles on abdominal segments 3–6 (which bear fleshy false legs) lie near the top border of the dark side–stripe in southern armyworm but towards the middle of the dark side–stripe in common armyworm.

The moths (adult stage) of the two species are, however, easily identified by the colour of their forewings. The moth of southern armyworm (Fig. 26) is about 20 mm long and has a wingspan of about 40 mm. Its forewings are grey coloured with darker markings. The moth of common armyworm is also about 20 mm long with a wingspan of up to 43 mm. The forewings vary in colour from red–brown to yellow–brown and are speckled with small black dots; a darker patch with a small white dot is often visible near the centre of each wing. The hind wings of both species range from light to dark grey, becoming darker around the outer margin.
Distribution
Armyworms are found throughout the pastoral areas of Tasmania, but outbreaks are most commonly reported from King Island, the north–west, south–east and Derwent Valley. In some years infestations are restricted to certain districts, perhaps because winds and storms that carry moths from the mainland dump them locally.

Damage—pasture
Damage to pasture is caused by the caterpillar stage which chews the leaves of legumes and grasses, bites the stems of grasses below the seed heads or severs the stems at the base. Young caterpillars will damage emerging grass during spring but the older caterpillars do most damage in summer. The amount of damage caused over a given period is often dependent on weather conditions. A succession of cold nights restricts caterpillar activity and therefore little feeding occurs. Cereals are more susceptible to damage than pastures. Pastures can be fouled by very high populations and the forage may be rejected by stock.

Life cycle—pasture (Figs 27 and 28)
Although southern armyworm, unlike common armyworm, does persist in Tasmania through winter, serious infestations are usually derived from immigration of moths from the mainland when the preceding climatic events foster breeding in the mainland source areas. Although moths of both species immigrate in spring, those of common armyworm are rare in spring and more common in summer.

Moths derived from locally overwintered southern armyworm caterpillars emerge in late spring, after most immigrant moths have arrived. Cooler temperatures in Tasmania delay their growth compared to mainland cohorts. That is why the flight activity of southern armyworm forms two peaks centred on mid October (immigrants preceding locals) and early March (local emergences). The moths are active at night.

Typically caterpillars of southern armyworm hatch in spring from eggs laid by immigratory moths. Newly hatched caterpillars undergo several moults. The caterpillars reach full size by January, often as susceptible cereal crops dry off. The fully fed caterpillars dig into the soil and form chambers in which they rest as shiny, brown pupae. Moths emerge from those pupae in March, lay eggs and give rise to a generation of caterpillars that grows slowly through winter and usually experience high mortality on wet ground.

The generation time averages 20 weeks during spring and summer and 32 weeks in autumn and winter. The summer duration is increased by a prolonged pupal resting period in the soil, which prevents the moths from emerging before rain and fresh grass. Generations can overlap. Moths and developing caterpillars have been found in every month of the year.

Southern armyworm moths lay their eggs between the sheaths of pasture grasses, cereals and maize, but laying also occurs at any suitably sheltered site at the base of a sward. Migratory southern armyworm moths seem to prefer to lay eggs in pastures locked up for hay rather than short grass. Common armyworm moths frequently lay eggs in dead twisted foliage.
Rarely, the winter generation of caterpillars starts at a high level in autumn and fares well in a dry winter. Such an event occurred in the Fingal Valley and northern Midlands in 2002 when an unusually late and abnormally large cohort of moths appeared in April (a month after the average date) to give rise to damaging numbers of large caterpillars in September when the usual migratory moths were starting to arrive from the mainland.

It is not known whether some of the moths emerging in autumn emigrate to the mainland but some certainly remain in Tasmania to generate winter infestations of caterpillars, as described above.

It is doubtful that significant numbers of common armyworm, if any, overwinter in Tasmania. Moths arrive from July to February. Fully grown caterpillars of common armyworm are most typically seen in February and March. Hence, the peak activity of common armyworm caterpillars usually occurs a month or so after southern armyworm.

**Southern armyworm life cycle**

![Image of Southern armyworm life cycle]

Fig. 27
Control—pasture

Predicting outbreaks:
Since the mid 1950s the Department of Primary Industries and Water has monitored the size of annual immigrations of southern armyworm by catching moths in one or more light traps. The current trap is located at Devonport. When catches are well above average a warning is issued. However, low numbers of moths can still initiate infestations if their eggs and caterpillars fare better than usual, while large numbers of moths may fail to initiate infestations if their caterpillars perish in inclement weather. Irrespective of forecasts, farmers should examine their pastures and cereals regularly for signs of armyworms.

However, common armyworm moths are not reliably detected in light traps. Fermentation traps are effective in monitoring flights of these moths but no permanent fermentation traps are operated in the state.

Detection of infestations:
Look for leaves with holes and scalloped edges. A line of holes may emerge as leaves unfold. Check for the presence of armyworms in a pasture by looking closely at the bases of the plants where the caterpillars can usually be found sheltering during the day in a curled–up position. Alternatively use a torch to look for caterpillars at night on the stems and seed–heads of the plants. Another sign of an armyworm infestation is the presence of lopped grass seed heads.
To spray or not
Armyworm numbers in pasture can reach several hundred per square metre in severe infestations. Pastures, unlike cereal crops, are able to sustain high armyworm populations in late spring without significant damage. The main problems usually result from the fouling of pasture, especially hay paddocks.

The size of the caterpillars is also important. It is not worth spraying a pasture because of signs of a few small armyworms. The young caterpillars tend to spend most of their time at the base of the sward where they are difficult to see and where they are less likely to be killed by insecticide. The best strategy is to wait until the caterpillars are well grown when they will be more active in their feeding and therefore more likely to pick up a lethal dose of insecticide.

Damage—forage and grain
Forage plants in the brassica family are not at risk from armyworm. Armyworms favour plants in the grass family, including cereals. Annual grasses are susceptible to attack if a dense stand is present at the time of spring moth flights when eggs are laid into grass foliage. They are also at risk if caterpillars move from adjoining pasture.

Damage to cereal plants is caused by defoliation but more importantly, by loss of seed heads. Mature caterpillars bite the stems below seed heads or sever the stems at the base. Young caterpillars will damage young cereals in spring but the older caterpillars do the most damage by climbing up the stems of cereals to feed, usually at night—90% of leaf consumption occurs during the final stage in caterpillar growth. Look at the bases of plants for caterpillars sheltering during the day. Cereal crops are much more sensitive to damage than pasture grasses, especially if large, voracious caterpillars are forced by drying foliage to feed on grain heads and the upper stems, which hold remaining moisture. As a rough guide, a single head of grain dropped in each square metre is equivalent to a grain loss in cereals of 10–15 kg/ha. Further, since each mature caterpillar may drop one head of grain, the number of caterpillars per square metre can give an idea of potential loss.

The foliage and silks of maize sustain armyworm caterpillars but leaf loss is rarely significant. Maize is generally grown after the main moth flights threaten young plants.

Life cycle—forage and grain
See pasture section. Armyworm moths tend to lay eggs in dense grassy foliage inserting clusters of eggs between the stem and sheath. They are less likely to establish in emerging grass or cereal and the caterpillars will require several weeks to reach a size where leaf consumption accelerates dramatically. In the meantime, there are opportunities for natural controls such as predators, parasites and weather to reduce caterpillar density.

Control—forage and grain
A handful of large caterpillars per square metre can rapidly cause serious grain loss and insecticide should be applied if such a situation is developing.
True cutworms
Brown or pink cutworm, Agrotis munda
Common cutworm or bogong moth, Agrotis infusa

Description
Two species of the genus Agrotis are recorded as regular pests in Tasmania. A third, the variable cutworm, Agrotis porphyricollis, is common in Tasmania but rarely causes economic damage. Moths of a fourth species, the black cutworm, Agrotis ipsilon, are rare vagrants blown from warmer states where they are a pest.

Moths of Agrotis species characteristically hold their wings flat above the body with left and right forewings completely overlapped rather than in an inclined position with left and right not overlapped as in armyworm and budworm moths.

The moth of common cutworm is also known as bogong moth and was well known to the natives of Victoria and nearby highland districts as a source of summer food. It is a large, dark grey moth with a wingspan of around 45 mm (Fig. 29) or a moth 35 mm long when the wings are in repose.

The moth of the brown cutworm (also known as pink cutworm) is grey and smaller having a wingspan of around 36 mm (Fig. 30) or 20 mm long when the wings are in repose.

Eggs of common cutworm are dome–shaped, vertically ridged, 0.7 mm diameter and 0.4 mm high. They are laid in clusters in the soil or, if it is too wet or dry, on foliage and stems. Eggs of brown cutworm are 0.6 mm diameter.

The caterpillars grow from 1 mm to 40 mm long (brown cutworm) or to 50 mm long (common cutworm) (Fig. 31). Hatchling caterpillars are cream with faint grey tones and bear sparse, short hairs with expanded rather than pointy tips. Older caterpillars are counter–shaded grey to dark grey on top with a dirty cream underside. They characteristically curl tightly when disturbed. Brown cutworm caterpillars may have a pink tinge.

The pupa of the common cutworm is a shiny, brown capsule 20 mm long. It rests in a soil chamber, 20–150 mm deep, from which the moth emerges. The pupa of brown cutworm is around 16 mm long.
The pupae of these two true cutworms have a rough cuticle around the spines on the tip of the abdomen whereas armyworms have smooth cuticle around these spines.

**Distribution**

These are native species that disperse widely across Australia by migratory flights in spring. Cutworms disperse across Tasmania.

**Damage—forage**

Look for moths at house lights and in recently cultivated paddocks. Moths prefer to lay eggs in moist, recently cultivated soil. Cutworm caterpillars prefer young, broad-leafed plants, including brassica forage and lucerne, but sometimes damage emerging grass such as new pasture following old pasture or minimum tillage in spring. The first signs of an infestation are numerous small holes in foliage chewed by tiny caterpillars. Larger caterpillars cut stems of tender plants causing them to fall—hence the name cutworm. They feed by night and shelter in the soil by day. Seedlings (including vegetable transplants) are particularly susceptible to cutting by older caterpillars that have established on young weeds during fallow periods. Damage occurs mostly in late spring to summer.

**Life cycle—forage (Figs 32 and 33)**

The two cutworm moths are native species with core winter breeding areas on the Australian mainland. Three overlapping generations of common cutworm occur through the cool months in these core areas of the mainland. Although a few may overwinter in Tasmania, most migrate to Tasmania in spring. Winter infestations have never been recorded in Tasmania. The numbers reaching Tasmania annually depend mostly upon preceding conditions through winter in their core breeding areas. Winds dump them anywhere from Sydney to Hobart or even on the sea. Many common cutworm moths shelter in northern Tasmanian high country scree slopes, such as Mt Barrow. There they await a return flight to the mainland in autumn when the new growth of broad-leafed plants in the black soil plains of NSW provides suitable habitat. However some common cutworm moths do not rest through summer in the Tasmanian high country but lay eggs in agricultural districts where food-plants are emerging.

The brown cutworm has 2–3 overlapping generations through the warmer months in core mainland breeding areas. The moths also breed in lowland Tasmanian crops in spring and summer but lack the summer resting strategy and two-way migration of common cutworm moths. However, moths emerging in autumn from a Tasmanian summer generation may attempt to return to the mainland.

The eggs incubate for three weeks in September, two weeks in October or around ten days in November. The caterpillars require 4–12 weeks to grow to full size. Initially the newly hatched caterpillars stay on foliage during the day but soon become reclusive and hide in the soil by day to feed at ground level on foliage at night or dull days. Fully grown caterpillars typically occur in November—December for brown cutworm and December—January for common cutworm. Pupation can pass in as little as three weeks but most moths do not emerge from the summer pupae until February.
It is not known whether most of the summer breeding generation of either species attempt to migrate to the mainland for winter or attempt to breed locally by laying eggs in autumn. If the latter occurs, they have never caused economic infestations. It is not known whether the summer—resting, montane moths migrate northwards in autumn.

**Control—forage**

Examine paddocks, including pasture, for older cutworm caterpillars and apply insecticide or cultivate soil to reduce their density before planting forage or new pasture. Clean tillage also removes weed seedlings that help new populations of young caterpillars to establish. Weeds used by cutworm include fat hen, dandelion, sowthistle, dock, shepherd’s purse, morning glory and others. Very young caterpillars (2 mm) may be found on foliage during the day but older caterpillars hide by day in the surface soil and trash at the base of plants. Monitor for two weeks after sowing forage brassicas in spring. Heavy rainfall and irrigation restrains cutworms, possibly by drowning eggs and young caterpillars. As with diamondback moth, dryland forage is more prone to infestation by cutworms than irrigated forage.

**Common cutworm life cycle**

![Common cutworm life cycle diagram](image-url)
Brown cutworm life cycle

- Mainland moths immigrate & lay eggs
- Moths emigrate or lay eggs locally
- Pupae in soil
- Caterpillars grow
- Pest absent

Fig. 33
Chevron cutworm

*Diarsia intermixta*

**Description**
This moth is of similar size to armyworm and brown cutworm moths with a wingspan around 38 mm. Male moths are fawn or dull orange (Fig. 34) while females are dull purple. Like true cutworms of the genus *Agrotis* they hold their wings flat on the back in repose. Eggs are dome-shaped, 0.65 mm diameter, 0.45 mm high and bear 28–38 vertical ridges. They are laid on foliage in clusters of 10–90 eggs but with small spaces between eggs. Caterpillars grow from 1 to 30 mm long (Fig. 35). They are counter–shaded, dark brown to black above and pale below. A series of paired, black, short oblique marks (like chevrons) occur along the back and a pale mark traverses the rear end. The pupa is a shiny brown capsule that rests in a chamber in the soil. The moth emerges from the soil.

**Distribution**
This minor pest occurs across the state with records dating back to at least 1918.

**Damage—forage**
Unlike true cutworms, *Agrotis* species, the caterpillars of chevron cutworm do not characteristically cut young plants at the base. They chew on foliage and occasionally defoliate brassica forage and chew into tubers and other underground storage organs (Fig. 36). Brassica forage is most frequently damaged in autumn and winter. Caterpillars also infest the silks of maize but are unlikely to cause economic damage. The chevron cutworm also feeds on fodder beet, carrots, potato, buckwheat, sweet corn, blackcurrant, ginseng, clover, brassica weeds, docks, chickweed and capeweed.

**Life cycle—forage** (Fig. 37)
There are perhaps three generations annually. Moths fly in any month but peak flights are most often in spring and autumn or, in some years, in summer rather than autumn. Flight periods vary considerably from year to year and sometimes appear to involve three peaks. Eggs are laid on foliage. They incubate for 2–3 weeks in spring but only 7–10 days in summer. Caterpillars feed exposed
on foliage but may shelter near the bases of plants during the day. They require several weeks to complete six stages after which the non-feeding pupal stage occupies 2–3 weeks. At a mean temperature of 19ºC the life cycle is 7 days egg, 40 days caterpillar and 18 days pupa, which is shorter than other noctuid moths such as cutworms and armyworms.

**Control—forage**
Control is seldom necessary. Many caterpillars succumb to parasitic wasps. Severe damage is often limited to small areas within a paddock. Apply an insecticide registered for caterpillars if severe defoliation is developing, but remember that broad spectrum insecticides harm the natural enemies of this and other pests.
**Green cutworms**

*Neumichtis nigerrima, Neumichtis spumigera* and *Neumichtis saliaris*

**Description**
The moths of these three species have wingspans of 36 mm, 38 mm and 34 mm respectively. *N. nigerrima* is a mostly black moth while the other two species are mostly brown. Unlike chevron cutworm and true cutworms of the genus *Agrotis*, they hold their wings steeply inclined, not horizontally, over their backs in repose. Eggs are dome–shaped, 0.65–0.71 mm in diameter, bear 17–31 vertical ridges and are laid singly on foliage. Caterpillars are either green or brown (Figs 38 and 39) and grow from 1 mm to 35 mm long. They all bear a pair of small but distinct white spots on the top of the hind end. These are more conspicuous than any other pairs of faint pale spots along the back (Fig. 38). The pupa is a shiny brown capsule that rests in a soil chamber from which the moth emerges.

**Distribution**
These very minor pests are native species. They occur across the state.

**Damage—forage**
Unlike true cutworms, *Agrotis* species, the caterpillars of green cutworm do not cut young plants at the base. They chew on foliage. Brassica forage is susceptible to attack in autumn and winter, but green cutworm caterpillars also feed on broad–leafed weeds and vegetable crops such as lettuce and carrots. One or two species of green cutworms often occur in mixed populations with the chevron cutworm.

**Life cycle—forage**
There may be two generations annually. Moths fly mostly in spring and autumn. Eggs require 2–3 weeks incubation in spring and autumn. Caterpillars require several weeks to complete six stages, but much longer through winter, after which the pupal stage occupies 2–3 weeks in the warmer months. At 19ºC the life cycle is 8 weeks for *N. nigerrima*, which is fast relative to other noctuid moths such as cutworms and armyworms, and 11 weeks for *N. spumigera*. 
Control—forage
Control is seldom necessary. Many caterpillars succumb to parasitic wasps. Severe damage is often limited to small areas within a paddock. Apply an insecticide registered for caterpillars if severe defoliation is developing, but remember that broad spectrum insecticides harm the natural enemies of this and other pests.

Fig. 39 Caterpillars of diamondback moth (top) and green cutworm, *Neumichtis nigerrima* (bottom). These may occur together on forage brassicas
Grass anthelid caterpillar

*Pterolocera amplicornis*

**Description**

The adult male is fawn with dark wing veins, has a 30 mm wingspan and very feathery antennae. Female moths are almost wingless, dark and 20 mm long. Caterpillars have dense, felt–like hair. They are brown and black with two pairs of conspicuous yellow spots on each segment of the body (Fig. 40). The pupa forms in a flask–shaped, silken cocoon within a vertical, silken–lined tunnel in the soil.

Other hairy caterpillars that may cause concern because they are occasionally abundant in rough pasture are those of the arctiid moths (Fig. 41), *Phaos interfixa* Walker and *Spilosoma glatignyi* (Le Guillou). They are not likely to cause economic injury to the pasture.

The spiky, black caterpillar of the meadow argus butterfly is occasionally abundant in pasture in March. It feeds on plantain weeds but not on grass or clover.

**Distribution**

The grass anthelid moth is a native species that is wide spread in open habitats. Sporadic, dense infestations of the caterpillars have been recorded from the northern Midlands, Fingal Valley, Midlands and Derwent Valley.

**Damage—pasture**

Pasture infestations described as heavy often result in little economic damage. Hundreds of caterpillars per square metre once caused light damage to Yorkshire fog grass and perennial ryegrass over 15 hectares. Such dense populations often succumb to disease epidemics. The caterpillars are most apparent in spring.

**Life cycle—pasture (Fig. 42)**

There is one generation annually. Female moths cannot fly and lay eggs in batches. Male moths fly in autumn. Mature caterpillars appear by spring. Summer is spent underground in the non–feeding pupal stage.
Control—pasture
Winter waterlogging, diseases and beneficial insects usually restrain populations of grass anthelid. In extreme situations, an insecticide registered for control of caterpillars in pasture could be used but such broad spectrum insecticides should be used sparingly as they will severely disrupt the natural enemies of many other pests.

Grass anthelid caterpillar life cycle

Fig. 42
Blackheaded pasture cockchafers

Acrossidius tasmaniae
Acrossidius pseudotasmaniae

Description
The adult stage is a shiny, dark brown or black beetle 10–12 mm in length (Fig. 43). Eggs are pale creamy yellow in colour and about 2 mm in diameter. Clusters of 10–50 are laid about 100 mm below the soil surface. The eggs hatch into C–shaped, white or greyish–white grubs with a shiny brown or black head capsule and three pairs of legs at the front end (Fig. 44). When fully grown in winter they are about 15 mm long. The pupa, is whitish, soft–bodied and about 10 mm in length.

The grubs make vertical holes in the soil (Fig. 45.), about the diameter of a pencil and remain below ground during the day. At night they emerge to feed on the foliage of clovers, grasses and some weeds, often biting off pieces of food which they store in their tunnels for later consumption.

Distribution
These pests are native species. They are widespread and serious pests of pasture in Tasmania, especially in coastal areas and on well–drained, light soils. Almost all grazing properties in the state carry populations of one or other of the two species involved. There is some evidence that Acrossidius pseudotasmaniae is more abundant in lower rainfall areas and that this species is restricted to Tasmania. Acrossidius tasmaniae also occurs in New South Wales, Victoria and South Australia, and has been introduced to New Zealand where it is known as the ‘Tasmanian grass grub’.

Damage—pasture
The damage caused by these pests results from feeding by the grubs and is usually in the form of bare patches which appear in the pasture from mid–autumn to late winter. Sown perennial species seem to disappear first. Closer examination of the soil surface will reveal tunnel entrances which are usually next to a low mound of thrown–up soil. In heavy infestations this soil can partly bury much of the pasture. Unlike corbie tunnels there is no silken lining associated with the holes of the pasture cockchafer (Fig. 45).
Heavy rain and frosts deter the grubs from feeding and damage can develop rapidly on the return of warm weather after extended periods of such conditions. Heavy grazing in autumn or winter can aggravate damage because the new regrowth is within easier reach of the grubs.

Pasture cockchafers survive best on light-textured, well-drained soils. It is common for infestations to be confined to well-drained crests in paddocks, although continuous infestations over many hectares are prevalent in bad years.

High concentrations of grubs and damage are commonly seen around trees, along fence lines and close to other obstacles. This is probably because flying beetles collide with these and lay their eggs in the vicinity.

Assessing damage—pasture
The best month to check for grubs in the paddock is May. Pay close attention to sandy or loamy paddocks that had bare patches in January or February, or a history of pasture cockchafer attack. A spade-square sample to a depth of 200 mm is a useful unit to measure with and a series of these should be taken diagonally across a paddock, say, every 20 paces. If average numbers exceed six grubs per spade-square then action should be taken.

Life cycle—pasture (Fig. 46)
The adult beetles emerge from the pupal stage in the soil in mid to late summer. Swarming flights are common at dusk on calm, warm evenings and the beetles are strongly attracted to light. These swarming flights serve to bring the sexes together for mating and assist dispersal. It is possible that beetles can fly several kilometres although the majority probably lay eggs in the same paddock from which they emerged.

The eggs hatch after 3–4 weeks and the grub passes through three instars, or growth stages, to become fully grown in the late autumn or winter. Feeding finishes in early to mid-spring and a non feeding prepupa, yellowish in colour, is formed prior to the pupa in early summer.

Control—pasture
Short, open pasture is more attractive to the egg-laying beetles than rank pasture whereas the reverse is true for corbies and redheaded pasture cockchafer. A high clover component also favours pasture cockchafer.

Cocksfoot and fescue grasses have good tolerance to surface and root-feeding cockchafers. Phalaris is resistant to all cockchafers and corbies. It is not yet known how effective the novel endophytes MaxP and ARI, plus other new novel endophytes yet to be released, will be in protecting fescues and ryegrass from cockchafers.

When corbie is also present, the optimum time for spraying is mid to late May when both pests can be controlled with the same insecticide.

For best results sprays should be applied before the end of June. Timing is important, because grubs do not come to the surface and feed every night. During periods of warm, dry weather or during a cold, frosty period they may not feed for several days. Feeding seems to be most intensive on moist nights after rain.
Therefore the best time to spray is at the end of a dry spell, a few hours before rain is anticipated. Allow at least four hours for the spray to dry and adhere to foliage.

Failures reported in relation to control attempts on this pest are often the result of poor timing of spray application.

In some areas in some years, pasture cockchafers are attacked by a pathogenic fungus which often causes population numbers to drop below damaging levels. This fungus, *Cordyceps gunnii*, is widespread in the State but cannot be relied upon as a control measure at the present time. In the latter stages of infection grubs turn a pinkish colour and become mummified in the soil. In the spring a long, slender fruiting body grows out of the immobile larva to reach the surface where it forms spores ready to infect the next year’s grubs (see Fig. 102, page 83).
Blackheaded cockchafer life cycle

- Pupae in soil
- Beetle flights & egg laying
- Non-feeding prepupal stage
- Young grubs developing
- Large grubs causing damage to pastures

Fig. 46
Redheaded pasture cockchafer
*Adoryphorus couloni*

**Description**

The adult stage is a stout, shiny black beetle about 15 mm long (Fig. 47). The ovoid eggs are 2–3 mm in length and pearly–white in colour. The grubs are soft–bodied and white with three pairs of yellowish legs, a hard, reddish brown head capsule and the posterior quarter of the body is a little swollen (Fig. 48). The head capsule appears rough or matte in contrast to the shiny capsule of blackheaded pasture cockchafer. The body wall is transparent. The white colouration of the grub derives from fatty tissue under the skin and the greyish appearance of the rear end results from soil in the gut. The posterior end of the grub is more opaque than in blackheaded pasture cockchafer (Fig. 44). When at rest the body is curved in the shape of a letter C. Grubs are sometimes called ‘curl grubs’ or ‘white grubs’. Newly hatched grubs are only 5 mm long but when mature, are robust and up to 30 mm in length. The grubs are less active when exposed than are blackheaded pasture cockchafer grubs, which retreat rapidly if placed on a spade.

The pupa, about 15 mm long, is soft–bodied and pale yellow–brown in colour.

**Distribution**

This pest is a native species. Before 1987 the redheaded pasture cockchafer occurred on King Island, but not Flinders Island, and across northern Tasmania as far south as Woodbury in the central Midlands. The cold high plateau around Oatlands slowed its spread into southern Tasmania. It now occurs in the southern Midlands, Derwent Valley, Bothwell district, Hobart, South Arm and other localities including Flinders Island. Its status in the Huon Valley and Channel is uncertain. It has not been recorded at altitudes above 200 metres.

Elsewhere this pest is distributed from southern New South Wales through Victoria to south–eastern South Australia. Prior to 1987 it spread to New Zealand where it now occurs on Banks Peninsula.
Damage—pasture
The adult beetle stage does not cause damage. However, the grub feeds on roots and humus in the root zone, usually within 50 mm of the soil surface. Because the grub spends its entire life feeding underground the effects of this pest can be difficult to appreciate unless populations are extremely high.

Damage is most serious in late autumn and is caused by two factors: severing of the roots during feeding and physical disruption of the roots during underground movement of the grubs. High numbers of the grubs undercut the pasture plants, severing them from their roots. This promotes uprooting by stock and birds (Fig. 49) which, during a dry spell, leads to plant death from moisture stress since the plants cannot tap soil moisture held at depth. Underground movement of grubs also makes the pasture feel spongy underfoot.

Grasses with weak, fibrous roots such as ryegrass are especially vulnerable to damage. In a mixed sward the ryegrass component is often uprooted completely by stock activity. The resulting gaps in the pasture allow fast-growing annuals such as barley-grass, storksbill and capeweed to establish. This trend to weediness is often the only symptom that is clearly visible unless the soil is turned over. Subterranean clover usually re-establishes itself satisfactorily after attack by this pest, provided sufficient soil seed reserves exist.

Damage first appears in late March and may be severe by May or early June when aggravated by bird activity. The forest raven is the main culprit in most regions (Fig. 49). Low soil temperatures in winter reduce the activity of grubs before more active feeding resumes in late August. Damage to pastures in spring is usually less severe than in autumn. There are several reasons for this: grub numbers will have declined through natural mortality; pathogens accelerate grub death when the soil temperatures are warmer; and the plants compensate much better because the spring growth flush enables them to re-establish a satisfactory root system given sufficient moisture is present.

Although the redheaded pasture cockchafer takes two years to complete its life cycle, the occurrence of overlapping generations means that grubs can be present every year. Usually, one generation is more abundant than the alternate generation so that, in any particular district, damage is seen every second year. The years of severe infestation are not necessarily the same for the Midlands and the north–west coast.

Life cycle—pasture (Fig. 50)
Adult beetles emerge from the soil at dusk in late winter and early spring (from the end of August until mid–October). Swarming flights which occur at this time help to disperse the beetles widely. During the night the female beetle tunnels into the soil to lay eggs, singly or a few at a time, at a depth of up to 80 mm. Each female may lay up to 25 eggs in her lifetime.

The eggs hatch in late spring, 6–8 weeks after being laid. The young grub then passes through three stages. The first two of these are passed rapidly so that by late summer–early autumn the final (third) stage is reached. This stage is the most damaging and feeds for almost 10 months. Feeding is intense during the autumn but is interrupted by the onset of cold weather in June.
At this time the grubs may dig to warmer depths in the soil and stop gaining weight. Nevertheless, the grubs do feed during spells of mild weather in winter. Active feeding and weight gain resume in early spring and continue until early summer when grubs reach full maturity and finish feeding. They then leave the root zone and dig deeper into the soil, often up to 200 mm below the surface, where they form a small cell by compacting the soil around them. Here they expel their gut contents, their body fat turns a yellowish colour and their body takes on a J-shape. This is the prepupal stage. After two weeks the grub’s skin is split off to reveal the pupa. The pupal stage lasts 6–8 weeks before the beetle emerges in February–March. However, the beetle remains in the pupal cell as a sexually immature adult for about six months until it digs its way to the surface to engage in locally synchronised flights in late winter–early spring. The beetles do not feed and rely on energy reserves laid down during the larval stage.

**Control—pasture**

Spraying will not control this pest in pasture. There are no synthetic insecticides that give effective, economical control of redheaded pasture cockchafers since their subterranean feeding habits create difficulties in the penetration and stability of chemicals. This contrasts to blackheaded pasture cockchafers, which feed above ground and are therefore susceptible to synthetic insecticides.

In existing pastures, management practices must be integrated and aimed at limiting damage as much as possible. When damage is noticed in mid–autumn, stock should be removed, particularly from ryegrass dominant pastures, and the paddock spelled until late winter. This will help prevent all the ryegrass being uprooted by grazing animals and maintain maximum leaf area needed to re-establish root growth. Although supplementary feed may have to be bought to carry displaced stock over winter, the expense will usually be repaid in superior spring production and the maintenance of desired botanical composition in the infested paddock.

Diversify feed sources on the farm away from total dependence on ryegrass pastures. This might entail sowing some autumn forage crops, storing extra hay in anticipation of a winter feed shortage aggravated by pests, or sowing down some areas of cockchafer tolerant pastures. Such pastures could include phalaris, cocksfoot or tall fescue with a small percentage of ryegrass mixed in. Dairy farmers are not advised to mix grasses in one paddock. Lucerne and oats are also relatively tolerant of cockchafer attack. It is not yet known if the novel endophytes MaxP and AR1, plus other new novel endophytes yet to be released, will protect some new fescues and ryegrass from root-feeding cockchafers.

If conditions are not too boggy, rolling of the infested pasture can be beneficial since this helps the sward or seed re-establish contact with the soil and may kill grubs close to the soil surface.

Large numbers of cockchafers can be destroyed by the trampling effect of block grazing stock. This should be done before the end of May while grubs are still close to the surface. This strategy should be employed when large numbers of grubs are first noticed, even if it means upsetting a carefully planned rotation, since the benefits in the longer term will be substantial.
A carefully planned rotation could include this strategy, but would not be advised if the grazing animals were milking cows.

A biological insecticide Chafer Guard™ (previously BioGreen) is available for redheaded pasture cockchafer. It is a potent strain of a native soil fungus, *Metarhizium anisopliae*, formulated as a granule that is mixed with seed, but not fertiliser, when sowing pasture. It causes fatal infections in the pest. It is best used as a preventative strategy whose benefits accumulate over several years and is not a quick remedial strategy for one season. See [www.beckerunderwood.com](http://www.beckerunderwood.com) or phone 1800 558 399 to speak to the Australian agents for this perishable product.

**Redheaded pasture cockchafer life cycle**

![Redheaded pasture cockchafer life cycle](image)
Yellowheaded pasture cockchafer is a descriptive group name for about 20 different species of scarab beetles that can occur in improved pastures in Tasmania. Only the four species listed above reach pest status and then only occasionally. They are root feeders like the redheaded pasture cockchafer.

**Description**

Yellowheaded pasture cockchafers are the root-feeding grubs of several beetles belonging to the family Scarabaeidae. This family also includes the redheaded and blackheaded pasture cockchafers.

The adult stage of all of them is a black or brown beetle. The grubs are soft, white, C-shaped, 5–30 mm long with a hard, yellow head capsule (Fig. 51). The pupae are whitish or pale brown and 5–15 mm in length depending on the species.

**Distribution**

These are native species. The shiny pasture scarab is widespread and occurs on most soil types. The small pasture scarab (Fig. 52) occurs on light-textured soils in the east and south-east, while the dusky pasture scarab (Fig. 53) is most commonly found in red soils on the north-west coast. The hairy scarab (Fig. 54) prefers light-textured soils in coastal areas, but is occasionally reported from the Midlands. All except the small pasture scarab occur on the Australian mainland. It is not uncommon for two or more species to occur together in the same paddock.

**Damage—pasture**

The grubs feed on roots and other organic matter in the soil and may cause symptoms in the pasture similar to those of the redheaded cockchafer. However, damage is usually not as extensive and is mostly confined to areas of less than half a hectare. Concentrated bird activity in the infested paddock in late autumn is often the first clue to an infestation.
Life cycle—pasture
Most yellowheaded pasture cockchafers, including the shiny pasture scarab and the small pasture scarab, have a life cycle of one year. Generally, adults fly at dusk in summer and commonly gather on trees, especially eucalypts, to feed and mate. The females return to the soil to lay eggs which hatch in late summer. Grubs feed from autumn through winter to early spring passing through three stages. When fully fed they dig deeper into the soil to pupate. Adults then emerge from the pupae in early to mid-summer.

A few species have a two-year life cycle. These sometimes spend six months or more underground in the adult stage between emerging from the pupa and joining in dispersal flights later in the year. The larval period of these species is also relatively long (perhaps 10 months).

Control—pasture
There are no effective chemicals available at present which give economical control. As for the redheaded pasture cockchafer, management practices should be adopted which minimise damage as much as possible. The comments made for redheaded pasture cockchafers in this regard generally apply to yellowheaded pasture cockchafers. Chafer Guard™ granules are not registered as effective against yellowheaded pasture cockchafers.
**Argentine stem weevil**  
*or ryegrass stem weevil,  
*Listronotus bonariensis*

**Description**  
The adult is a greyish brown beetle 3.0–3.5 mm in length (Fig. 55). The head is elongated into a snout—a characteristic of all weevils. Three pale longitudinal stripes are visible on the thorax (Fig. 56). The body is sparsely covered with small, stiff bristles. Eggs are cylindrical, less than 1 mm in length and when first laid are yellow but quickly darken to black. Hatchling grubs are about 1 mm in length, increasing in size to about 5 mm at maturity. They are legless and cream with a brown head capsule. The pupa is white and less than 3 mm long.

**Distribution**  
This species is not native to Tasmania. It first appeared in the 1970s. It now occurs widely in the agricultural districts of the state though no records exist for the major Bass Strait islands.

**Damage—pasture**  
In New Zealand the ryegrass and cocksfoot component of pastures is particularly susceptible to attack by this insect. However, it does not appear to be a serious problem in Tasmania. In New Zealand severe damage is caused by grubs mining in the grass stems (Fig. 57). This affects vegetative tillers that wilt and yellow from the centre outwards. Clover dominance can follow. Adult feeding, though normally not economically significant, produces narrow rectangular holes near leaf tips, giving a windowed appearance, which may be confused with slug damage (Fig. 58). Adults leave a fibrous frass on leaves. An endophytic fungus infesting local perennial ryegrass may confer resistance to the weevil.

Annual and short lived ryegrasses do not have endophyte–based resistance to Argentine stem weevil.

**Life cycle—pasture**  
In New Zealand there are 1–2 generations annually, varying with locality. Life cycle details for Tasmania have not been established. In New Zealand first generation grubs are present from October to mid–December and second generation grubs from January to April.
Eggs are inserted under the top layer of the sheaths and require one month to incubate in spring, two weeks in summer. Grubs tunnel in tillers for two months in winter–spring. The pupa forms in the soil. The adult weevils disperse by flight.

**Control—pasture**

In Tasmanian pastures and annual grasses, treatment of Argentine stem weevil infestations is usually not necessary. No insecticides are currently registered for its control in pasture but some are registered for control in turf. Nitrogen fertilisers may favour the pest.

**Damage—forage**

Brassica forage is not at risk. This pest prefers plants in the grass family.

Poor establishment, yellow streaking and wilting of maize seedlings is caused by the grub tunnelling in the stems. Minimum tillage when reseeding pasture increase the risk. In paddocks near Devonport it clearly prefers lighter grey soils to red ferrosols, although individuals can be found on both soil types.

**Life cycle—forage**

New Zealand experience suggests that the mode of infestation in maize is direct transfer of grubs from decomposing turfs to emerging maize seedlings. However, eggs have been found in Tasmanian sweet corn seedlings in December suggesting that, at least some, infestation is caused in spring by adult weevils.

**Control—forage**

No pesticides are registered for control in maize. A fallow period of four weeks between cultivation of pasture and planting maize will reduce infestation. Minimum tillage will increase the likelihood of transfer of infestations from pasture to maize.

---

Fig. 57 Grub of Argentine stem weevil with dark head emerging from tunnel in stem of maize seedling

Fig. 58 Holes in grass chewed by Argentine stem weevil beetles
Sitona weevil

*Sitona discoideus*

**Description**
The adult is a grey beetle with a short snout. The body is around 2.5–3.0 mm long. Eggs are 0.4 mm long and 0.3 mm wide. Grubs grow to 3 mm long. They are fat, white, legless grubs with a slight curve in the body and a yellow head capsule. The pupa is white and occurs in the soil (Fig. 59).

**Distribution**
This European species entered the state before 1972. It previously entered the Australian mainland in 1958. It occurs in the Midlands, Fingal Valley, northern and south-eastern districts of the state and on Flinders Island. It is a capable flier and can be widely dispersed by wind. Hence individuals can turn up almost anywhere. The related clover root weevil, *Sitona lepidus*, is a pest in New Zealand but is not known from Tasmania.

**Damage—forage**
Lucerne is particularly susceptible to attack but other legumes such as burr medic, spotted medic, white clover, red clover, subterranean clover and vetches are also attacked. Adult sitona weevils chew foliage causing a scalloped leaf-edge or complete defoliation (Fig. 60). Seedlings are susceptible. The grubs of sitona weevil tunnel into or chew upon the root nodules that supply nitrogen to legumes.

**Life cycle—forage**
The Tasmanian life cycle has not been studied but a one year cycle is likely. This pest appears to adapt its life cycle strategy to local climate. The following outline is tentative. Adults may feed sporadically through winter. In early spring adults commence feeding on lucerne and medic foliage and mature to reproductive stage. Each female lays up to 1300 eggs through spring. The eggs are laid on the soil surface and hatch in two weeks. The grubs at first tunnel into root nodules but older grubs feed externally on root nodules. Pupae form in soil chambers. Adults can also be found feeding on foliage in autumn. Adults are active fliers and can disperse many kilometres in autumn.

**Control—forage**
No control strategy has been developed for Tasmania.
**Whitefringed weevil**
*Naupactus leucoloma*

**Description**
The adult resembles a ‘sunflower seed with legs’ both in size and colour (Fig. 61). The body is 12 mm long. Eggs are laid in the soil. Grubs live in the soil and grow from 1 mm to 12 mm long. They are fat, white, legless grubs with a slight curve. Unlike cockchafer grubs the whitefringed weevil grubs are not dark internally at the hind end. The head capsule is pale and indistinct whereas in other weevils it is prominent and coloured (yellow, orange or brown) (Fig. 62). However, the dark chewing mouthparts are conspicuous in the head of whitefringed weevil grubs. The soft and white pupa rests in the soil.

**Distribution**
This South American species apparently entered the state in the mid 1980s although it was not officially recognised until 1992. It entered the Australian mainland around 1932. It occurs along the northwest coast, in the Tamar Valley and near Pipers Brook. There are spreading infestations developing around Scottsdale, Swansea and Ouse. Other spot infestations are likely to develop as a result of the adult weevils being carried in agricultural produce and machinery or the grubs in pot plants or the root balls of tree saplings.

**Damage— forage and pasture**
Lucerne is particularly susceptible to attack. The life of lucerne crops can be reduced to a couple of years by damage from this pest. The grubs chew furrows and pits along the roots (Fig. 63). Weevils bred on lucerne can lay 1600 eggs versus 200 for those bred on grasses and weeds. The grubs also damage the roots and tubers of vegetable crops and poppies. In pasture, the grubs attack clover and tap-rooted flat weeds. The grubs are least favoured by fibrous-rooted grasses.

Adults chew the foliage of many plants but this damage is usually not serious.

**Life cycle—forage**
The Tasmanian life cycle has not been studied but a two year cycle is likely. Grubs of all ages can be found in winter suggesting that there is not one synchronised
generation annually. However, adults are most common in late summer and early autumn and disperse by walking. All adults are females that cannot fly. They do not need to mate with males to lay viable eggs, so that a single weevil carried in forage or machinery can start a new infestation.

**Control—forage**

Farm to farm hygiene is important because this pest is not yet widespread. One female weevil can establish a new colony. They disperse as described above. Once established this pest is difficult to manage because the grubs are protected in the soil while pesticides aimed at adults have poor efficacy.

Victorian experience with potato crops suggests that high rates of soil insecticides in furrows at planting give only partial control. Regular soil cultivation is likely to disrupt the long life cycle. Soil fumigation may be needed when populations become too high.

![Grubs of whitefringed weevil chew the roots and stem bases of lucerne, tap-rooted weeds and vegetables](image)
Wingless grasshopper

Phaulacridium vittatum

Description
The adult grasshopper is 10–18 mm long and is usually greyish brown in colour. Some individuals also have a creamy–white stripe along each side (Fig. 64). Colouration within a population may vary from brown–black to pale tan. Most of the grasshoppers will have short non–functional wings but a few will have fully developed wings and are capable of flying short distances.

The young hoppers are 3–4 mm long and pinkish in colour when newly emerged from the egg. They rapidly darken to black. Juvenile hoppers of intermediate age are brownish black in colour and 6–10 mm in length.

Distribution
This pest was native to savannah woodland and natural grassland in southern Australia prior to European settlement. Although widespread within Tasmania, the wingless grasshopper is predominantly a pest on coastal sandy soils in eastern Tasmania and on the Bass Strait islands. It is also common in areas of south–eastern and south–western mainland Australia where annual rainfall exceeds 500 mm.

Wingless grasshoppers are not locusts which are much larger grasshoppers. Locusts do not occur in Tasmania except as rare individuals blown from the mainland.

Damage—pasture
Despite their name, wingless grasshoppers prefer broad–leafed plants to grass. They became a pest after 1950 as summer growing native grasses were displaced by clover, ryegrass and broad–leafed weeds.

Infestations often follow dry winters and springs. Thin, weedy pasture favours the survival of juvenile grasshoppers and is less able to withstand damage. Competition with livestock becomes intense and increases the likelihood of infestations in following years. Overgrazing may increase damage by encouraging broadleaf weed invasion.
In dry seasons a mermithid nematode ‘worm’, which is a major parasite and natural control agent of the wingless grasshopper, is less abundant.

By feeding on clovers in preference to grasses, wingless grasshoppers may cause legumes to almost disappear from a paddock. Grasses may establish poorly because young shoots are eaten by adult grasshoppers in autumn. Paddocks infested over a number of years tend to become weedy since the sown perennial species eventually succumb to insect attack. Outbreaks can recur for a succession of years until rainfall, denser and grassier pasture, and parasitism, hinder population growth.

**Life cycle—pasture (Fig. 65)**

The wingless grasshopper has only one generation each year.

Females mate and lay eggs in late summer and autumn. Eggs are deposited in the soil at a depth of 20 mm in compact bundles called ‘pods’. A female may lay up to 10 pods in her lifetime and each pod contains about a dozen eggs. The eggs remain dormant through the winter. Hatching occurs in November and can be spread over 3–4 weeks due to local variations in soil temperature related to aspect and pasture cover. Areas of concentrated laying are called ‘egg–beds’ and are commonly used year after year.

After hatching, the young hopper passes through five stages over seven weeks before becoming an adult. Young hoppers feed on clovers and flat weeds (not grass), in the vicinity of the egg–bed. Availability of suitable food is crucial at this stage and high survival only occurs if pasture cover is low and suitable food plants are abundant. Flowers of capeweed are a favoured host at this stage. Damage to this weed can be used as an indicator of the grasshoppers presence. Dense grass means that their favoured foods are difficult to find so that large numbers of grasshoppers are rarely found in rank pastures.

Older juveniles disperse more widely, often encouraged to move as annual grasses dry, and permit access to low–growing food plants. When numbers are high, dense streams of 10 mm long hoppers can be seen dispersing.

Adults begin to appear in late December–January and can survive on a wider range of plants. Under favourable conditions of abundant food and high temperature, egg–laying may commence in mid to late summer. Adults are not migratory and rarely move more than 50 metres a day. Under dry conditions populations may concentrate in moist flats, forage crops, gardens or irrigated areas where they can cause severe damage.

**Assessment of grasshopper numbers**

There is usually only a limited number of favoured breeding areas on each farm. Egg–beds are concentrated here. Typical breeding sites might be old sheep camps, sandy rises in undulating paddocks, or boundaries adjacent to native vegetation. Inspection of these sites for the presence of young hoppers 4–8 mm long should start in mid–November. A population in excess of 20 per square metre should be regarded as potentially damaging. Numbers in excess of 50 per square metre would warrant spraying. A dry spring period and high stocking rate (which limit grass cover and encourage flat–weeds), favour high survival of young hoppers.
Fig. 65

Wingless grasshopper life cycle:
- EGGS LAYED
- EGGS OVERWINTER IN SOIL
- Nymphs growing & feeding
- ADULTS FEEDING
- ADULTS DIE

Fig. 66
Five Tasmanian grasshoppers:
The wingless grasshopper is top left.
The yellow-winged grasshopper is bottom left.
The other species are not pests.
**Control—pasture**

Unlike locusts the wingless grasshopper does not invade areas from remote breeding grounds. Instead numbers build up locally and the need for control is at or near the site of the population increase. Revegetation of egg–beds with dense perennial grasses and trees will remove breeding sites.

For chemical control early detection is critical to success. Control can be obtained by well–timed strategic spraying with insecticides. Start spraying when hatching is complete (2–3 weeks after initial hatching is noted), but before the young hoppers disperse from breeding areas in late December (5–6 weeks after hatching). A follow up spray against adults may be needed, before any reverse migration back to breeding areas. It is best to spray in the morning because hoppers shelter in the heat of the day. There are about four weeks in which to locate and assess trouble spots and to spray where necessary.

Boom sprayers can be used where the terrain permits. Aerial spraying with ultra low volume formulations of insecticide may be more appropriate for large areas.

Baits made from bran mixed with the insecticide malathion can also be used to protect valuable areas.

While considerable research into the bio–control of wingless grasshoppers has been done in Australia, no economical solution has yet been found. Monitor the website www.beckerunderwood.com for progress reports on Green Guard®, a fungal insecticide that is under development for locust and grasshopper control.

**Damage—forage**

In dry summers the grasshoppers are more likely to disperse from pasture to forage, vegetables, orchards, pine shelter belts and gardens to defoliate plants.

**Life cycle—forage**

See pasture section. The grasshoppers have only one generation annually. Their eggs must survive winter in an uncultivated, lightly vegetated, sandy soil. Hence grasshoppers are unlikely to originate within forage crops but move into them from elsewhere. Understanding this cycle is key to timely control.

**Control—forage**

Consult the section on ‘control in pasture’ above. Spot spraying the young juveniles before they disperse from ‘egg–beds’ is most economical. In late summer protective spraying of forage crops, orchards or gardens may be necessary. Spraying at six to ten day intervals may be required and barrier spraying of surrounding pastures to a width of at least 10 metres will reduce the rate of re–infestation.
Black field cricket
Teleogryllus commodus

Description
Adult crickets are about 25 mm long and glossy black in colour (Fig. 67). The hind legs are enlarged like those of a grasshopper but the antennae are longer than the body whereas grasshoppers have short antennae.
The female has a rod–like egg–laying tube (ovipositor) projecting from the end of her body. The eggs are yellowish and about 2 mm long. Immature crickets, called nymphs, resemble miniature adults, but lack wings, and have a narrow white band across their body.

Distribution
The black field cricket occurs widely in the pastoral areas of eastern and southern Australia and is most commonly found on heavy–textured soils. This pest is abundant on Flinders Island, in parts of the Midlands and in the south–east of the State.

Damage—pasture
The black field cricket can be especially troublesome in the autumn after a dry summer. All pastures that have an open sward, are over grazed or heavily stocked, are prone to the soil drying and cracking and subsequent infestation. Black cracking clay sites are favoured because the crevices that develop in summer and autumn offer the crickets good protection from birds and dehydration.
Crickets feed outwards from cracks (Fig. 68) and consume most types of pasture plants, including germinating seeds. Ryegrass succumbs rapidly, cocksfoot, tall fescue and phalaris are more tolerant of damage but even these will suffer in heavy infestations.

Assessing damage—pasture
It is useful to walk around infested areas at dusk on warm evenings and observe the extent of cricket activity. Numbers can be easily underestimated because of their habit of sheltering in cracks during the day. A more accurate method of assessing numbers involves placing hessian bags, folded in half, over soil cracks.
Crickets shelter under the bags and by observation their numbers can be estimated by quickly lifting the bags next day. Ten bags per paddock will give a good guide and it pays to make several counts at different times. Research done in Victoria suggests an average of 5–7 crickets per bag (or one or more bags with at least 20 crickets) warrants control measures. Bare areas along the margins of cracks are another sign that enough crickets are present to warrant treatment.

Life cycle—pasture (Fig. 69)
There is only one generation each year. Adult crickets die with the onset of winter. Eggs are laid from late summer to late autumn in the crowns of grasses or singly, but loosely clustered about 10 mm deep in the soil. Each female can lay up to 1000 eggs. The eggs remain dormant through winter. They hatch in spring. The juveniles (nymphs) grow slowly over the summer. They are fully grown and mature by February or March when breeding takes place. Males may be heard chirping at dusk at this time. Crickets are mostly active at night when they feed and lay eggs.

Control—pasture
In areas susceptible to cricket damage cracking of the soil can be minimized by maintaining a ground cover. Areas on the farm attacked regularly could be sown to cocksfoot, tall fescue, phalaris or lucerne all of which tolerate cricket attack better than ryegrass.

Control can be achieved using bait mixed with the insecticide malathion. Instructions for doing so can be found on the label of certain formulations of malathion. Whole wheat, barley or oats can be mixed with insecticide in a cement mixer. The mixture should be held for 24 hours to assist penetration of the insecticide into the grain.

A spinner–type fertiliser spreader can be used to distribute the bait. Baiting is best done in February or early March prior to the serious damage that can occur later in autumn. Crickets that eat poisoned crickets also die.
Black field cricket life cycle

- **Eggs hatch**: Eggs hatch from the soil in mid-June.
- **Nymphs growing & feeding**: Nymphs grow and feed from June to December.
- **Adults feeding**: Adults feed and mate from January to April.
- **Eggs laid**: Eggs are laid in May.
- **Eggs overwinter in soil**: Eggs overwinter in the soil from May to June.
- **Adults die**: Adults die in July.

Fig. 69
Lucerne flea
*Sminthurus viridis*

The lucerne flea is a tiny, wingless, yellow–green, globular insect (Fig. 70). It belongs to a group known as 'springtails' and can leap into the air by means of a forked 'spring' under its abdomen. The eggs are pale yellow and about 0.3 mm in diameter (Fig. 70). The newly hatched juveniles or nymphs are about 1 mm in length and gradually increase in size to about 3 mm in the adult stage.

**Distribution**
Lucerne flea is an introduced species. It is most troublesome as a pest of pastures in the north–west, especially from Table Cape westwards, but also around the Devonport area. It does occasional damage to pastures in the north–east particularly in the Winnaleah area. Lucerne flea does not occur in pastures in the isolated regions of the west coast and is uncommon in midland areas of the State. Although it occurs in isolated pockets in pasture in the east and south–east, it is not regarded as a major pest in these areas, except in unusually favourable seasons.

**Damage—pasture**
The lucerne flea eats the green tissue of leaves (Fig. 71) and despite its common name, does more damage to the clover component of pasture than to lucerne. Young nymphs initially eat small holes in leaves creating a speckled appearance (Fig. 72); older nymphs and adults eat out larger, window–like holes. In an advanced stage of an attack by the insects only the veins and ragged portions of the clover leaves are left (Fig. 75).

A heavy infestation of lucerne flea in pasture can reduce the amount of feed available to stock and reduce the palatability of the remaining feed by fouling. Newly sown pastures that are attacked may not establish properly.

**Life cycle—pasture (Fig. 73)**
The lucerne flea is best suited to moist conditions and temperatures around 11–16 °C. Dry or excessively wet conditions cause a rapid drop in numbers.

Eggs are laid in batches of about 50–60 in moist situations usually on the soil surface, or beneath decaying
leaves and debris. After they are laid the eggs are coated with a fluid incorporating excreted soil that dries on exposure to the air.

The eggs endure a period of arrested development under the dry conditions experienced during summer. Hatching, triggered by cool, moist weather after autumn rains, may take up to a fortnight or longer at lower temperatures. The nymphs undergo about five molts before becoming sexually mature and able to lay eggs. The length of the nymphal stage varies from 4–6 weeks after the opening autumn rains, and during spring, but may last 8–9 weeks during the cooler winter months.

Severe outbreaks are likely when there are good rains late in the summer. Such unusually early rains lengthen the period over which the flea is active and increase the possibility of severe damage in the autumn.

Populations start to increase in autumn and usually reach a high level by early winter. Populations decline in winter because of the less favourable weather conditions but some activity continues. Flea populations normally reach their highest levels during the warm, moist conditions in spring (Fig. 73).

In non–irrigated pastures flea numbers decrease with the approach of hot dry weather in summer. However their eggs survive and hatch after the onset of late summer or autumn rains.

Hatching occurs during summer if pastures are irrigated, and this may lead to damage. Poorly drained, damp areas of non–irrigated pasture may also carry small populations during summer.

Under normal conditions when hatching starts in autumn the lucerne flea can complete up to four generations a year in Tasmania, although the number may vary according to prevailing conditions.

Fig. 73 Pattern of lucerne flea activity in Tasmania based on monthly averages from four sites in north–west Tasmania from May 1976–April 1980
Control—pasture

Grazing management:
Close grazing opens the sward to expose the lucerne flea to the detrimental effects of heat and dryness. Because pastures stocked with sheep are more closely grazed than cattle paddocks they are usually less susceptible to flea damage.

Predators:
Research into biological control of lucerne flea conducted in Tasmania in the mid to late 1970s showed that several species of introduced predatory mites of European origin attacked the lucerne flea, but were unable to prevent populations reaching outbreak levels. One of these predators, the pasture snout mite, *Bdellodes lapidaria*, is not suited to the climate of the far north–west, where the flea problem is worst, and it has little effect on flea numbers.

Between 1985 and 1990, a new program was initiated that resulted in the successful introduction and establishment of an additional European predator, the spiny snout mite, *Neomolgus capillatus* (Fig. 74). As the predator has a slow natural rate of spread, a redistribution program between 1992 and 1998 resulted in the transfer of over 1.6 million of the predators from established sites to 840 new sites on over 300 of the state’s dairy properties. The predator is continuing to slowly spread from these established sites.

Efficacy data collected from Tasmanian dairy pastures showed that the addition of the spiny snout mite resulted in effective control of autumn populations of the lucerne flea, with mean density reductions of around 93%. However, the control of spring populations was variable and less effective. Although mean density reductions of around 84% were recorded in spring, the spiny snout mite and other mite predators usually became active too late in the spring to prevent lucerne flea reaching damaging levels. Even so, the ability of the spiny snout mite to effectively control autumn populations of lucerne flea is enough to warrant an additional program to further accelerate the dispersal of this species, particularly around the properties where it is already established.
The spiny snout mite has a similar life cycle to lucerne flea, with populations showing peaks in both autumn and late spring.

**Chemical sprays:**
At present, insecticides are still an effective way of controlling lucerne flea. For optimum control the spraying must be carefully timed. The four generations of the flea each year overlap so that a complete range of flea stages from unhatched eggs, newly hatched nymphs, through to egg-laying adults may exist at any one time in a given population. Eggs are not killed by insecticides.

**When to spray:**
The best method of achieving optimum control is to spray in autumn shortly after the hatching of the over-summering eggs when most of the individuals in the population will be immature and not laying eggs. However, spraying is often necessary during spring, once populations start to increase again after the cooler winter weather. Correct timing of spring sprays is more difficult. The age of the fleas in the population in spring is less uniform, and some eggs will be present because flea activity will have been continuous since autumn.

There are several main points to consider: the eggs of the flea are resistant to insecticides; the eggs usually hatch after a good autumn break; the hatching takes about two weeks; and it takes about another 4–6 weeks before the fleas mature and start laying more eggs. The aim is to spray after all eggs have hatched but before the fleas are mature enough to lay more eggs. If spraying is delayed for too long, a follow-up spray may be necessary to control a second generation.

The critical time to spray in autumn is 2–5 weeks after the opening rains (Fig. 73). The critical spraying time should be assessed by inspecting the infested pasture at weekly intervals after the autumn break. Although populations usually fall to less damaging levels in winter, close watch should be kept for the resumption of higher levels of activity in spring—these usually occur with the onset of milder weather conditions. Because of the vigorous pasture growth that occurs during spring, flea damage is often overlooked. However, it should be remembered that the spring flea populations lay the over-summering eggs that give rise to populations in the next autumn.

**Life cycle—forage**
Lucerne flea is primarily a pasture pest associated with clover and other legumes. It can persist after minimal cultivation to damage the seedlings of subsequent forage and cash crops as well as seedlings of many pasture grasses and legumes.

**Damage—forage**
Lucerne flea also attacks forage plants and lucerne. Damaged foliage will show many pinhead-sized, window-like holes that eventually coalesce into larger, ragged holes. Established plants will usually compensate for a little foliar damage, but young stands may be completely defoliated, resulting in reduced survival.
Control—forage
Lucerne flea can persist after pasture is minimally cultivated. Prior to planting spring brassica forage, apply an insecticide for lucerne flea control before applying herbicides to kill the existing pasture. Alternatively, sow insecticide–treated seed. Control weeds in fallow and headlands for one month prior to sowing. See pasture sections above for details of life cycle and spray strategy in pasture. Monitor emerging forage plants for the first fortnight after sowing in spring. Spray if emerging plants display leaf damage.
Earth mites
Redlegged earth mite, *Halotydeus destructor*
Blue oat mite or pea mite, *Penthaleus major*
Blue oat mite, *Penthaleus falcatus*

Description
These mites are similar in appearance. When fully grown they are about the size of a pin head and have a dark blue or black body with red legs. Blue oat mites (Fig. 76) are recognised by the red spot near the hind end of the body. Their legs are redder than the salmon–pink legs of the redlegged earth mite (Fig. 77).

Distribution
These pests are not native species. The redlegged earth mite (Figs 78 and 79) was first recorded on the Tasmanian mainland at Devonport in 1940, having previously been known from King and Flinders Islands. It appeared in Australia in 1917 from South Africa. It now occurs in the north, south and east of the state but is more restricted in its occurrence than the widespread blue oat mite, *Penthaleus major*. The largest populations of redlegged earth mites are found on lighter, well drained soil types particularly in coastal areas. There are two species commonly called blue oat mite. The blue oat mites occur in most pastures throughout agricultural areas of the State. *Penthaleus falcatus* is much less abundant than *P. major* but is notable for being highly resistant to some insecticides.

Damage—pasture
Redlegged earth mite:
This is a major problem for many growers, and may cost the industry several million dollars annually. The redlegged earth mite commonly damages clover and other legumes in pasture. The mites feed by rasping the surface layers of plant tissues and sucking up the released juices. Damaged foliage takes on a silvery, bleached appearance as with subterranean clover (Fig. 80) or leaves can become twisted and distorted as with white, balsana, persian and arrow leaf clovers. Legumes may be damaged at any age, but seedlings and young plants of subterranean clover are the most susceptible.
Blue oat mites (two species):
Damage appears similar to that described above for redlegged earth mite. Large populations of the blue oat mites often occur in pastures but do not cause significant injury to legumes in Tasmania. In pasture *P. major* feeds mainly on grass leaves, while *P. falcatus* mainly feeds on broad-leaved weeds such as cat’s ear and bristly ox-tongue. Blue oat mites often co-exist, and perhaps compete, with redlegged earth mite. The juveniles may feed on soil microflora and this requirement may influence their distribution and abundance.

**Life cycle—pasture**
Redlegged earth mites occur as both sexes and reproduce sexually. All adults of blue oat mites are females and produce populations of genetically distinct clones. Over 20 clones of *P. major* have been detected in Australia.

The period of redlegged earth mite activity starts with the opening autumn rains and continues through winter and spring until the approach of the hot and dry summer months. Over autumn and winter, females lay their non diapause eggs on soil or host plants. Irrigation may prolong the period of activity. The summer months are spent as eggs retained within the body of the dead female on or near the soil surface. These summer, or diapause, eggs hatch with the opening rains and cooler temperatures of autumn. There are two or three generations of the mites each year. As with lucerne flea the peak periods of activity occur in spring and autumn/early winter with numbers being reduced by cooler winter temperatures. Blue oat mites have a broadly similar life cycle.

**Control—pasture**
**Identification:**
It is very important to determine which species is present or whether there is a mixed population. The risk of damage and the timing of control differ for the three species. *P. falcatus* is the most problematic species to control because it is strongly resistant to several pesticides.
Chemical sprays:
Insecticide applications take three forms—bare earth treatments, seed dressings and foliar treatments. Residual bare earth treatments probably interfere with natural enemies of several other pasture pests. Seed dressings are more selective but may fail under high pest pressure. Single foliar treatments require precise timing.

The rarer blue oat mite, *P. falcatus*, is naturally resistant to all earth mite insecticides registered as of 2001. This includes a 50-fold tolerance to omethoate compared to redlegged earth mite. The more widespread blue oat mite, *P. major*, is rarely involved in insecticide control failures, although it possesses some tolerance to some synthetic pyrethroid and organo–phosphate insecticides. Redlegged earth mite is the least tolerant species, except in the case of phosmet, to which *P. major* is the least tolerant species.

The TIMERITE® information package (www.timerite.co.au) developed by CSIRO provides the date for a single mid spring spray that controls redlegged earth mite through to the following autumn. The date is when the largest proportion of adults is present and the lowest proportion of summer eggs. The calculation of the date is determined more by the latitude than temperature or rainfall. Spraying on this date reduced populations of redlegged earth mite by 88% in many trials in eastern Australia between 1997 and 2003. A supplementary spray may be needed well before mid spring if populations are large enough to cause loss of clover. Personal observation and monitoring of pasture will reveal if early populations are high.

This strategy does not apply to blue oat mites.

Chemical control of redlegged earth mite can be attempted in autumn when the majority of eggs have hatched and the mites are not yet mature enough to lay eggs. Unlike the spring date, the autumn date varies with weather from year to year. This period may occur 2–5 weeks after a good autumn break. However, unhatched eggs, which are resistant to sprays, may hatch several days or a few weeks after spray application. Because of this a second spray application is often necessary. Farmers should also keep a close watch for the resurgence of populations in spring.

Cultural methods:
Minimum tillage often favours earth mites, but crops of wheat, oats, chick peas, lentils or narrow–leafed lupins reduce the summer egg–load, provided that weeds are not present as alternate food for the mites. Heavy stocking in early to mid spring reduces mite populations by defoliating mite food plants and by trampling. It also helps new clover to germinate.

Damage—forage
Besides clover in pasture, the redlegged earth mite also damages grass, seedling lucerne, peas, oats, canola, forage and vegetables. Damaged leaves become yellow, then white. Historical records suggest that peas and oats were most often attacked by blue oat mites in Tasmania—possibly both *P. major* and *P. falcatus* but only the former was recognised when the records were compiled. On the mainland, at least, *P. falcatus* is the species of ‘blue oat mite’ most often found infesting canola. It is not known whether this applies in Tasmania but it is significant because this species has resistance to many pesticides.
**Life cycle—forage**
See pasture section. Note that earth mites are usually a problem in emerging forage crops as the result of carry-over from preceding pasture.

**Control—forage**
Prior to planting spring brassica forage, include an insecticide for earth mites with the herbicide when killing existing vegetation. Mites may also invade from neighbouring paddocks. Remove weeds from fallow at least one month before sowing. See pasture sections above for details of life cycle and spray strategy in pasture. Monitor seedlings for two weeks after sowing forage brassicas in spring and spray if foliar bronzing develops rapidly. Seedlings of some high value crops, such as canola, have a low damage threshold. A seed dressing will protect seedlings if previous experience indicates a high risk.

![Clover leaves silvered and bronzed by redlegged earth mite sucking](image)

*Fig 80*  Clover leaves silvered and bronzed by redlegged earth mite sucking
Several aphids infest forage crops such as brassicas, lucerne and cereals. They include:

**Grey cabbage aphid, *Brevicoryne brassicae*** in brassicas  
**Turnip aphid, *Lipaphis erysimi*** in brassicas  
**Green peach aphid, *Myzus persicae*** in brassicas  
**Spotted alfalfa aphid, *Therioaphis trifolii*** in lucerne  
**Blue green aphid, *Acyrthosiphon kondoi*** in lucerne  
**Pea aphid, *Acyrthosiphon pisum*** in lucerne.

All are introduced species but they usually do not need control. Small populations will usually succumb to natural controls before causing economic damage. These natural controls include fungal diseases, parasitic wasps and predators such as brown lacewing, hoverflies and ladybirds. However, the use of broad spectrum insecticides, such as many cheap synthetic pyrethroids and organophosphates used against other pests, will destroy the predators and parasitoids of aphids. This will lead to high densities of aphids, which may then need costly control by insecticides. It is often cheaper and simpler to use selective insecticides for the other pests, such as caterpillars, in the first place.
Small pointed snail

*Cochlicella barbara*

**Description**
This small snail, also known as conical snail, has a characteristic tapering conical shell drawn to a blunt point (Fig. 83). The shell is pale in colour with a brown spiral band or brown streaks. Newly hatched snails are almost transparent and about 2 mm in length. Adult snails have a shell between 8 mm and 12 mm in height and about 5 mm in maximum diameter. A similar but larger species, the pointed snail, *Cochlicella acuta*, does not occur in Tasmania but is a pest on the mainland.

**Distribution**
This pest was introduced to Australia from southern Europe. Although widely distributed in pastoral areas, small pointed snails reach pest status only on King and Flinders Islands and in the north–west, particularly the Circular Head district, where they seem to be favoured by relatively high annual rainfall (more than 1000 mm per year). Populations are generally higher in rank pastures than in short pastures. In marginal areas snails may be largely confined to ditches or low–lying poorly drained areas.

**Damage—pasture and lucerne**
Heavy infestations may occur in autumn and spring. These snails feed on a wide variety of broad–leafed plants including legumes and many weeds. After a severe attack, plants take on a tattered appearance and may be liberally coated with dried shiny mucus. Nevertheless, such damage is rarely on a large scale or of lasting significance. This pest causes severe damage to emerging pastures on King Island. The major hazard to pasture presented by this pest is fouling. Stock, especially cattle, are deterred from feeding when snails are abundant.

**Life cycle—pasture**
The life cycle of this pest in Tasmania has not been studied.

**Control – pasture and lucerne**
Unless populations are high or extensive, control is generally not necessary. Block grazing stock can decrease populations by trampling. Soil cultivation, burning and rolling will also reduce snail populations prior to planting lucerne.
Slugs

Black-keeled slug, *Milax gagates*

Brown field slug, *Deroceras panormitanum*

Grey field slug or reticulated slug, *Deroceras reticulatum*

Hedgehog slug, *Arion intermedius*

Description

The adult of the grey field slug is fawn with grey meshlike markings, 50 mm long and exudes sticky, white milky body mucus if poked (Fig. 84).

Its close relative, the brown field slug is uniformly brown to grey, 35 mm long and exudes clear, non-sticky mucus (Fig. 85).

Black-keeled slug is uniformly black, 50 mm long and has a crease or keel along the back.

Eggs are translucent, soft spheres about 1 mm in diameter and found in clusters in the soil (Fig. 86). Juveniles resemble miniature adults.

The hedgehog slug, *Arion intermedius*, is pale yellow in colour, 15–20 mm long and its skin is rough in texture. It may also cause some damage but usually eats fungus.

Distribution

These are not native species but are sometimes found in native habitats. The pest slugs probably occur in most agricultural districts.

Damage—*forage*

Slugs attack a broad range of plants and are most damaging when they feed on germinating seeds and the seedlings of grass, forage and cereals. The black-keeled slug appears most voracious in captivity and probably feeds deeper in the soil than *Deroceras* species but the latter appear to be more abundant and are probably responsible for many losses in germinating crops.

Life cycle—*forage*

Breeding occurs in spring and autumn or when favoured by moisture and sufficient heat. Individuals take 6–9 months to develop. New cohorts of juveniles appear in spring and autumn but irrigation favours more continual breeding.
Control—forage

Slug populations proliferate in moist pasture, particularly where a range of broadleafed seedlings and shelter occur. Close grazing will hinder slug prosperity. Intensive tillage before sowing crops may be needed to reduce populations. Inspect paddocks in advance. Slugs can survive long periods without food if moisture is available so that fallow periods alone may not reduce populations. They are sensitive to desiccation.

Three types of baits are available, (based on the solid fuel compound metaldehyde, the carbamate insecticide methiocarb or the newer type based on chelated iron) but are expensive when used over large areas. Placement and density of baits is important. Some germinating seeds may be eaten before the cotyledons emerge and may be more attractive, and accessible, to slugs in the soil than mouldy, deteriorated baits lying exposed on the soil surface.

Various predatory beetles such as the shiny black ground beetles, Carabidae, prey on slugs but these are harmed by broad spectrum insecticides which may used for other pests.

Biological sprays based on nematodes have been used overseas, for turf management, but are too expensive for broad acre crops.

Fig. 87 Grey field slugs chewing holes in a swede tuber

78
Biological diversity & pasture pest control

plus

glossaries
In natural environments many different species of plants and animals, including insects, co–exist in relative stability. This is the result of thousands of years of co–evolution whereby a complex series of interactions prevents any one species becoming too abundant.

An improved pasture or crop, however, is an artificially simplified habitat with limited biological or physical diversity. Biologists believe that such a situation leads to a high likelihood of pest outbreaks since only a few herbivorous insects are favoured by the new pasture environment which meets all their needs for food and shelter.

Most of Tasmania’s major pasture pests (in particular corbie and cockchafers) are native insects which lived in the native grasslands under open woodland. This vegetation community occupied much of what is now the pastoral zone. In their natural habitat these insects were kept in check by a range of predators and parasites which shared their environment. Birds, lizards, beetles, spiders, ants, parasitic wasps and flies all helped to regulate grass–feeding insects.

Unfortunately some crucial resources needed for the survival of these ‘policeman’ of the natural world are often lacking in improved pastures. For example, parasitic wasps and flies require nectar–bearing flowers for energy to search for prey, predatory beetles need logs or bark for concealment, and birds need shrubs and trees for shelter and nesting sites.

An added problem is that beneficial species are often more sensitive to insecticides than the target pest. Using a cheap, broad spectrum insecticide to control one pest may destroy the natural enemies of another pest and create a new problem. It is well worth integrating all the benefits and costs when selecting an insecticide. A more selective spray may be more effective in the long run even if it is more expensive initially, or more difficult to apply. Selective insecticides can help to maintain biodiversity.

For these reasons environmental diversity is encouraged.
in the pastoral landscape. This can be achieved by further planting of trees and shrubs as well as preservation, free from grazing, of remaining patches of native vegetation. Trees with suitable nectar–bearing flowers for insects include *Eucalyptus, Leptospermum, Melaleuca* and *Bursaria*. A variety of trees and shrubs with different flowering seasons will maximise the availability of nectar for beneficial species. The reversal of tree decline on farms is a necessity and rural tree–planting programs must be further encouraged.

Although broad acre clear–felling of trees to open up new land for pasture has been the practice in the past, a more considered attitude to trees on pastoral properties is needed in the future. In addition to their value as a habitat for predators and parasites of insect pests, trees are useful in erosion control and help moderate climatic extremes. Properly situated wind breaks reduce soil moisture loss and shelter livestock.

Some natural enemies of the pests of grassland or broad acre crops rely more on grassland habitats than woodland and hedgerows. Simple raised, drained, permanent grassy banks conserve their numbers in areas prone to waterlogging or cultivation. These are sometimes called 'beetle banks'.

Simply leaving parts of paddocks unsprayed can also help beneficial insects to re–establish after broad spectrum insecticides are used.

Some pasture pests such as lucerne flea and redlegged earth mites have been introduced from overseas without their natural predators and parasites. Free of this regulation, they can achieve high population levels that can lead to damage. For these pests it is necessary to discover their specialised predators in their country of origin and import them in a carefully planned biological control program.

Other useful insects may need to be introduced as more becomes known about what is lacking in an improved pasture ecosystem. For example no native insects effectively recycle the nutrients in cattle dung in all seasons. However, certain African and European beetles are highly specialised, and effective, at burying this material, thereby returning the nutrients to the soil in a few days.
It is important to add key predators, key parasites or key habitat for those beneficial insects that provide the desired outcomes. This requires biological knowledge of the pasture or forage system.

Further research will assist in bringing our agricultural ecosystems towards a sustainable natural balance, thereby lessening the effect of pests and reducing our dependence on insecticides. The inevitable development of insecticide resistant pests makes it imperative that this work is successful.

**Biological control in Tasmania:**

Spiny snout mite was introduced in the late 1980s and now helps to control lucerne flea as described in that section.

The introduction, and wide dispersal, of five species of dung beetles is another success for biological control.

Several species of tiny European wasps were introduced in the late 1940s and now restrain populations of diamond back moth and cabbage white butterfly in forage brassicas (Fig. 96).

Certain ladybirds, like the 11-spotted ladybird and white collared ladybird, were accidently introduced into Tasmania but now reduce populations of many introduced aphid species in pasture and forage.

Many predators and parasites already present in pastoral ecosystems suppress many pests.
Fig. 98 This microscopic parasitic wasp is about to inject its eggs into a cabbage white butterfly prepupa.

Fig. 99 The fuzzy cadaver of a diamondback moth caterpillar killed by a natural fungus.

Fig. 100 The white collared ladybird is an aphid predator that has recently arrived in Tasmania.

Fig. 101 Predators like this 11-spotted ladybird suffer parasitism by a particular tiny wasp whose cocoon forms under the dead beetle.

Fig. 102 The fungus Cordyceps grows out of a cockchafer grub that it has killed.
Dung Beetles

Dung beetles help provide:

- A cleaner pasture with less unpalatable rank growth
- Faster nutrient cycling
- Soil aeration
- Soil moisture retention
- Reduction of fly breeding habitat
- Reduction of round worm habitat
- Reduction of contamination of waterways.

Several native dung beetles occur on farmland, but few can handle cattle, sheep and horse dung. Five species of exotic dung beetles have been introduced to Tasmania to dispose of dung more effectively.

These beetles carry dung down into tunnels and lay eggs in it. The eggs hatch into grubs that eat the buried dung and grow before transforming, via a non feeding pupal stage, into adults (beetles). The adults emerge from the soil to disperse and reproduce.

The best known is the large, 20–25 mm long, iridescent blue–black *Geotrupes spiniger*. Adults of this species can be collected in autumn when they fly for half an hour at dusk and dawn. Place wire netting on a plastic sheet, then place fresh dung on the wire just before flying time. At the end of flying time, lift the wire with the dung off the sheet. The beetles will be underneath. Release the beetles downwind of fresh cow pats in the cool of the evening. Do not plough this area for one month at least. About 300 beetles should be released to maximise success.

Three other species are half the size of *Geotrupes spiniger*. They are *Onthophagus binodis*, *Onthophagus taurus* and *Euoniticellus fulvus*. These beetles fly in summer rather than autumn. Collection of these smaller species is best done in January and February. First slice a thin layer of turf and invert it on the soil. Place fresh dung on it daily for a couple of days at mid morning because these beetles fly at midday. Do this where stock have been absent for ten days. After several days place the turf and dung onto wire netting on top of a plastic sheet (dung side up). The beetles will tunnel down under the netting. Later lift the wire plus soil and dung to reveal the beetles. About 1,000 may be required to establish a new population.

A fifth species, *Bubus bison*, was released in recent years near Waterhouse.

The veterinary treatment, ivermectin, can kill the grubs and beetles in pats within six days of stock drenching. Keep drenched animals away from release sites for six days.
Earthworms

Earthworms improve soil fertility and their tunnels aid water, air and root penetration. They also accelerate nutrient recycling. Native earthworms fare poorly in managed pasture. However, several introduced earthworms of European origin can thrive on farmland.

They include the small field worm, *Aporrectodea caliginosa*, and purple worm, *Aporrectodea trapezoides*, in the top 100 mm of agricultural and garden soils; large field worm, *Aporrectodea longa*, in high rainfall areas and dung worm, *Lumbricus rubellus*, where organic matter is high. Tiger worm, *Eisenia fetida*, thrives in compost and manure heaps but not in pasture or cropping soils.

Species of earthworms suitable for pastures are not readily available from commercial earthworm farms. Move turfs, 50–75 mm deep, containing pasture species from source areas to establishment areas in late autumn to early spring. Invert the turf at its destination to provide a food source for the worms. Place 100 turfs per hectare.

Conditions that favour earthworms are: plant material with high nitrogen; good soil moisture; minimum tillage; soil pH about 5.5 and minimum soil compaction. Some insecticides and fungicides seriously harm earthworms but fenitrothion and chlorpyrifos, often used against corbies and cockchafers, and most herbicides and ivermectin drenches cause only minimal disruption.
Glossary

Abdomen
The posterior part of the insect body and comprising several segments. The other two divisions of the insect body are the head and thorax. The thorax bears wings and segmented legs. The abdomen of most caterpillars also bears pairs of unsegmented false, fleshy legs.

Biomass
The weight of all the bodies of the organisms in question. Such a measure facilitates comparison of the likely food consumption of 10,000 cockchafers versus, for example, ten sheep.

Conventional seedbed
A firm, fine, bare, even soil surface into which seed is sown using a seed drill. Fertiliser is often placed into the seedbed using the seed drill in addition to the seed.

Cultivation
Total disturbance of the soil to a depth usually not greater than 300 mm using ploughs and other implements to create a conventional seedbed.

Diapause
Arrested development of any stage in the life cycle which synchronizes growth (hatching of eggs, emergence of moths etc.) with suitable environmental conditions.

Direct Drilling
The placement of seed into the soil surface without any disturbance of the soil other than that caused by the seed drill at the time of sowing.

Fallow
A conventionally prepared seedbed, or an area in which all plants have been killed using a herbicide, which is left unsown for a period of time to permit additional weed, pest and disease control before sowing a new pasture or crop.

False Leg (proleg)
Abdominal leg of a caterpillar. These are not segmented, or articulated, and generally appear as pairs of fleshy lobes bearing minute hooks at their tips.

Frass
Pellet–like droppings of caterpillars.

Head Capsule
The sclerotized (a hard 'shell'), compact case that encloses the head of an insect. It is inelastic whereas the 'skin' of the thorax and abdomen is elastic enough to permit some growth before it needs to be shed (or moulted) and replaced. The head capsule size is diagnostic for each stage.

Instar
A stage of larval growth. Insects pass through several larval stages (commonly 3–6) before becoming adults. Moults of the skin in the immature stages (larvae and nymphs) separate each instar and permit expansion of the body. Adult insects remain one size, usually fixed for the species, and do not shed their skin.

Larva
Caterpillar (moth), grub (beetle) or maggot (fly) stage in an insect life cycle occurring between the egg and pupal stages. Larvae are specialised for feeding and may occupy the longest period
of the life cycle. They do not reproduce and generally have restricted dispersal ability. They are very different in form, and often have a different diet, to the adult stage unlike the nymphs of other insect groups (for example, grasshoppers). Nymphs differ from larvae in that they resemble wingless adults and often eat similar food.

**Minimum cultivation (also may be called minimum tillage)**

One or more shallow cultivations to level a rough soil surface, to break up surface organic matter layers and/or to create a shallow seedbed.

**Moulting**

To shed the exterior skin in the process of larval or nymphal growth.

**Nymph**

Young and sexually immature stage that hatches from an egg; smaller but similar to the adult in appearance and often with a similar diet. Because nymphal juveniles are similar to the adult there is no need for a transitional pupal stage, whereas larval juveniles (caterpillars, grubs or maggots) require the non-feeding pupal stage to reconstruct (or metamorphose) their bodies.

**Oversowing**

A term used to describe the sowing of seed of new or the same species into a pasture in order to change or improve the botanical composition of a pasture.

**Parasitoid**

A parasite is an organism that lives at the expense of another, which it does not usually kill. A parasitoid is a parasite that kills its host. Many parasitoids of insects are the larvae of wasps and flies that feed within the host caterpillar or grub. Many are highly specific to one or a few hosts, but some are more generalised. One to many parasitoids can occur within one host but the number that emerge from a host is characteristic of each parasitoid.

**Pasture renovation**

A term used to describe a range of activities from the sowing of a completely new pasture to simply increasing the content of desired species in existing pasture.

**Pathogenic**

Causing or producing disease.

**Prepupa**

An inactive non-feeding stage between the end of the larval stage and the start of the pupal stage.

**Predator**

An organism that eats more than one other species of organism during its life.

**Pupa**

Non-feeding stage between larva and adult with restricted mobility. The pupal stage during which caterpillars such as corbies and armyworms metamorphose into moths often has the appearance of a brown, shiny capsule. Whereas the pupal stage in which beetle grubs such as cockchafers and weevils metamorphose into adult beetles, is often soft and creamy white.

**Spiracle**

An aperture allowing air to enter the system of respiratory tubes within the body of an insect. They often look like a series of tiny ‘portholes’ along the sides of caterpillars and grubs.

**Sheaths**

Green tissue at the bases of the leaflets of grasses that wrap around the stem.
# Names of pest and beneficial species

<table>
<thead>
<tr>
<th>Common name</th>
<th>Genus</th>
<th>Species</th>
<th>Author</th>
</tr>
</thead>
<tbody>
<tr>
<td>Argentine stem weevil</td>
<td>Listronotus</td>
<td>bonariensis</td>
<td>(Kuschel)</td>
</tr>
<tr>
<td>Black field cricket</td>
<td>Teleogryllus</td>
<td>commodus</td>
<td>(Walker)</td>
</tr>
<tr>
<td>Black-keeled slug</td>
<td>Milax</td>
<td>gagates</td>
<td>(Dreparnaud)</td>
</tr>
<tr>
<td>Blackheaded pasture cockchafer</td>
<td>Acrossidius</td>
<td>pseudotaasmaniae</td>
<td>(Given)</td>
</tr>
<tr>
<td>Blackheaded pasture cockchafer</td>
<td>Acrossidius</td>
<td>tasmaniae</td>
<td>(Hope)</td>
</tr>
<tr>
<td>Blue green aphid</td>
<td>Acyrthosiphon</td>
<td>kondoii</td>
<td>Shinji</td>
</tr>
<tr>
<td>Blue oat mite (pea mite)</td>
<td>Pentaleus</td>
<td>falcatu</td>
<td>(Qin and Halliday)</td>
</tr>
<tr>
<td>Blue oat mite</td>
<td>Pentaleus</td>
<td>major</td>
<td>(Dugès)</td>
</tr>
<tr>
<td>Brown cutworm</td>
<td>Agrotis</td>
<td>munda</td>
<td>Walker</td>
</tr>
<tr>
<td>Brown field slug</td>
<td>Deroceras</td>
<td>panormitanum</td>
<td>(Lessona and Pollonera)</td>
</tr>
<tr>
<td>Brown lacewing</td>
<td>Micromus</td>
<td>tasmaniae</td>
<td>(Walker)</td>
</tr>
<tr>
<td>Cabbage white butterfly</td>
<td>Pieris</td>
<td>rapae</td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Cabbage white butterfly parasite</td>
<td>Cotesia</td>
<td>glomerata</td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Cabbage white butterfly parasite</td>
<td>Cotesia</td>
<td>rubecula</td>
<td>(Marshall)</td>
</tr>
<tr>
<td>Cabbage white butterfly pupal parasite</td>
<td>Pteromalus</td>
<td>puparum</td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Chevron cutworm</td>
<td>Diarsia</td>
<td>intermixta</td>
<td>(Guenée)</td>
</tr>
<tr>
<td>Common armyworm</td>
<td>Mythimna</td>
<td>convecta</td>
<td>(Walker)</td>
</tr>
<tr>
<td>Common cutworm</td>
<td>Agrotis</td>
<td>infusa</td>
<td>(Boisduval)</td>
</tr>
<tr>
<td>Corbie</td>
<td>Oncopera</td>
<td>intricata</td>
<td>Tindale</td>
</tr>
<tr>
<td>Damself bug</td>
<td>Nabis</td>
<td>kinbergi</td>
<td>Reuter</td>
</tr>
<tr>
<td>Diamondback moth</td>
<td>Plutella</td>
<td>xylostella</td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Diamondback moth larval parasite</td>
<td>Diadegma</td>
<td>rapi</td>
<td>(Cameron)</td>
</tr>
<tr>
<td>Diamondback moth larval parasite</td>
<td>Diadegma</td>
<td>semiclausum</td>
<td>(Hellén)</td>
</tr>
<tr>
<td>Diamondback moth pupal parasite</td>
<td>Diadromus</td>
<td>collaris</td>
<td>(Gravenhorst)</td>
</tr>
<tr>
<td><strong>Common name</strong></td>
<td><strong>Genus</strong></td>
<td><strong>Species</strong></td>
<td><strong>Author</strong></td>
</tr>
<tr>
<td>-------------------------------</td>
<td>-------------</td>
<td>--------------</td>
<td>--------------------</td>
</tr>
<tr>
<td>Dusky pasture scarab</td>
<td><em>Sericesthis</em></td>
<td><em>nigrolineata</em></td>
<td>(Boisduval)</td>
</tr>
<tr>
<td>Eleven–spotted ladybird</td>
<td><em>Coccinella</em></td>
<td><em>undecimpunctata</em></td>
<td>Linnaeus</td>
</tr>
<tr>
<td>Grass anthelid caterpillar</td>
<td><em>Pterolocera</em></td>
<td><em>amplicornis</em></td>
<td>Walker</td>
</tr>
<tr>
<td>Grey cabbage aphid</td>
<td><em>Brevicoryne</em></td>
<td><em>brassicae</em></td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Green cutworm</td>
<td><em>Neumichtis</em></td>
<td><em>nigerrima</em></td>
<td>(Guénéé)</td>
</tr>
<tr>
<td>Green cutworm</td>
<td><em>Neumichtis</em></td>
<td><em>saliaris</em></td>
<td>(Guénéé)</td>
</tr>
<tr>
<td>Grey garden slug</td>
<td><em>Deroceras</em></td>
<td><em>reticulatum</em></td>
<td>(Müller)</td>
</tr>
<tr>
<td>Green peach aphid</td>
<td><em>Myzus</em></td>
<td><em>persicae</em></td>
<td>(Sulzer)</td>
</tr>
<tr>
<td>Hairy scarab</td>
<td><em>Saulostomus</em></td>
<td><em>villosus</em></td>
<td>Waterhouse</td>
</tr>
<tr>
<td>Hedgehog slug</td>
<td><em>Arion</em></td>
<td><em>intermedius</em></td>
<td>Normand</td>
</tr>
<tr>
<td>Lucerne flea</td>
<td><em>Sminthurus</em></td>
<td><em>viridis</em></td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Meadow argus</td>
<td><em>Junonia</em></td>
<td><em>villida ssp. calybe</em></td>
<td>(Godart)</td>
</tr>
<tr>
<td>Oxyycanus grass grub</td>
<td><em>Oxyxcanus</em></td>
<td><em>fuscomaculatus</em></td>
<td>Walker</td>
</tr>
<tr>
<td>Pasture snout mite</td>
<td><em>Bdelliodes</em></td>
<td><em>lapidaria</em></td>
<td>(Kramer)</td>
</tr>
<tr>
<td>Pea aphid</td>
<td><em>Acyrtosiphon</em></td>
<td><em>pisum</em></td>
<td>(Harris)</td>
</tr>
<tr>
<td>Redheaded pasture cockchafer</td>
<td><em>Adoryphorus</em></td>
<td><em>couloni</em></td>
<td>(Burmeister)</td>
</tr>
<tr>
<td>Redlegged earth mite</td>
<td><em>Halotydeus</em></td>
<td><em>destructor</em></td>
<td>(Tucker)</td>
</tr>
<tr>
<td>Shiny pasture scarab</td>
<td><em>Scitala</em></td>
<td><em>sericans</em></td>
<td>Erichson</td>
</tr>
<tr>
<td>Small pastures scarab</td>
<td><em>Sericesthis</em></td>
<td><em>nigra</em></td>
<td>(Lea)</td>
</tr>
<tr>
<td>Small pointed snail</td>
<td><em>Cochlicella</em></td>
<td><em>barbara</em></td>
<td>(Linnaeus)</td>
</tr>
<tr>
<td>Spiny predatory shield bug</td>
<td><em>Oechalia</em></td>
<td><em>schellenbergii</em></td>
<td>Guérin–Méneville</td>
</tr>
<tr>
<td>Spiny snout mite</td>
<td><em>Neomolgus</em></td>
<td><em>capillatus</em></td>
<td>(Kramer)</td>
</tr>
<tr>
<td>Spotted alfalfa aphid</td>
<td><em>Therioaphis</em></td>
<td><em>trifolii</em></td>
<td>(Monell)</td>
</tr>
<tr>
<td>Sitona weevil</td>
<td><em>Sitona</em></td>
<td><em>discoideus</em></td>
<td>Gyllenhal</td>
</tr>
<tr>
<td>Southern armyworm</td>
<td><em>Persectania</em></td>
<td><em>ewingii</em></td>
<td>(Westwood)</td>
</tr>
<tr>
<td>Transverse ladybird</td>
<td><em>Coccinella</em></td>
<td><em>transversalis</em></td>
<td>Fabricius</td>
</tr>
<tr>
<td>Turnip aphid</td>
<td><em>Lipaphis</em></td>
<td><em>erysini</em></td>
<td>(Kaltenbach)</td>
</tr>
<tr>
<td>White fringed weevil</td>
<td><em>Naupactus</em></td>
<td><em>leucoloma</em></td>
<td>(Boheman)</td>
</tr>
<tr>
<td>Wingless grasshopper</td>
<td><em>Phaulacridium</em></td>
<td><em>vittatum</em></td>
<td>(Sjöstedt)</td>
</tr>
<tr>
<td>Winter corbie</td>
<td><em>Oncopera</em></td>
<td><em>rufobrunnea</em></td>
<td>Tindale</td>
</tr>
</tbody>
</table>