NSW GOVERNMENT | Department of Primary Industries

Canola growth & development

WWW.DPI.NSW.GOV.AU

PROCROP Profitable, sustainable cropping

Disclaimer

The information contained in this publication is based on knowledge and understanding at the time of writing (August 2011). However, because of advances in knowledge, users are reminded of the need to ensure that information upon which they rely is up to date and to check currency of the information with the appropriate officer of Department of Primary Industries or the user's independent adviser.

Recognising that some of the information in this document is provided by third parties, the State of New South Wales, the author and the publisher take no responsibility for the accuracy, currency, reliability and correctness of any information included in the document provided by third parties.

The product trade names in this publication are supplied on the understanding that no preference between equivalent products is intended and that the inclusion of a product name does not imply endorsement by Department of Primary Industries over any equivalent product from another manufacturer.

ALWAYS READ THE LABEL

Users of agricultural or veterinary chemical products must always read the label and any permit, before using the product, and strictly comply with the directions on the label and the conditions of any permit. Users are not absolved from compliance with the directions on the label or the conditions of the permit by reason of any statement made or not made in this publication.

Produced by Department of Primary Industries, August 2011

ISBN 978 1 74256 212 4

Acknowledgments

This book is part of the PROCROP series, a project funded by the National Heritage Trust.

Authors

This book was compiled by Jan Edwards, formerly District Agronomist, DPI, Cowra and Kathi Hertel, District Agronomist, DPI, Dubbo.

Editor

Jan Edwards, formerly DPI, Cowra

Editorial Assistance

Ann Munroe, Scientific and General Editor, Sydney

Layout

Barry Jensen, DPI, Orange

Illustrations

Barry Jensen, DPI, Orange

Template design

Belinda Gersbach, DPI, Orange

Other contributions and assistance

We would like to thank our expert reviewers:

Don McCaffery, Technical Specialist, DPI, Orange

Graeme McIntosh, Project Officer, CropMate, DPI, Dareton

Rachael Whitworth, District Agronomist, DPI, Griffith

Photography

Lowan Turton, DPI, Menangle

Jan Edwards, DPI, Cowra

Julie White, DPI, Tocal

PROCROP
Profitable, sustainable cropping

Contents

Preface

This book describes the growth and development of the canola plant from germination to pod filling. The environmental factors and management actions that influence each growth stage are provided as a practical reference for managing crops.

The aim of *Canola growth and development* is to link plant physiology and crop management. It will help agronomists and farmers to understand the life cycle of the canola plant and the factors that influence crop growth and development. It will also help in identifying the growth stages of the canola plant by using the decimal growth scale. All of this knowledge can then be applied to crop management to maximise yield and profit.

There are four chapters in the book, covering the progression of key stages in the life cycle of the canola plant and its growth and management. Included in each chapter are practical exercises to demonstrate how knowledge of plant physiology can be applied in the paddock.

Introduction

by Jan Edwards

Growing canola

Canola belongs to the botanical family Brassicaceae. Other species in this family are mustard, turnip, wild radish, cauliflower, cabbage and broccoli. They have been cultivated for their edible roots, stems, leaves, buds, flowers and seeds.

Canola is an altered form of rapeseed. The word 'rape' in rapeseed comes from the Latin word 'rapum', meaning turnip. Rapeseed was cultivated as early as 2000 BC in India and was later introduced to China and Japan around 0 AD. It was grown in Europe in the 13th century and was first grown commercially in Australia in 1969.

In 1988, breeding changes to the oil quality of rapeseed produced canola (Canadian oil low acid). Canola varieties must have oil that contains less than 2% **erucic acid**, and the meal must contain less than 30 µmol g^{-1} **glucosinolates**. Australian canola varieties typically contain less than 0.5% erucic acid and less than 20 µmol g^{-1} glucosinolates.

Canola seed is crushed to obtain an edible oil that is used in cooking. A by-product of the crushing process is a protein-rich meal that is used for stockfeed.

Canola is the third-largest crop produced in Australia, behind wheat and barley. Australia produces about 2 million tonnes of canola annually. Half is crushed domestically and the other half is exported to Japan, China, Pakistan and Bangladesh.

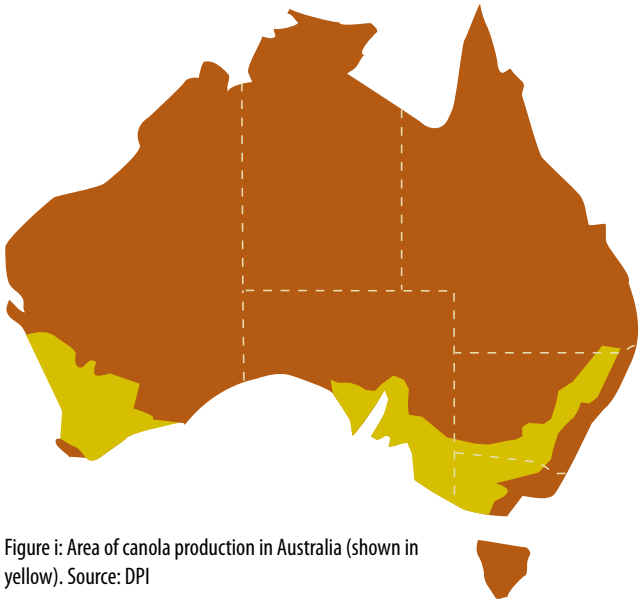


Figure i: Area of canola production in Australia (shown in yellow). Source: DPI

NSW production

New South Wales produces between 20%–25% of the national crop. Although canola is grown in almost all areas of the cropping zone (Figure i), production is centred around central and southern New South Wales, where canola is the main rotational crop. The major limitation of production is the sensitivity of canola to high levels of soil aluminium.

Production of canola in NSW fluctuates with seasonal conditions and price (Figure ii). Production in NSW during the period 1993–2009 averaged 272,500 t/year at an average yield of 1.35 t/ha. In 2000, NSW canola production peaked at 619,000 t, and in the drought year of 2007 only 43,400 t was produced.

Erucic acid
A monounsaturated omega-9 fatty acid.

Glucosinolates
Compounds derived from glucose and amino acids. They are responsible for the bitter, 'hot' taste associated with condiment mustard. When seeds are crushed, the glucosinolates end up in the seed meal.

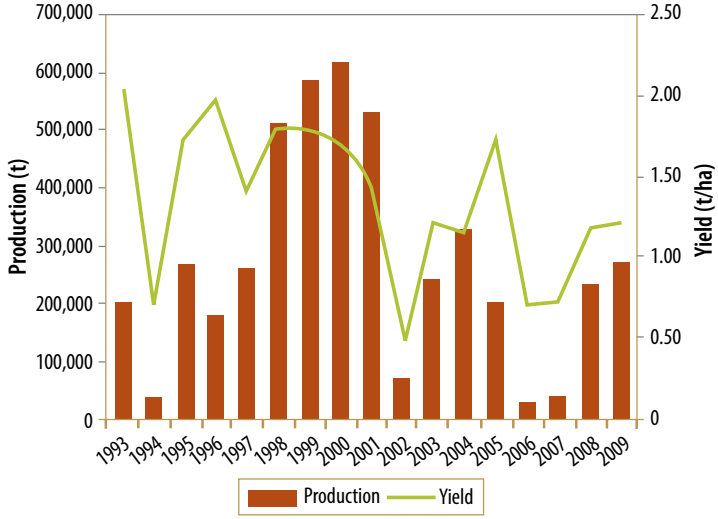


Figure ii. Production and yield of canola in NSW from 1993–2010. Source: Scott (2010)

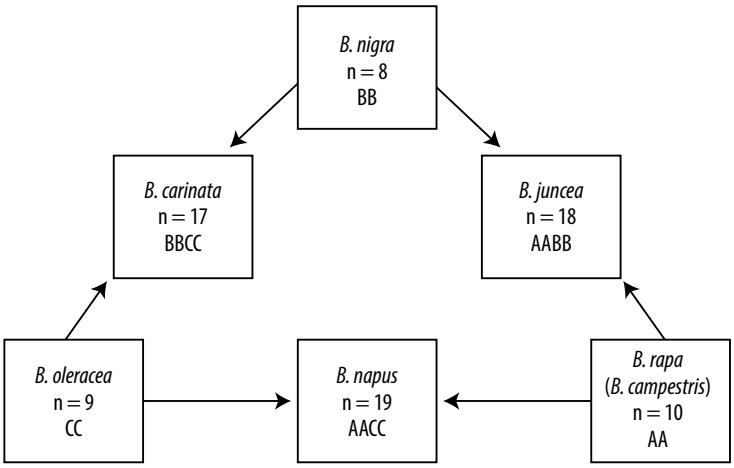


Figure iii. The 'Triangle of U' showing the relationship between the brassica species and the origin of *Brassica napus*. Source: U (1935), Office of the Gene Technology Regulator (2008)

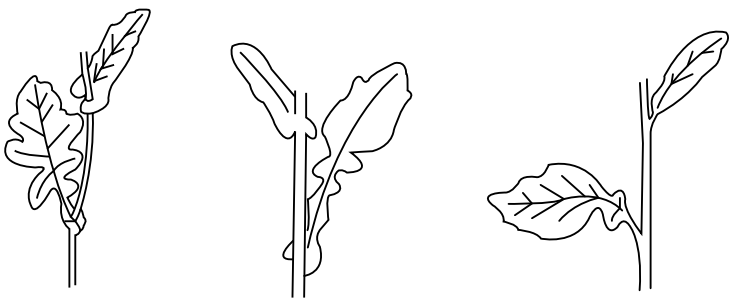


Figure iv. Distinguishing *B. napus* from other brassicas. The leaves of canola (*Brassica napus*) (centre) only half-grasp the stalk. The leaves of turnip (*Brassica campestris*) (left) grasp the stalk completely. The leaves of mustard (*Brassica juncea*) (right) do not reach the stalk. Source: Bengtsson et al. (1972)

Canola types

Conventional

The first rapeseed varieties were introduced into Australia from Europe and Canada in 1969. Under Australian conditions these varieties were late flowering (and so restricted to the higher rainfall zones) and very susceptible to blackleg.

From 1970 to 1988, conventional breeding techniques were used to improve yield, adaptation, blackleg resistance and seed quality (low erucic acid, low glucosinolates). These varieties were based on *B. rapa* (formerly known as *B. campestris*). They had earlier maturity and tolerance to pod shattering.

In 1988 the first varieties were released that combined blackleg resistance with higher yield. These varieties were based on *B. napus* material from Asia and Europe. From this time, there was a complete swing to breeding *B. napus* varieties.

Brassica napus is thought to have formed originally from natural crosses (hybridisation) of *B. rapa* and *B. oleracea* (Figure iii). It is distinguished from other species by the shape of the upper leaves: the lower part of the leaf blade half-grasps the stalk (Figure iv).

Triazine-tolerant canola

Triazine-tolerant (TT) varieties were first commercialised in 1993, with the release of the variety Siren. Genes for tolerance to the triazine group of herbicides were bred into conventional canola varieties. This enabled the control of *Brassica* weeds, which were previously unable to be controlled in standard canola varieties.

The triazine-tolerant trait is associated with reduced conversion of sunlight into biomass (i.e. reduced radiation-use efficiency). Triazine-tolerant varieties are therefore generally less vigorous as seedlings and produce less biomass than conventional varieties. This results in 10% to 15% lower yields and 1% to 3% lower oil contents than in conventional varieties. Another effect of the triazine-tolerant trait is a delay in plant development.

Hybrids

Hybrids were first released in 1988. Hybrid varieties are produced using controlled pollination of a female parent by a male parent (the source of pollen). The progeny (the F1 hybrid) contain the best characteristics of both parents, known as hybrid vigour. Hybrid varieties are typically associated with larger seeds, strong seedling vigour and greater biomass production.

Specialty canola – high oleic/low linolenic (HOLL)

Specialty canolas were bred by traditional means to increase the content of the monounsaturated fat oleic acid and decrease the level of the polyunsaturated fat linolenic acid in the oil. This type of oil is more stable at higher temperatures and more suited for deep frying. This gave a high oleic/low linolenic (HOLL) canola.

IMI-tolerant canola

IMI-tolerant varieties are tolerant to imidazolinones (IMIs), the active ingredients of herbicides such as OnDuty® and Intervix®. They are grown as part of the CLEARFIELD® production system. IMI-tolerant canola varieties were developed by selection of naturally occurring mutations from conventional canola varieties. Unlike the TT gene, the gene for IMI tolerance is not associated with a yield penalty.

Condiment (Indian) mustard

Condiment mustards are varieties of *Brassica juncea* grown for their hot, peppery taste. Although related to juncea canola (see below), condiment mustards have different meal and oil qualities. The level of glucosinolates in the meal after crushing is much higher in condiment mustard and is responsible for the hot and spicy taste of table mustard. The oil has a distinct 'nutty' flavour, but the erucic acid level is sufficiently low to make it suitable for human consumption. Indian mustard is the preferred oilseed in many parts of South Asia, northern and western China and eastern Russia. It has a reputation for having greater drought and shattering tolerance than canola.

Juncea canola – *Brassica juncea*

Juncea canola is the name given to plants bred from *Brassica juncea* to have all the oil and meal quality specifications of canola. The oil has high levels of oleic acid and low levels of erucic acid, and there are low levels of glucosinolates in the meal (Table i).The meal can be substituted for canola meal in animal diets. Juncea canola has the same market end-use as canola.

Table i. Typical seed quality characteristics for canola, juncea canola and condiment mustard when grown in the low rainfall zone.

CHARACTERISTIC	CANOLA	JUNCEA CANOLA	CONDIMENT MUSTARD
Oil %	36–42	34–40	34–40
Oleic acid %	57–63	57–63	variable
Linoleic acid %	18–25	18–25	variable
Linolenic acid %	8–13	8–13	variable
Erucic acid %	<1	< 1	1–20
Glucosinolate in meal (µmol/g – 10% MC)	< 30	< 30	110–160
Allyl glucosinolate in meal (µmol/g – 10% MC)	0	< 1	NA

Source: DPI

Growth
The accumulation of dry matter.

Development
The progression from vegetative growth to reproductive development.

Juncea canola is being developed as a drought- and heat-tolerant alternative to canola for the low rainfall zone. It also has excellent seedling vigour (similar to that of hybrid canola) and is more tolerant of shattering than canola. Because it is a relatively new crop, breeding, selection and agronomic research have not progressed as far as with canola. The first commercial varieties were grown in 2007.

Roundup Ready®

Roundup Ready® (RR) varieties have been bred by genetic modification technology to be tolerant of the herbicide glyphosate. This allows glyphosate to be sprayed over canola in the early stages of growth without affecting the development of the crop. The first varieties were grown commercially in 2008.

Industrial mustard

Industrial mustard is a *B. juncea* type that is not suitable for either of the edible markets because of its high levels of erucic acid and/or glucosinolates. Industrial mustard is grown for use in a number of industrial products, including biodiesel.

Winter types for grazing

Unlike the other canola varieties, which are spring types, winter types require a period of cold (vernalisation) before they will flower. This makes them suitable for a dual-purpose role. They can be grazed during winter, then locked up for harvest in late spring. There is currently only one winter type commercially available.

Life cycle

Growth and **development** of the canola plant is a complex process. During the life cycle of the plant, many of the growth stages overlap.

Growth and development are continuous but can be divided into easily recognizable stages. These are shown in Table ii. The length of each growth stage is influenced by temperature, moisture, light (day length), nutrition and variety, with temperature and moisture being the most important environmental factors regulating growth and development of canola.

Defining canola growth stages

Canola has seven principal stages (see Table ii and Figure v). These stages overlap:

- germination and emergence (stage 0)
- leaf production (stage 1)
- stem extension (stage 2)
- flower bud development (stage 3)
- flowering (stage 4)
- pod development (stage 5)
- seed development (stage 6).

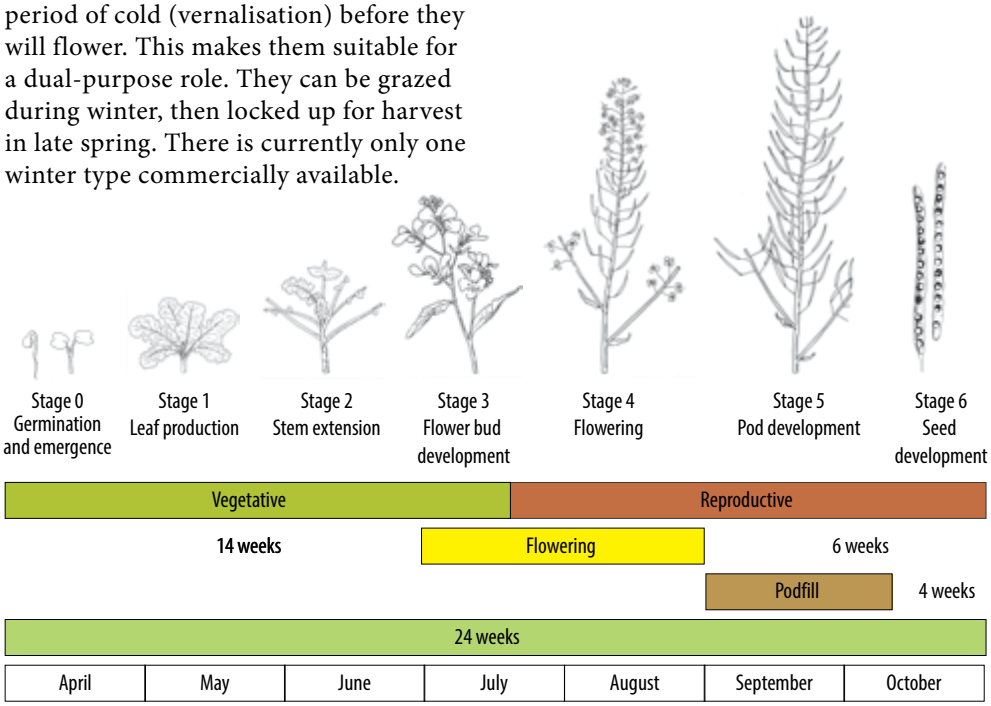


Figure v. Growth stages of canola. Source: DPI

Table ii. Stages of development in canola (*B. napus*).

STAGE	DECIMAL SCORE
Dry seed	0.0
Imbibed seed	0.2
Radicle emerged	0.4
Hypocotyl extended	0.6
Cotyledons emerged	0.8
LEAF PRODUCTION (LOST LEAVES ARE COUNTED BY THEIR SCARS)	
Both cotyledons unfolded and green	1.00
1st true leaf exposed	1.01
2nd true leaf exposed	1.02
5th true leaf exposed	1.05
10th true leaf exposed	1.10
20th true leaf exposed	1.20
STEM EXTENSION	
No internodes detectable (rosette)	2.00
One internode detectable	2.01
Two internodes detectable	2.02
Five internodes detectable	2.05
Ten internodes detectable	2.10
Twenty internodes detectable	2.20
FLOWER BUD DEVELOPMENT	
Only leaf buds present	3.0
Flower buds present but enclosed by leaves	3.1
Flower buds visible from above (green bud)	3.3
Flower buds raised above leaves	3.5
First flower stalks extending	3.6
First buds yellow (yellow bud)	3.7
More than half of flower buds on raceme yellow	3.9
FLOWERING	
1st flowers opened	4.1
20% of all buds on raceme flowering or flowered	4.2
50% of all buds on raceme flowering or flowered	4.5
80% of all buds on raceme flowering or flowered	4.8
All viable buds on raceme finished flowering	4.9
POD DEVELOPMENT	
Lowest pods more than 2 cm long	5.1
20% of potential pods on raceme more than 2 cm long	5.2
50% of potential pods on raceme more than 2 cm long	5.5
80% of potential pods on raceme more than 2 cm long	5.8
All potential pods on raceme more than 2 cm long	5.9
SEED DEVELOPMENT	
Seeds present	6.1
Most seeds translucent	6.2
Most seeds green	6.3
Most seeds green-brown mottled	6.4
Most seeds brown	6.5
Most seeds dark brown	6.6
Most seeds black but soft	6.7
Most seeds black and hard	6.8
All seeds black and hard	6.9

Source: Modified from Sylvester-Bradley and Makepeace (1984)

Decimal growth scale

Effective crop management depends on being able to correctly identify the growth stage of the crop. A growth scale provides a common reference for describing growth stages. An example of where this is important is in the timing of fertiliser and chemical applications.

The canola plant

Canola is an annual plant that grows to between 70 and 170 cm high. In Australia canola is a winter-growing oilseed crop that is sown in autumn and matures in late spring, giving a 5- to 6-month growing season. The main structures of the canola plant are the leaves, stem, branches, roots, pods and seeds (Figure vi).

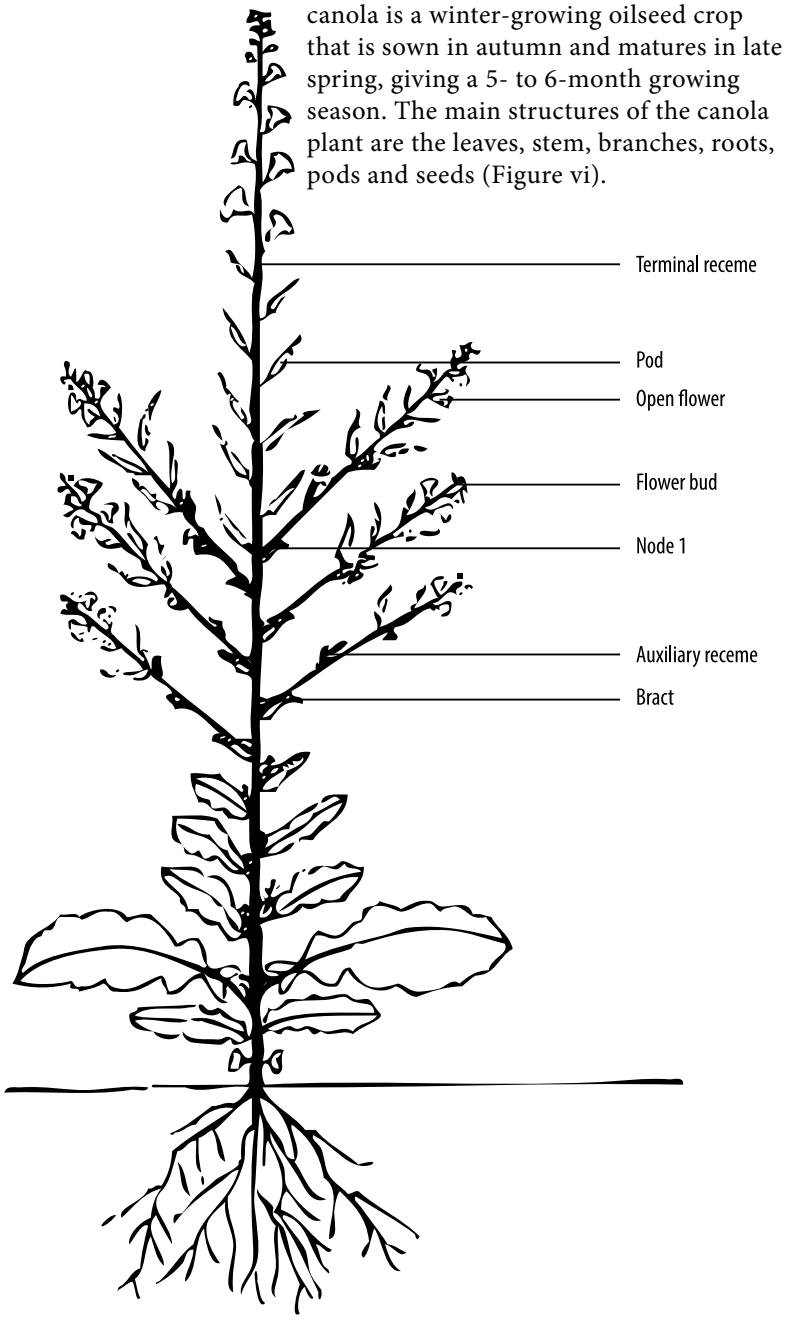


Figure vi. The mature canola plant. Source: Adapted from Tayo and Morgan (1975)

Leaves

The first true leaf to develop and fully expand appears frilly or ruffled at the edges. Each leaf is attached to the stem at a node.

The canola plant does not produce a definite number of leaves. A well grown canola plant can produce between 10 and 15 leaves. The oldest leaves at the base of the plant are the largest, and the youngest leaves at the top of the plant are the smallest (Figures vii, viii and ix).

The leaves of the mustard plant (*B. juncea*) are a lighter green and have indented vein patterns. The mustard leaf appears rippled compared with the surface of a canola leaf (see Figure ix).

Figure vii: The leaf of *B. napus*. Source: F. Tome

Figure viii: The leaf sheath of *B. napus*. Source: F. Tome

Stem and branches

The canola plant has a main stem that supports the plant (Figure x). Along the stem are the internodes: there are between 15 and 20 nodes per stem, at a spacing of 5 to 10 cm. The main stem elongates to between 70 and 170 cm, reaching its maximum length at flowering. At the end of the main stem is a flower head.

The stem is an important photosynthetic structure throughout the period of pod and seed growth.

The thickness of the stem is related to plant density. Plants in low-density crops have thicker stems and are more resistant to **lodging**.

By the time the plant matures, the tissues in the centre of the stem have usually collapsed, leaving a hollow, fibrous tube.

Branches form from buds on the main stem. The buds develop in the axil of the leaves. The branches develop one to four leaves and a flower bud.

Lodging
Describes when the crop canopy lies over or leans towards the horizontal, usually because of heavy rain and wind. Sometimes described as 'tabling'.

Figure x. Canola stem. Photo: Lowan Turton, DPI

Roots

Canola plants have a taproot system. The taproot acts as a reservoir for nutrients and **assimilates**. Secondary roots develop from the taproot, growing outward and downward. The root system of a mature canola plant can penetrate as deeply as 120 cm.

Canola has abundant fine roots with the ability to branch and proliferate in zones of higher nutrient content, such as around fertiliser bands or granules. In addition, canola roots can increase their root hair number and length in response to low phosphorus conditions.

Flowers

The canola plant is **indeterminate**, and it is able to initiate new reproductive flower heads throughout most of its life.

Canola flowers are yellow and consist of four petals and six stamens (Figure xi). The four petals, which narrow at the base, form the shape of a cross, which is where the original family name of 'Cruciferae' (now Brassicaceae) comes from. The flowers develop in terminal **racemes** (bunches at the end of the stem).

The female part of the flower is called the carpel. The carpel consists of three parts:

- the ovary, which becomes the seed after fertilisation
- styles, which are extensions of the ovary
- stigmas, which are specialised filaments on which the pollen falls and germinates.

Figure xi. Canola flower head. Photo: Lowan Turton, DPI

Pods

The seed pod (sometimes called a silique) is an elongated capsule with a prominent mid vein. The pod is approximately 6 to 9 cm long. The pod has a beak or tip 1 to 2 cm long. It is made up of two carpels separated by a false septum (Figure xii). A mature seed pod is fragile and can easily shatter, causing loss of seeds.

The pods on a mustard plant (either condiment or juncea canola) are held more upright than on a canola plant (Figure xiii).

Assimilates
Products of photosynthesis.

Indeterminate
Some plants like canola and the pulse crops are indeterminate; that is, they flower over a long period of time and can initiate new flowers when conditions are favourable. This is in contrast with determinate crops such as wheat, which flower once in a short period.

Raceme
An unbranched flower head with flower buds connected by short stalks called pedicles. This creates a bunch of flowers at the end of the stem.

The seed

Seeds begin developing first on the lowest third of the branches of the main stem. The seeds mature 30 to 40 days after fertilisation. Canola seed is black, whereas mustard seed is yellow.

Between 15 and 25 seeds are produced per pod. At maturity, the seed makes up 60% of the weight of the whole pod.

There are approximately 280 000 to 300 000 seeds per kilogram (Table iii). Each seed weighs between 2.5 and 5 mg.

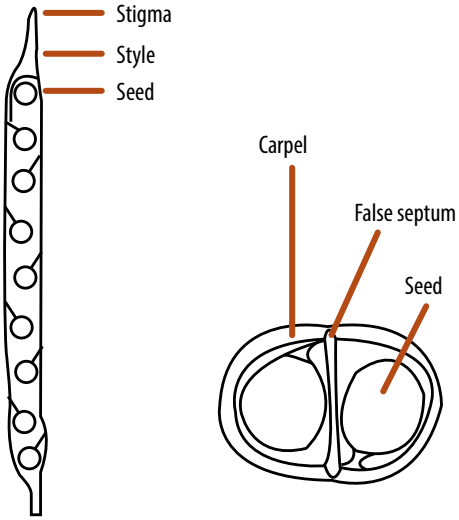


Figure xii. The brassica pod shown lengthwise (left) and as a cross-section (right). Source: Bengtsson et al. (1972)

Table iii. Normal variation in seed size of different types of brassica oilseeds.

OILSEED TYPE	1000-SEED WEIGHT (G)
Winter canola	4.5–5.5
Winter turnip canola	3.0–4.0
Summer rape	3.5–4.5
Summer turnip rape	2.0–3.0
Brown mustard	2.0–3.0

Source: Bengtsson et al. (1972)

The canola seed can be broadly divided into three components (Figure xiv):

- **seed coat** (pericarp) and **aleurone** (or bran) **layer**
- **endosperm**
- embryo

The seed coat (the outer protective covering of the seed) accounts for between 12% and 16% of the seed weight. It has a low oil content and high fibre content. The aleurone layer lies just underneath the seed coat and is rich in protein.

Brassica species have very little endosperm compared with other plant seeds, such as those of cereals. The thin layer of endosperm surrounds the embryo.

Seed coat (pericarp – testa)
The outer protective covering of the seed.

Aleurone layer
Aleurone (from Greek *aleuron*, flour) is a protein found in the protein granules of maturing seeds. The aleurone layer is the outermost cell layer of the seed coat.

Endosperm
A nutritive tissue within the seed surrounding the embryo. The endosperm in the mature canola seed is very small.

Figure xiii. Difference between the shape of the pods on a mustard plant (left) and a canola plant (right) Photos: Michel Dignaud, DPI

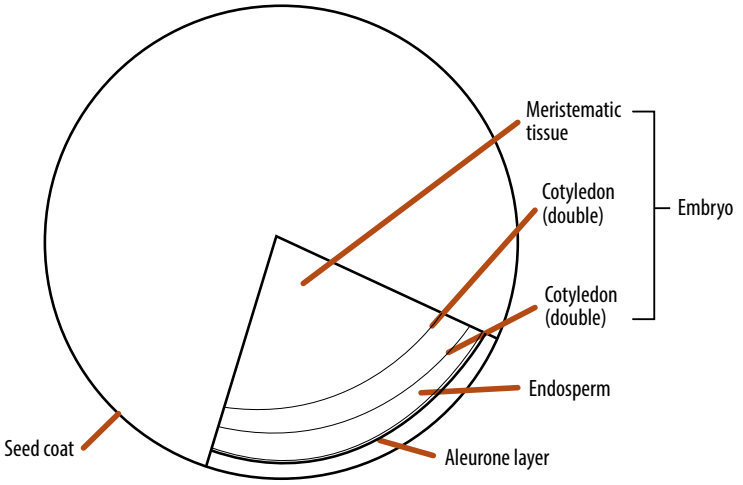


Figure xiv. The structure of a canola seed. Source: Adapted from Bengtsson et al. (1972)

Cotyledons
Embryonic leaves that emerge before the first true leaves. The cotyledons of *B. napus* seedlings are smooth on the underside, whereas the cotyledons of *B. rapa* are hairy and wrinkled. The cotyledons serve as the food storage organs in canola.

Ovule
The part of the plant that, after fertilisation, develops into the seed.

Meristematic tissue
Central part of the canola seed from which the radicle (root), hypocotyl (stem) and epicotyl (bud) develop.

Radicle
Part of the seed embryo that grows into the primary root.

Most of the embryo consists of two **cotyledons**, which contain 30% to 50% oil (Figure xv). The cotyledons are folded so that one occupies the outer space in the **ovule** and enfolds the other, 'inner', cotyledon. About 80% of the oil in the canola seed is concentrated in droplets in the cells of the cotyledon. Also within the embryo is the **meristematic tissue**. From this tissue the **radicle** (root), hypocotyl (stem) and epicotyl (bud) develop.

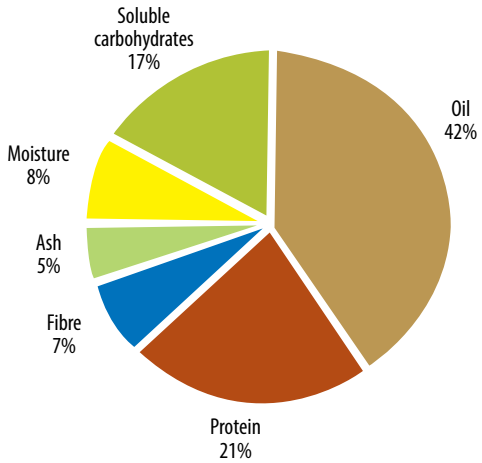


Figure xv. Major components of the canola seed. Source: Adapted from NSW Agriculture, Canola Agfact

At maturity, the canola seed contains:

- 70% of the P, N, Zn, Fe and Mg
- 30% to 35% of the Cu, Mn, S, Ca and K
- less than 20% of the Na and Cl.

The rest is in the pod walls.

Canola oil

Oil is extracted by mechanically crushing the seed. The oil is then processed by using heat and/or chemicals. Approximately 73% of canola in Australia is processed by addition of solvents, 25% by expeller treatment and 2% by cold-pressing.

The seed typically has an oil content ranging from 35% to 45%. The oil content is generally expressed as a percentage of the whole seed at 8% moisture content. The oil contains:

- 10% to 12% linolenic acid (omega-3)
- < 0.1% erucic acid
- 59% to 62% oleic acid
- 12% to 22% linoleic acid.

Canola oil is high in unsaturated fats (93%) and has no cholesterol or trans-fats. It has the lowest saturated fat content (7%) of any common edible oil. When canola is processed to form canola oil, all traces of protein are removed from the residue that becomes the seed meal.

Seed meal

The seed meal is what is left over after the oil is removed. It contains proteins, carbohydrates, minerals and fibre. The exact composition of seed meal depends on the oil extraction method. The protein content varies each season and increases as the oil content decreases. Typically, seed meal consists of between 36% and 39% protein, 1.5% to 2% fat, 11% to 13% fibre and less than 10 µmol g^{-1} glucosinolate.

The minimum protein content of seed meal, as determined by the AOF (Australian Oilseeds Federation) is 36%, measured at 12% moisture.

References and further reading

AOF 2007, *Australian Canola Meal Guide for the Feed Industry*. Australian Oilseeds Federation, Sydney

AOF 2007, *Quality of Australian Canola*. Australian Oilseeds Federation, Sydney

Appelqvist LA, Ohlson R (eds) 1972, *Rapeseed—Cultivation, Composition, Processing and Utilisation*. Elsevier Publishing Company, London, New York, Amsterdam.

Bengtsson L, Von Hofsten A, Loof B 1972, Botany of rapeseed. Chapter 3 in *Rapeseed – Cultivation, Composition, Processing and Utilisation*. (Appelqvist L, Ohlson R eds). Elsevier, Amsterdam, London, New York.

Bell JM, Hickling D 2003, Canola meal feed industry guide. Section II. Composition of canola meal. Canola Council of Canada. www.canola-council.org

Beversdorf WD, Weiss-Lerman J, Erickson LR, Souza Machado V 1980, Transfer of cytoplasmically-inherited triazine resistance from bird's rape to cultivated oilseed rape. (*Brassica campestris* and *B. napus*). *Canadian Journal of Genetics and Cytology* 22, 167–172.

Colton B, Potter T 1999, History. In *Canola in Australia: the First Thirty Years* (Salisbury PA, Potter TD, McDonald G, Green AG eds). Organising Committee of the 10th International Rapeseed Congress, Canberra.

Mailer R 1999, Product quality. Chapter 14 in *Canola in Australia: the First Thirty Years*. (Salisbury PA, Potter TD, McDonald G, Green AG eds). Organising Committee of the 10th International Rapeseed Congress, Canberra, pp. 71–74.

Mailer R 1999, Quality of Australian canola. In *Canola in Australia: the First Thirty Years* (Salisbury PA, Potter TD, McDonald G, Green AG eds). Organising Committee of the 10th International Rapeseed Congress, Canberra.

Office of the Gene Technology Regulator 2008, Biology of *Brassica napus* L. (Canola), p. 2. www.ogtr.gov.au/internet/ogtr/publishing.nsf/content/canola-3/$FILE/biologycanola08_2.pdf

www.ogtr.gov.au/internet/ogtr/publishing.nsf/Content/canola-3/$FILE/biologycanola08_2.pdf

Ryan MH, Kirkegaard JA, Angus JF 2006, *Brassica* crops stimulate soil mineral N accumulation. *Australian Journal of Agricultural Research* 44: 367–377.

Scarisbrick DH, Daniels RW (eds) 1986, Oilseed rape physiology. Chapter 3 in *Oilseed Rape*. Collins, London

Scott F 2010, NSW Grains Report Summary 1993–2010, Industry & Investment NSW, Orange. www.dpi.nsw.gov.au/aboutus/resources/periodicals/newsletters/grains-report-nsw

Sylvester-Bradley R, Makepeace RJ 1984, A code for stages of development in oilseed rape (*Brassica napus* L.). In *Aspects of Applied Biology 6: Agronomy, Physiology, Plant Breeding and Crop Protection of Oilseed Rape*. 26–28 March 1984. Association of Applied Biologists, Churchill College, Cambridge UK, 399–419.

Tayo TO, Morgan DG 1975, Quantitative analysis of the growth, development and distribution of flowers and pods in oil seed rape. *Journal of Agricultural Science*, Cambridge 85, 103–110.

U N 1935, Genome analysis in *Brassica* with special reference to the experimental formation of *B. napus* and peculiar mode of fertilization. *Japanese Journal of Botany* 7, 389–452.

1. Germination and emergence

by Kathi Hertel and Jan Edwards

Chapter Snapshot

Introduction

Under the right conditions, a viable canola seed germinates. Chapter 1 is about the processes by which the first shoot emerges from the ground and root growth begins. The phases covered in this chapter are germination, emergence and establishment.

Learning Outcomes

At the end of this chapter, you will be able to:

- describe the germination process and the roles of moisture, temperature and oxygen
- explain plant emergence and establishment and the roles of moisture and temperature
- understand the factors that influence hypocotyl length
- recognise the qualities to look for when selecting seed
- conduct a germination test to determine germination percentage

Radicle
Part of the seed embryo that grows into the primary root.

Germination – Growth stages 0.0–0.3

Germination starts after the seed absorbs moisture and ends when the **radicle** splits the seed coat and the shoot pushes upward through the soil.

The energy for germination comes from the oil (lipids) stored in the cotyledons in the seed. The lipids are converted to sucrose, which provides the energy source for the seedling until it begins photosynthesis. Amino acids and essential proteins are also used during early seedling growth.

Germination has three phases, each characterised by large changes in rates of water absorption (Figure 1–1). These phases are:

- rapid hydration
- lag period
- a second phase of rapid hydration, with visible germination

Figure 1–2. Germinating canola seeds, showing swelling, then splitting of the seed coat, emergence and extension of the radicle, and emergence of the cotyledons. Photos: Lowan Turton, DPI

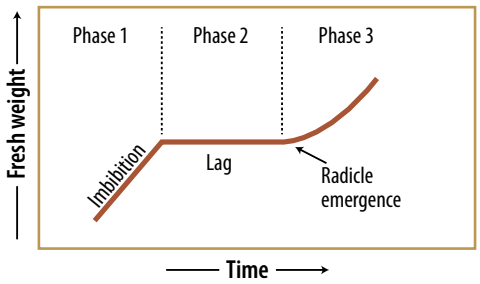


Figure 1–1. Germination phases of canola. Source: Boyd and Van Acker (2004)

Phase I – Water absorption

Before planting, canola seed has a moisture content of between 7% and 8%. After the seed is placed in the soil it imbibes (absorbs) water, swells and becomes sticky (because of the presence of a gelatinous substance in the seed coat). Seed metabolic activity increases.

The speed of water absorption depends on soil moisture content and seed size. Small seeds such as canola have a large surface-to-volume ratio, so imbibition happens very quickly.

Phase II – Lag phase

During the lag phase, seed metabolism increases but the seed water content changes very little. Enzymes in the embryo are activated and break down stored proteins to amino acids, starch to glucose, and oil to fatty acids and glycerol. These are translocated to the active growing point in the seed, where they are used for embryo growth. It is possible at this point for germination to be suspended if the soil is very dry.

Phase III – Visible germination

A second period of rapid water absorption occurs. The seed continues to imbibe until it reaches a moisture content of about 24%. This will be about 12 hours after Phase I began. The seed is now at a 'point of no return'. It will not survive desiccation at this point.

The seed coat swells and ruptures. Approximately 6 hours later, the radicle emerges through the protective seed coat (Figure 1–2). This is the first visible sign of germination and marks the end of germination and the start of seedling

growth. The phase concludes with the emergence of the hypocotyl.

The emergence of the radicle causes breakdown of the endosperm and the seed coat. This releases sugars and signal molecules, which are thought to trigger the expression of defence genes that protect the emerging seedling from pathogen attack.

Emergence – Growth stages 0.6–1.0

Emergence takes 4 to 15 days, depending on soil temperature. The number of days to 50% emergence is important, as the first 50% of seedlings that emerge account for the majority of yield.

Canola emergence is referred to as **epigeal** – that is the **hypocotyl** pulls the heart-shaped cotyledons above the soil surface. Once at the surface and exposed to light, the cotyledons turn green and begin photosynthesis. The cotyledons allow the very young seedling to photosynthesise until the emergence of the first true leaves (Figure 1–3).

The small size of the canola cotyledon makes the plant vulnerable during emergence and early establishment. The growing point is above the soil, between the two cotyledons. With the growing tip exposed, canola seedlings are susceptible to insects, frosts, soil erosion, hail or any other hazard that can damage the plant below the cotyledons.

Establishment

The plant is established once it has roots and a shoot. It is no longer relying on reserves in the seed, as it is producing its own energy. The leaves are now able to photosynthesise and the roots are able to take up water and nutrients.

A crop is said to be established when 50% of the plants have germinated and emerged and are developing with strong seedling vigour.

Factors affecting germination and emergence

Dormancy

There are two types of **dormancy**: **primary** and **secondary**. Canola seed has no primary dormancy. However, secondary dormancy may be induced in some varieties if the seed is exposed to low soil water levels. Other factors that can induce secondary dormancy are long periods of darkness, low oxygen supply, and temperatures greater than 20°C.

Once dormant, seeds will not germinate. Secondary dormancy can be removed if the seeds are exposed to low temperature (2°C to 4°C) or alternating warm and cold temperatures.

Figure 1–3. Emergence of a canola plant. Left: cotyledons. Right: first true leaf. Photos: Jan Edwards

Epigeal emergence
Cotyledons are raised above the soil surface by the hypocotyl. By comparison, in wheat and other cereals the cotyledons remain below the ground (hypogeal).

Hypocotyl
Part of the seedling between the crown and the roots. A pinched hypocotyl is an indication of blackleg.

Primary dormancy
Caused by the interaction of abscisic acid and environmental conditions in the late stages of seed development. Primary dormancy prevents the seeds from germinating immediately after harvest.

Secondary dormancy
Induced by stresses (e.g. light, moisture and temperature) during seed development, seed storage or germination.

Abscisic acid
A dormancy hormone found in seeds. It can inhibit water uptake by the embryo tissues and therefore inhibit seed germination.

Moisture

Soil moisture is critical for both germination and emergence. Canola has to absorb a high percentage of its weight in water before germination begins. It will germinate when the seed moisture content has risen to about 24%.

Water absorption is a passive process. The ability of seeds to absorb water depends on the difference in water potential between the seed and the surrounding soil. Seeds can absorb water even at very low soil water potentials, but low water potentials may induce secondary dormancy (see previous page).

Seed size influences the rate of seed water absorption. Small seeds have a large surface-to-volume ratio, which means that less time is required to absorb adequate moisture for germination.

In soils with a low moisture content, the germination rate will be lower and emergence slower (Table 1–1).

Table 1–1. Effect of soil moisture content on final emergence percentage and days to 50% emergence

TOTAL SOIL WATER CONTENT (% WEIGHT)	FINAL EMERGENCE %	DAYS TO 50% EMERGENCE
18	82	9
15	59	12
13	45	13
11	4	–

Source: Modified from Canola Council of Canada (2003)

- A trial was established in a growth chamber at a constant temperature of 8.5°C/10°C (day/night).
- The higher the total soil water content, the higher the final germination percentage.
- The higher the soil water content, the quicker the time to 50% seedling emergence.

Waterlogging

Canola is sensitive to waterlogging during germination. When soils become waterlogged, the oxygen supply in the soil solution rapidly decreases. Oxygen is essential for seed germination. Without oxygen, seeds cannot continue their metabolic processes, and germination ceases. Prolonged waterlogging can kill canola seeds and seedlings.

Temperature

Germination

Low temperature affects both the process and the rate of germination. The period of transition from Phase II to III of germination is believed to be the most sensitive to temperature.

Low temperatures slow the rate of water absorption and reduce the production of proteins required for germination. Sustained low temperatures damage the seed embryo. The result is slow, 'staggered' germination, resulting in poor crop establishment.

Once the seeds have absorbed enough water, soil temperature plays a major role in advancing germination. The optimum temperature range for canola germination is 15°C to 20°C. Soil temperatures below 10°C result in progressively poorer germination and emergence. At 2°C seeds will absorb water and germinate, but germination rarely occurs at temperatures below 2°C. For example, at Cowra, there is a 10% probability that the 10 cm soil temperature will be less than 10°C on the 8 May (Figure 1–4).

Low temperatures also slow the rate of germination. At low temperatures the protein synthesis needed in the seed for germination is slowed. For example, at 22°C the seed will germinate in 1 day compared with 11 days at 2°C (Figure 1–5).

Emergence

The thermal time requirement for emergence is about 115 **degree-days** (Figure 1–6). From sowing to emergence generally takes between 4 and 15 days. The colder the soil, the longer it will take. Temperatures less than 10°C reduce germination and emergence rates. The combination of reduction in germination rates and longer germination times at temperatures below 10°C results in poor emergence.

Degree-days
Degree-days are a measure of accumulated temperature used to explain the relationship between plant development and temperature. This accumulated temperature is calculated as the average daily temperature minus a base temperature and is recorded as degree-days (°Cd). The base temperature is the minimum temperature at which the plant grows, and this varies for each crop. For canola, the base temperature is 0°C during vegetative growth and 5°C in the reproductive phase.

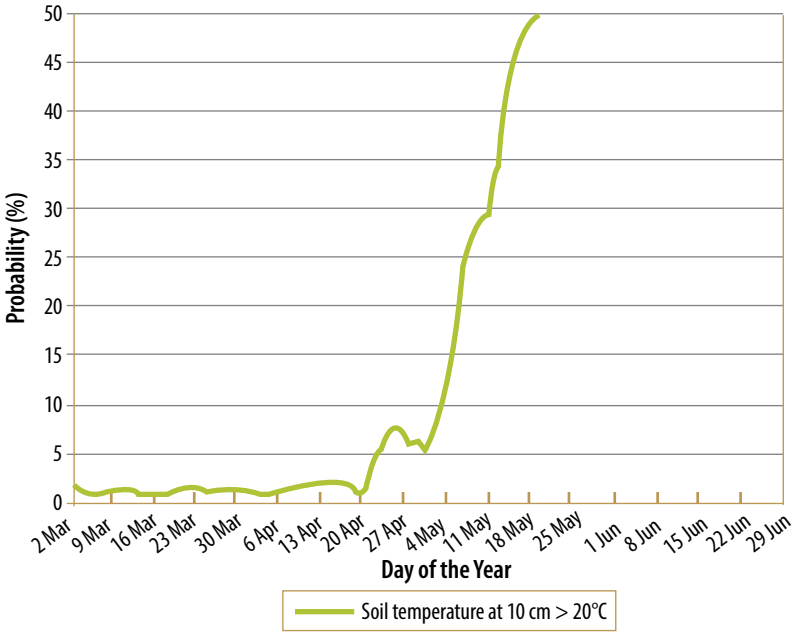
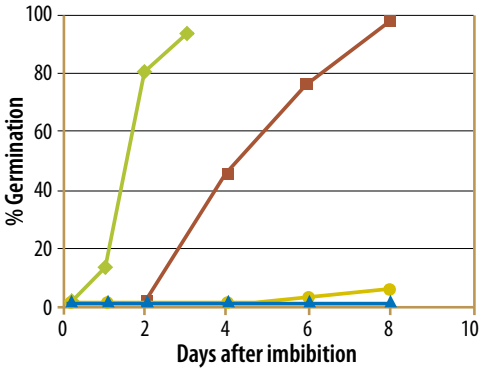
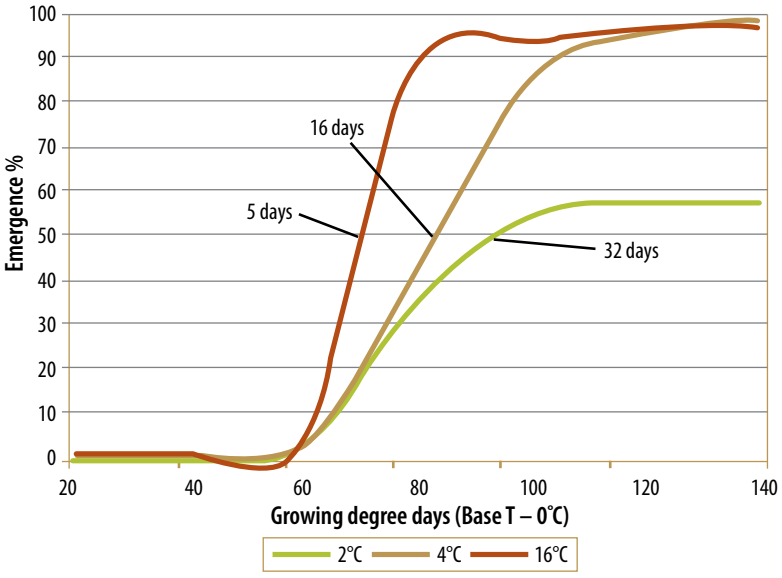


Figure 1–4. Probability that the 9 am soil temperature at 10 cm deep is less than 10°C at Cowra, NSW (1960–2009). Source: Data from Bureau of Meteorology and Office of Environment and Heritage



- Germination time varied with temperature
- At 22°C, germination percentage increased rapidly and peaked by day 3.
- At 10°C, germination increased gradually until day 2, then increased rapidly 4 days after imbibitions, reaching 95% germination by day 8.
- At 6°C and 2°C, germination never reached higher than 10%.

Figure 1–5. Effect of temperature on the percentage germination of the *B. napus* variety Westar. ◆ 22°C, ■ 10°C, ● 6°C and ▲ 2°C. Source: Nykiforuk and Johnson-Flanagan (1999)



- The colder the soil, the longer it takes to reach 50% emergence.
- At 2°C it takes 32 days to reach 50% emergence; at 4°C it takes 16 days; at 16°C it takes 5 days.

Figure 1–6. Effect of temperature on emergence percentage. Lines point to times of 50% emergence. Source: Canola Council of Canada (2003)

Establishment

Once established, canola is relatively frost tolerant, but damage can occur during the cotyledon stage and the seedlings can die if frosted. Plants become more frost tolerant as they develop.

Seedling growth and vigour is reduced at temperatures below 7°C, and occasionally seedlings will die.

Soluble carbohydrates accumulate when there is a rapid reduction in leaf temperature. This accumulation suppresses photosynthesis, and therefore seedling growth rates, during the cooler winter months.

Seed size

Canola seeds are smaller than other grains such as wheat, barley or lupins. They weigh only 3 mg each. The 1000-seed weight of canola is typically 3 to 6 g. Seed size varies according to the growing conditions in which the seed was grown. There are also varietal differences. Generally, hybrid varieties have larger seeds.

Seed size plays an important role in crop establishment. Larger seeds produce more vigorous seedlings and improved crop establishment (Table 1–2). There is also an interaction with sowing depth. Larger seeds establish more plants, particularly if sown 3 cm or deeper.

Table 1–2. Effect of seed size and sowing depth on plant establishment.

SEED SIZE (MM)	SOWING DEPTH (CM)			
	4.5	3.0	1.5	MEAN
	NO. OF PLANTS/M²			
> 1.7	41.7	64.2	77.0	61.0
1.4–1.7	26.6	43.2	73.3	47.7
< 1.4	23.0	33.0	78.5	44.8
Mean	30.5	46.8	76.3	

Source: Kathi Hertel, unpublished data, DPI

- Similar plant numbers established when seeds were sown at 1.5 cm deep, regardless of seed size.
- Larger seeds (greater than 1.4 mm) established more plants when sown at 3 and 4.5 cm deep.

Seed quality

Seed quality is important to ensure good establishment. Canola seed should have a germination percentage above 85%. Planting high-quality seed is essential for rapid, even crop establishment. Early seedling growth relies on stored energy reserves in the seed. Good seedling establishment is more likely if the seed is undamaged, stored correctly, and from a plant that has had adequate nutrition.

Seed moisture content, age of seed, seed size and germination percentage all contribute to overall seed quality. There can be substantial differences in the performance of commercial certified seed lots from different sources. These differences can be as large as differences among varieties.

There are a number of factors that can greatly affect germination. They include seed size, seed handling and harvest timing.

Seed size

The larger the seed, the larger the cotyledon and the lipid reserves. Although seed size does not alter germination, larger seeds emerge earlier and faster than medium-sized and small seeds. This is because larger seeds germinate more rapidly and their roots are longer than those of smaller seeds. In adequate moisture, medium-sized seeds will emerge in 5 or 6 days.

Seed size is usually measured by weighing 1000 grains; this is known as the 1000-seed weight. The 1000-seed weight varies among varieties and from season to season. As a result, sowing rates should be altered according to seed weight to achieve the desired plant population.

Harvest timing

The timing of windrowing can also affect germination. If the crop is not windrowed at the correct time, seed development can stop, resulting in unripe seeds with reduced germination ability. See Chapter 4.

Seed chlorophyll

High seed chlorophyll levels can reduce seedling vigour and increase seedling mortality. Chlorophyll levels below 35 mg/kg are desirable. Canola seed harvested from plants suffering frost during seed-filling or severe moisture stress may have elevated chlorophyll levels. See Chapter 4.

Seed handling

Besides seed size, germination can also be affected by seed handling procedures. Care needs to be taken when harvesting canola seed to ensure it is not cracked. Cracking can reduce germination. This topic is covered further in Chapter 4.

Seed storage

The aim of storage is to preserve the viability of the seed for future sowing and maintain its quality for market.

Canola is more difficult to store than cereals because of its oil content. The oil content makes canola more prone to deterioration in storage. For this reason, canola should not be stored on farm for more than one summer.

The rate at which canola deteriorates in storage depends on the:

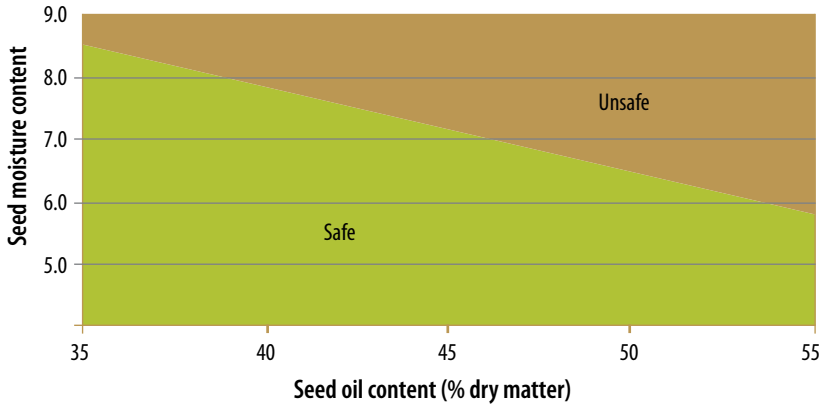
- storage temperature
- seed moisture content
- seed oil content
- relative humidity
- storage time
- percentage of green or immature seeds in the sample
- amount of weathering after physiological maturity.

Monitoring of seed moisture of canola is necessary during storage, as a moisture content of 6% to 8.5% can be unsafe, depending on the seed oil content (Figure 1–7).

High temperatures or moisture can cause a number of reactions in the seed, resulting in:

- increased levels of free fatty acids, causing off-flavours in the oil
- oxidation and browning reactions, which taint the oil
- changes to the oil profile of the seed owing to reactions involving chlorophylls, carotenoid pigments, flavonoids and phenols.

Canola should be stored at or below 8% moisture and at temperatures below 25°C (but preferably below 20°C).



- Safe storage limits are determined by the oil and moisture content of the seed.
- Canola falling into the potentially unsafe area above the line should not be stored for any lengthy period of time unless appropriate action is taken, such as lowering the moisture content and seed temperature.

Figure 1–7. Potential unsafe storage limits for Australian canola varieties (at 60% equilibrium relative humidity and 25°C).
Source: www.australianoilseeds.com/__data/assets/pdf_file/0006/4110/Oilseeds_Flyer.pdf

A seed is a living organism that releases moisture as it respires. Canola can maintain a high respiration rate for up to 6 weeks following harvest before becoming dormant. Moulds can develop in canola within 11 days at 10.6% moisture at 25°C. Poor seed storage can reduce seed quality and reduce emergence (Table 1–3).

Sowing

Soil pH

Canola should not be grown in soils with a pH_{Ca} less than 5.2 or an aluminium concentration above 5%. As soils become more acid, more aluminium is released into the soil solution. The presence of aluminium has no effect on the germination process, but high concentrations have been reported to affect root growth (Figure 1–8). Addition of lime to the soil before planting can increase soil pH, improving emergence and root development (Figure 1–9).

Seed bed

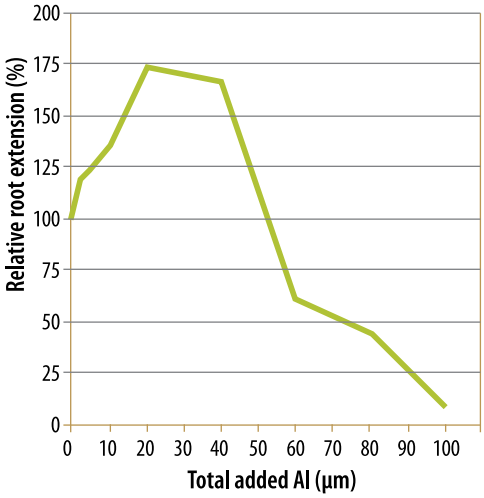
Seed–soil contact, especially under dry conditions, is crucial for helping moisture diffuse into the canola seed. Emergence of canola seedlings can be reduced by the

Table 1–3. Changes in germination percentages of canola samples stored at three temperatures and moisture contents.

STORAGE TEMPERATURE (°C)	MOISTURE CONTENT (%)	OSCAR 1	OSCAR 2	PINNACLE
		Germination (%) before storage		
		99.6	96.5	99.9
		Germination (%) after storage		
20	6	99.8	97.5	99.6
	7	99.9	96.6	99.8
	8	99.8	96.9	99.6
25	6	99.8	97.5	99.8
	7	99.5	96.4	99.3
	8	99.5	92.4	99.6
30	6	99.9	97.3	99.5
	7	96.5	71.6	95.6
	8	94.4	26.3	87.0

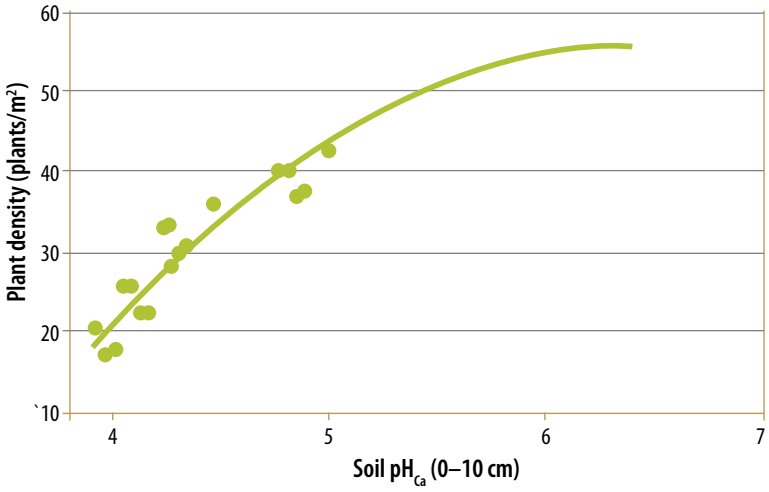
Source: Reuss and Cassells (2003)

- A high moisture content and warm temperatures during storage for 18 months led to decreased germination.
- The effect of temperature on germination percentage was greater if the moisture content was also high.
- When temperature and moisture content are increased during storage, germination rates fall.



- Root growth of 4-day-old canola seedlings exposed to aluminium concentrations of between 20 and 40 µm was up to 75% greater than that of control seedlings.
- Higher aluminium concentrations caused the tap root to become stunted and reduced secondary root development and the formation of lateral roots.

Figure 1–8. Effects of aluminium on canola roots. Source: Clune and Copeland (1999)



- There was an increase in density of canola from 17 to 60 plants/m^2 with increasing rates of lime application.
- Canola emergence increased by 15% for each unit increase in soil pH$_{Ca}$ above 5.2.

Figure 1–9. Effects of lime on canola emergence and plant population. Source: Scott et al. (2003)

formation of soil crusts in hardsetting, **sodic or dispersing soils**. Sodic or dispersing soils that surface-seal will reduce the emergence of canola seedlings.

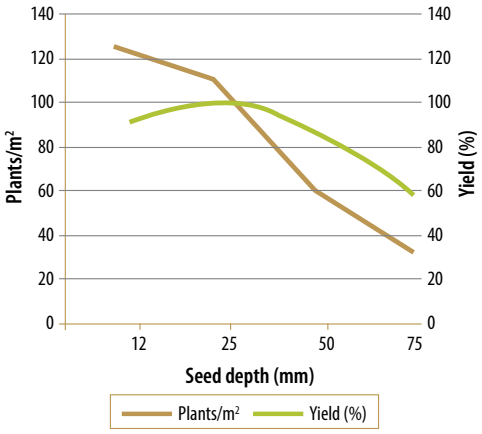
A firm, moist seedbed provides uniform seed germination and rapid seedling growth. Adequate soil moisture at the seedling and elongation stages promotes the development of a strong, healthy plant less prone to lodging and with a maximum amount of leaf growth by the end of July.

Sowing depth

Sowing depth has a major influence on seedling vigour, which subsequently affects seedling establishment and crop performance (Figure 1–10 and Table 1–4). A sowing depth of between 1.2 and 2.5 cm is ideal.

Deep seed placement increases the risk of failed emergence. Deeper-sown seeds grow longer hypocotyls and have shorter root systems, smaller leaf area and less leaf and root dry mass. Leaf expansion is slower.

Sodic or dispersing soil
In a sodic soil a high proportion of sodium ions relative to other positive ions is present in the soil or the soil water. These soils are 'dispersing', because when they are wetted the clay particles are forced apart.



- Plant population and yield were highest when canola was sown at 25 mm.
- When sown shallower than 25 mm, seed was susceptible to dryness and heat.
- When the seed was sown deeper than 25 mm, where the soil was colder, the rate of emergence was reduced.

Figure 1–10. Effect of sowing depth on plant population and yield. Source: Modified from Canola Council of Canada (2003)

Deeper sowing reduces light, and the hypocotyl responds to this by elongating, reducing the chance of seedling emergence. Plants with longer hypocotyls have reduced leaf area from an early stage. Leaves are also slower to expand reducing dry matter. The longer hypocotyls are thinner and more susceptible to mechanical damage. Root length is also reduced.

The hypocotyl is the shoot that emerges from the seed. Seeds planted more than 2 cm deep or into more than 5 t/ha of stubble develop elongated hypocotyls. This depletes the seed reserves more quickly than in seeds with shorter hypocotyls. This can contribute to slower growth of plants in surface-mulch treatments, and the slower growth can be further compounded by low temperatures. The increased length can also decrease tissue density and make the seedlings more susceptible to damage. These seedlings are mechanically fragile, increasing the likelihood of collapse of the plant and possible death.

Plants with longer hypocotyls have smaller root systems, less leaf area, and less leaf and root biomass. As a result, plants that allocate more resources to the hypocotyls at the expense of leaves and roots have lower relative growth rates.

Table 1–4. Effect of sowing depth on plant establishment (plants/m^2). Trial sown at Mingenew in Western Australia at 18 cm spacing.

SOWING DATE	SOWING DEPTH (CM)			
	2.0	4.5	5.0	7.5
	NO. OF PLANTS/M^2			
28 April	45.1	26.8	31.5	6.8
22 May	120.6	118.1	102.4	12.1

Source: Walton (1998)

- Plant establishment was reduced at both sowing dates with deeper sowing.
- The April sowing was subject to drier soil than the May sowing, hence the lower establishment numbers.
- Sowing deeper than 5 cm gave unacceptable establishment rates.

Sowing time

Recommended sowing times for canola in NSW are shown in Table 1–5. Up-to-date tables are published each year in the *Winter crop variety sowing guide*. Sowing a variety outside of the sowing window increases the risk of frost damage or of high temperatures at flowering and grain fill.

Table 1–5. Recommended sowing times for canola in New South Wales.
Source: *Winter crop variety sowing guide*, DPI (2011)

WEEK	APRIL				MAY				JUNE			
REGION	1	2	3	4	1	2	3	4	1	2	3	4
Northern – West												
Northern – East												
Central – West												
Central – East												
Southern – West												
Southern – East												
Southern – Irrigation												

Best sowing time

Earlier or later than desirable, possible yield reduction.
Earlier – too vegetative, lodging, disease and/or frost risk.
Later – spring moisture and heat stress.

Too late for good yields, unless favourable spring.

Nutrition

Table 1–6 shows the amounts of nutrients removed per tonne of canola.

Nitrogen (N)

Nitrogen is an essential nutrient for plant growth and development. It affects final yield and oil content. However, canola is extremely sensitive to nitrogen placed with the seed – more so than wheat, barley and chickpeas. Placing nitrogen fertiliser at high rates with the seed can reduce germination, plant establishment and root growth.

The reduction in germination percentage is due to an increase in soil solution osmotic potential and a decrease in water uptake by seeds. There may also be toxicity effects.

The reduction in emergence percentages is greater in drier and colder soils. Rainfall soon after sowing reduces the effect of the fertiliser by leaching some of it away from the germinating seed.

Higher rates of nitrogen in the form of ammonia reduce root growth and cause stunting. The roots are also thicker and may have a brown, scorched appearance near the tip. Root length is also affected; this may cause problems with uptake of other nutrients.

The amount of nitrogen that can be placed directly with the seed at planting varies with rainfall and with row spacing. DPI recommends a maximum of 20 kg N/ha in higher rainfall areas and 10 kg N/ha in lower rainfall areas, or when planting on wider row spacings. Incitec Pivot has developed recommendations based on the type of sowing (direct drill or broadcast) and the row spacing (Table 1–7).

Phosphorus (P)

Phosphorus is an essential nutrient required at germination and in early seedling growth to promote rapid early root growth.

Phosphorus is the most limiting plant nutrient in southern and central NSW canola-growing soils.

Phosphorus needs to be applied close to the seed at planting. It is not a mobile nutrient like nitrogen, so the roots need to be in direct contact with it in the soil.

The main phosphorus forms taken up by roots from the soil solution are primary and secondary phosphate ions (H_2PO_{4-} and HPO_4^{-2}).

Table 1–6. Nutrients removed per tonne of canola.

	MINERAL ELEMENT CONTENT (KG)						MINERAL ELEMENT CONTENT (G)				
	N	P	S	K	CA	MG	CU	ZN	MO	MN	FE
Biomass at flowering	28.2	3.2	7.4	29.6	11.4	2.6	5.1	23.4	–	47.2	–
Grain	34.3	5.3	5.8	8.5	3.8	3.1	3.7	35.7	40.0	31.5	55.0
Stubble	5.6	2.8	5.7	9.2	6.8	1.8	3.7	9.7	–	25.5	–

Source: adapted from Brennan (2006), *Irrigated Cropping Forum* (2007), Price (2006), Santonoceto et al. (2002)

Table 1–7. Amounts of nitrogen that can be sown with canola seed, as determined by calculations of seed bed utilisation.

	25 MM SEED SPREAD, E.G. DISCS, KNIFE POINT ROW SPACING (CM)			50 MM SEED SPREAD ROW SPACING (CM)		
SOIL TEXTURE	15 CM	22.5 CM	30 CM	15 CM	22.5 CM	30 CM
SEED BED UTILISATION	17%	11%	8%	33%	22%	17%
Light (sandy loam)	10 kg N/ha	5 kg N/ha	0 kg N/ha	20 kg N/ha	15 kg N/ha	10 kg N/ha
Medium–Heavy (loam to clay)	15 kg N/ha	10 kg N/ha	5 kg N/ha	30 kg N/ha	20 kg N/ha	15 kg N/ha

Source: Jim Laycock, Incitec Pivot, adapted from 'Fertiliser management in direct seeding systems'. *Better Crops* 81(2), 1997.

Seedbed utilisation

Seedbed utilisation is a term that describes the volume of soil through which fertiliser is mixed. Generally, the higher the seedbed utilisation, the higher the rate of fertiliser that can be applied with the seed.

$$\text{Seedbed utilisation (\%)} = \frac{\text{width of seed row} \times 100}{\text{row spacing}}$$

Phosphorus levels in the plant. Canola seedlings take up phosphorus rapidly during early growth, but not as rapidly as nitrogen. The phosphorus level remains fairly high in the leaves (0.3% to 0.4%) until late flowering, when significant translocation occurs into developing pods and seeds.

By maturity, 75% to 80% of the phosphorus in above-ground dry matter is in the seed.

Canola seed contains 0.7% to 0.8% phosphorus – approximately double that of cereal grains. Canola stems and pods at harvest contain only 0.1% to 0.2% phosphorus.

Phosphorus requirements. The seed can supply enough phosphorus to support growth for about 7 days. After that, the plant needs to take phosphorus up from the soil.

Although canola requires large amounts of phosphorus for growth, maximum responses are often attained at lower rates of phosphorus than for wheat or barley. This is because canola, a non-mycorrhizal plant, is very effective in absorbing phosphorus from the soil and from fertiliser. Canola releases organic acids near the root tip. These acids increase the availability of soil phosphorus.

Phosphorus placement. Canola is more sensitive than cereal grains to damage from fertiliser salts placed with the seed. Levels of phosphorus greater than 15 to 20 kg/ha applied in the seed row have reduced crop emergence severely. Damage increases with decreasing soil water content.

Banding phosphorus near the seed can be as effective as placement within the seed row in terms of availability to the plant and will avoid damage to germination. In one study, application of up to 40 kg P/ha placed beside the row did not reduce emergence but increased yield.

Phosphorus deficiency. Deficiency symptoms can appear by the second week of growth, since canola seedlings are able to obtain sufficient phosphorus from seed reserves for the first week of growth.

Phosphorus-deficient leaves may be dark green, bluish green or purplish (from an accumulation of anthocyanin pigments).

Canola plants suffering from phosphorus deficiency can experience slow leaf expansion and have smaller and fewer leaves. Mildly deficient plants may look normal but are small.

Root growth is less affected than shoot growth by phosphorus deficiency. A severe deficiency restricts root development, but not as dramatically as stem and leaf growth.

Phosphorus deficiency affects the maturity and development of reproductive tissue. Even a mild phosphorus deficiency can result in maturity delays of several days. In addition to a flowering delay, a phosphorus deficiency can reduce the number of flowers and seeds per pod. Also, a phosphorus deficiency can cause leaves to die and drop early, contributing to the overall yield loss.

Sulfur (S)

Sulfur is required in relatively large amounts. Canola requires approximately 10 kg S per tonne of seed.

Sulfur deficiency symptoms may occur as early as 21 days after sowing. Symptoms commonly include reduced plant growth and leaf thickening. The leaves may be pale green, with pale yellowish mottling between the veins and cupping of younger leaves. Older leaves can show purple pigmentation on the undersides; the leaf margins can roll inwards, giving the leaf a tubular appearance.

The sulfur level in canola plants is highest in the early seedling stage, when young leaves make up most of the dry matter. As plants develop, the overall sulfur level declines, but not as dramatically as with nitrogen.

Zinc (Zn)

Canola requires zinc on alkaline soils. Applying zinc as a seed treatment is insufficient because of both the small seed size of canola and the low sowing rate. Zinc is therefore applied with fertiliser, or later in the season as a foliar spray.

Plant population

Plant population, which is determined by sowing rate, germination percentage and establishment percentage, is an important determinant of biomass at flowering and therefore yield.

Crops with low plant densities tend to yield poorly. Low-density crops can compensate with increased pod and seed production per plant, but they are more vulnerable to disease, pests, weed competition and environmental stress.

The current recommendation for NSW is a target plant population range of 30 to 60 plants/m^2, depending on the region (see DPI's *Winter crop variety sowing guide*). This is generally achieved with a sowing rate of 2 to 3 kg/ha.

Increasing the sowing rate increases competition between plants, creating thinner main stems and fewer, less productive branches.

Reducing the sowing rate creates plants with thicker main stems and more branches, delays leaf area development, reduces biomass at flowering and ultimately reduces yield.

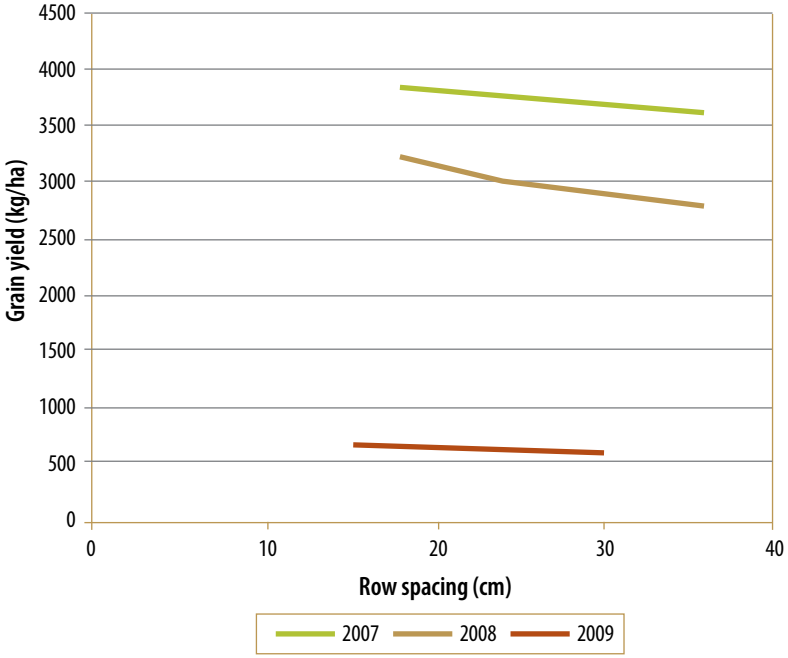
Row spacing

Canola has traditionally been sown at 15 cm row spacing, but the adoption of stubble retention and no-till farming systems has resulted in a trend to wider row spacing and the possibility of inter-row sowing using GPS guidance systems.

Recent experiments in southern NSW have shown that widening row spacing in canola does appear to reduce yield when the row space is increased to 35 cm (Figure 1–11).

Stubble cover

Sowing canola into heavy stubble can suppress canola emergence and establishment and reduce subsequent crop performance. Generally, there is little effect on canola emergence and crop establishment if less than 3 or 4 t/ha of stubble is present.



- There is some evidence that both canola and mustard branch more at lower plant populations.
- Even in dry years, wider rows appear to yield less.

Figure 1–11. Effect of row spacing on the grain yield of canola at Cowra in 2007, 2008 and 2009. Source: Jan Edwards, unpublished data

The effect of stubble is mainly physical rather than biochemical (i.e. it is not related to stubble toxicity). The effect is greatest if the stubble is lying directly over the sown row. The physical impact of stubble can be avoided if the stubble is moved away from directly above the emerging seedlings.

Stubble can reduce growth by causing:

- delayed plant and leaf emergence
- reduced shoot and root growth
- investment of energy in the hypocotyl at the expense of the roots and leaves
- increased incidence of seedling diseases.

Ultimately this gives fewer plants per unit area and reduces individual plant size, therefore reducing leaf area and biomass.

Stubble forces the plants to grow longer hypocotyls. It also delays leaf appearance, forcing the plant to rely for longer on its seed reserves.

Radiation effects

Stubble dramatically reduces the amount and spectral quality of light reaching the plants. The overall amount of usable light – particularly the amount of blue light and the red to far-red ratio – is reduced by stubble. This in turn reduces dry mass accumulation.

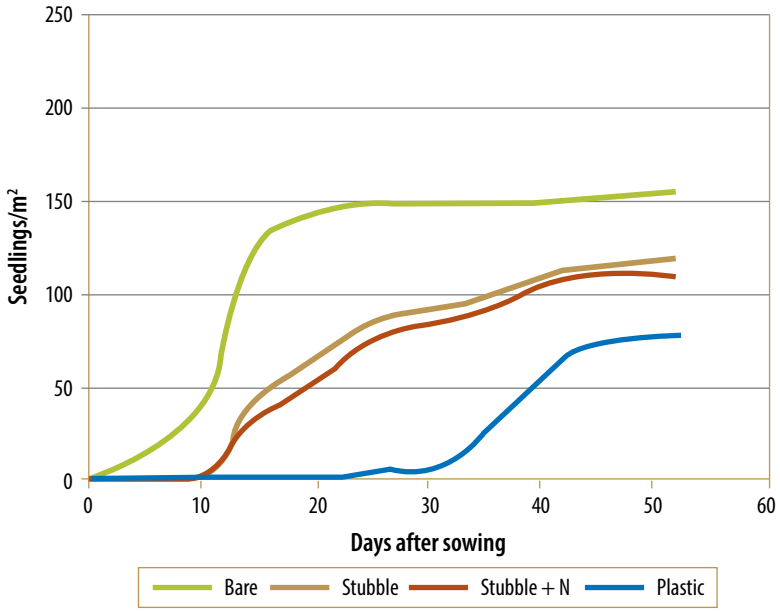
A reduction in the red to far-red ratio of light causes hypocotyl elongation. This effectively weakens the canola seedling. Hypocotyl elongation is influenced more by a decrease in the quality of the light spectrum than by shading.

In general, low light intensities reduce the photosynthetic rates of emerging seedlings and therefore their rates of growth and emergence above the surface-mulch layer (Figure 1–12).

Temperature effects

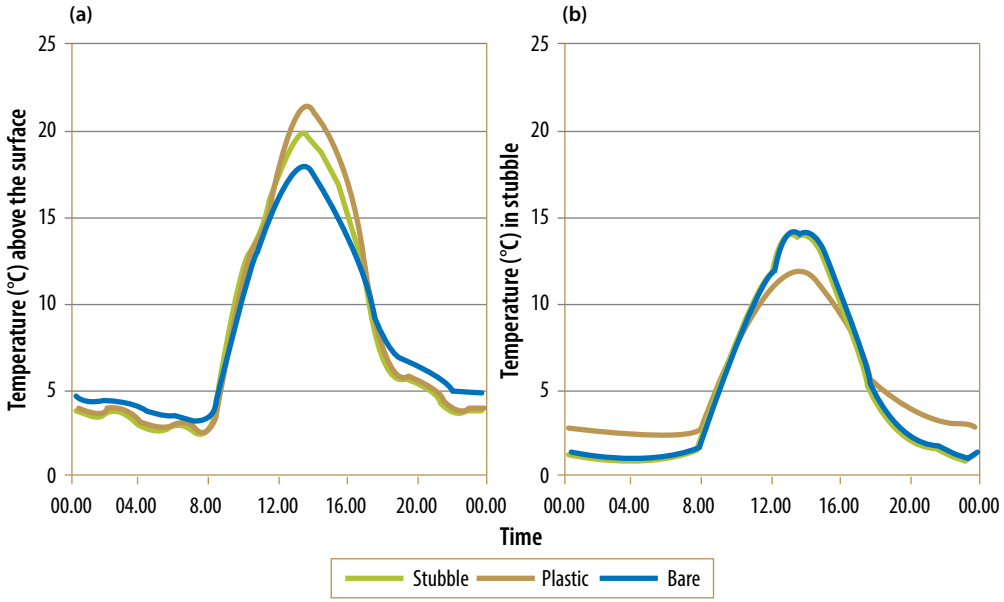
Stubble creates a greater diurnal variation in temperature at the soil surface than does bare ground (Figure 1–13). The growing point of canola seedlings is above the ground and is therefore exposed to this temperature variation.

Low temperatures may result in seedling death through freezing injury, and this causes lower seedling densities.



- Stubble reduces the rate of seedling emergence and the final number of seedlings per square metre.
- Wheat stubble at 5 t/ha reduced the rate of emergence of canola by 25%, plant establishment by 33%, vegetative biomass by 46% and yield by 26%.

Figure 1–12. Effect of 5 t/ha wheat stubble on emergence of canola. Source: Modified from Bruce et al. (2006a,b,c)



- There is a greater diurnal variation in temperature at the surface of stubble (5 t/ha wheat stubble) than of bare soil. (The stubble was raked off the plots immediately before sowing.)
- Stubble changes the temperature of the soil surface: the temperature is higher than that of bare soil.

Figure 1–13. Average air temperature measured (a) on the surface and (b) in the stubble. Source: Bruce et al. (2006a, b, c)

References and further reading

Boyd and Van Acker 2004, Imbibition response of green foxtail, canola, wild mustard seeds to different osmotic potential. *Canadian Journal of Botany,* 82(6), 801–806.

Brennan R 2006, *Nutrients in canola: maintaining the balance in your rotation.* DAFWA Farmnote 203/December.

Bruce SE, Kirkegaard JA, Pratley JE, Howe GN 2005, Impacts of retained wheat stubble on canola in southern NSW. *Australian Journal of Experimental Agriculture* 45, 421–433.

Bruce SE, Kirkegaard JA, Pratley J, Howe G 2006a, Growth suppression of canola through wheat stubble. I. Separating physical and biochemical causes in the field. *Plant and Soil* 281, 203–218.

Bruce SE, Ryan MH, Kirkegaard, Pratley J 2006b, Improving the performance of canola in retained wheat stubble. *Australian Journal of Agricultural Research* 57, 1203–1212.

Bruce SE, Ryan MH, Hely S, Kirkegaard JA, Pratley J 2006c, Growth suppression of canola through wheat stubble. II. Investigating impacts of hypocotyl elongation using simulated stubble. *Plant and Soil* 281, 219–231

Canola Council of Canada 2003, *Canola Growers Manual.* Canola Council of Canada, Alberta. www.canola-council.org/canola_growers_manual.aspx

Canola Council of Canada 2009, *Canola Growers Manual.* Canola Council of Canada, Alberta. www.canolacouncil.org/contents3.aspx

Clune TS, Copeland L 1999, Effects of aluminium on canola roots. *Plant and Soil* 216, 27–33.

Gunasekera CP, Martin LD, Siddique KHM, Walton GH 2006, Genotype by environment interactions of Indian mustard (*Brassica juncea* L.) and canola (*B. napus* L.) in Mediterranean-type environments:1. Crop growth and seed yield. *European Journal of Agronomy* 25, 13–21

I&I NSW 2011, *Winter crop variety sowing guide.* Industry & Investment NSW, Orange. www.dpi.nsw.gov.au/agriculture/field/field-crops/winter-cereals/winter-crop-variety-sowing-guide

Irrigated Cropping Forum 2007, *Oilseed News* 37, November. Irrigated Cropping Forum, Horsham Vic.

Nyborg M, Hennig AMF 1969, Field experiments with different placements

of fertilizers for barley, flax and rapeseed. *Canadian Journal of Soil Science* 49, 79–88

Nykiforuk CL, Johnson-Flanagan AM 1999, Storage reserve mobilisation during low temperature germination and early seedling growth in *Brassica napus*. *Plant Physiology Biochemistry* 37(12), 939–947

Price G (ed) 2006, *Australian Soil Fertility Manual*, 3rd edn, Fertiliser Industry Federation of Australia, CSIRO Publishing.

Reithmuller G, Alam R, Hamilton G, Hawksley J 2002, Large canola seed is best, particularly for deep sowing. *GRDC Update*, WA

Reuss R, Cassells J 2003, The effect of storage conditions on the quality of Australian canola (rapeseed), *Brassica napus* L. In *Advances in Stored Product Protection: Proceedings of the 8th International Working Conference on Stored Product Protection,* York, England, 2002. Credland PF, Armitage DM, Bell CH, Cogan PM, Highley E (eds). CAB International, Wallingford, Oxon, UK. pp 498–503

Santonoceto C, Hocking PJ, Braschkat J, Randall PJ 2002, Mineral nutrient uptake and removal by canola, Indian mustard, and Linola in two contrasting environments, and implications for carbon cycle effects on soil acidification. *Australian Journal of Agricultural Research* 53, 459–470.

Scott BJ, Fleming MR, Conyers MK, Chan KY, Knight PG 2003, Lime improves emergence of canola on an acidic hard setting soil. *Australian Journal of Experimental Agriculture* 43, 155–161

The Seed Biology Place 2009, Seed structure and anatomy. www.seedbiology.de/structure.asp

Walton G 1998, Don't sow canola too deep. *Highlights of Oilseeds Research and Development in Western Australia*. Grains Research and Development Corporation, ACT

IN THE PADDOCK

The following are some examples of activities that can be done in the paddock to illustrate the stages of germination and emergence that have just been discussed. These are practical exercises to help farmers assess the progress of their crop at these stages.

Seed size

Aim: to determine the 1000-seed weight of a seed lot.

The 1000-seed weight of canola is typically 3 to 6 g. To see whether your seed falls within this range:

1. Count out 200 seeds from each seed lot to be planted
2. Discard seeds that will be removed by cleaning/grading.
3. Weigh 200 seeds on scales accurate to 0.1 g.
4. Multiply the result by 5 to calculate the 1000-seed weight.
5. Repeat 5 times.

COUNT	1000-SEED WEIGHT (G)	
	SEED LOT 1	SEED LOT 2
1		
2		
3		
4		
5		
Average		

Calculating your germination percentage

Aim: to calculate the germination percentage of a seed lot.

An acceptable germination percentage is 85%. To compare your germination percentage:

1. Collect 50 seeds from each lot to be planted.
2. Lay four sheets of paper towel on top of each other and moisten (do not drench).
3. Place 50 seeds on the paper towels about 10 mm apart.
4. Roll up, sandwiching the seeds between the moist paper towels.
5. Soak a hand towel in water and ring out, then wrap it around the rolled-up paper towel and loosely secure with the rubber bands.
6. Place in a plastic bag, seal and place in a warm place (such as the kitchen bench near a window), and leave for 5 to 7 days.
7. Unwrap and count the number of seeds that have not germinated.
8. Do your calculation as follows:

   Germination % = [(number of seeds tested – number of seeds that did not germinate) ÷ 50] × 100.
9. Repeat five times.

IN THE PADDOCK

COUNT	GERMINATION %	
	SEED LOT 1	SEED LOT 2
1		
2		
3		
4		
5		
Average		

Calculating sowing rates

Aim: to calculate a sowing rate based on a target plant density.

1. Decide on a target plant density (see DPI's *Winter crop variety sowing guide*).
2. Calculate the 1000-seed weight.
3. Use the following formula to calculate sowing rates:

$$\text{Sowing rate (kg/ha)} = \frac{\text{(target density} \times \text{1000-seed weight in grams} \div 100)}{\text{(establishment \%} \times \text{germination \%)}}$$

COUNT	EXAMPLE 1	EXAMPLE 2	PADDOCK 1	PADDOCK 2
Target density	45	45		
1000-seed weight (g)	3	6		
Establishment	80% = 0.8	80% = 0.8		
Germination	90% = 0.9	90% = 0.9		
Sowing rate	1.9	3.8		

Sowing depth

Aim: to measure hypocotyl length and sowing depth.

1. Carefully dig up 10 plants along a row, including the seed and roots, in two paddocks.
2. Measure the depth from the seed to the soil surface – where the hypocotyl ends and the green stem begins.
3. Record the hypocotyl length of the 10 plants from each paddock in the table below; calculate the average hypocotyl length, and therefore the sowing depth.

IN THE PADDOCK

COUNT	HYPOCOTYL LENGTH (CM)	
	PADDOCK 1	PADDOCK 2
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		
Average		

Sowing implements and seed placement

Aim: to compare the sowing depths of different sowing implements.

1. Compare the sowing depths in paddocks that have been sown with different types of planters or in different soil types.
2. Examine the depths of different sowing rows. Was the machine level?
3. Discuss soil throw in zero till and implications for emergence.
4. Look at the deep furrows. Compare the growth of seedlings in deep furrows compared with areas with no furrows.
5. Look for differences in the length of the hypocotyl during cotyledon or leaf development.
6. Record the hypocotyl lengths of 10 seedlings from each paddock and compare the results.
7. Compare the emergence dates.
8. Press wheels: Examine the paddock for signs of crusting, poor seedling emergence and the effects of soil-type changes.
9. Stubble: Are there any differences in emergence visible? Did the sowing implement drag stubble? Can you observe any shading effects?

Establishment

Aim: to determine the uniformity of seedling establishment.

1. Check seed placement and moisture and stubble conditions at sowing.
2. Observe the evenness of seedling size.
3. Record the hypocotyl lengths of 10 seedlings from each paddock and compare the results.
4. Compare the emergence dates. Is there more than one emergence time/event?
   Hint: Compare between hybrid and conventional varieties and between sowing times.

IN THE PADDOCK

Plant population

Aim: to determine the establishment percentage and plant population in a paddock.

1. Count the plants along 1.0 m of row (either side of a 0.5-m row or along one row) at 10 random locations within the paddock.
2. Add the 10 counts together and divide by 10 to give the average number of plants/metre of row.
3. Multiply the average plant count by the row (tyne or disc) spacing factor:

   17.5 cm = 5.71 30 cm = 3.33

   20 cm = 5.00 33 cm = 3.03

   22.5 cm = 4.44 36 cm = 2.77

   25 cm = 4.00 40 cm = 2.50

   27.5 cm = 3.36 50 cm = 2.00

4. Repeat in a different paddock and record the results for both.

COUNT	PLANT POPULATION	
	PADDOCK 1	PADDOCK 2
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		
Total		
Average per m row		
Plants/m^2		

2. Vegetative growth

by Jan Edwards and Kathi Hertel

Chapter Snapshot

Introduction

Chapter 2 explains vegetative growth and the factors that affect the growth of the plant. The aim of this chapter is to help you make effective management decisions by understanding how a canola plant grows and how growth can be affected by different management strategies. The effectiveness of post-emergence inputs, such as herbicides, fertilisers and water, differs with stage of growth. Proper timing of application, based on the growth stage of the crop, can improve the efficiency of the input and prevent crop injury and economic loss.

Learning Outcomes

At the end of this chapter, you will be able to:

- identify the difference between growth and development
- describe how the root system grows
- explain photosynthesis, respiration and transpiration
- understand how dry matter or biomass is accumulated
- describe the role of plant nutrition during vegetative growth
- locate the main stem, branches, pods and seeds of a canola plant
- assess the dry matter or biomass of a crop.

Vegetative growth

It is important to distinguish between growth and development. **Growth** is an increase in size through the accumulation of dry matter, first as sugars, then as structural and storage materials in the leaves, stems and fruits. **Development** is the progress of a crop through the stages of its life cycle.

The vegetative stage – the period from sowing to first flower – can range from 40 to 60 days, depending on the date of sowing and the growing conditions.

Root growth

The function of the root system is to absorb nutrients and water for plant growth. Healthy roots, unrestricted by soil constraints or disease, are essential to maximise yield. Roots also synthesise growth regulators or plant hormones.

Canola has a taproot. The taproot, in the absence of major obstructions, grows vertically through the soil profile. Smaller roots grow in all directions, allowing the young canola plant to access water and nutrients. The canola root system can grow to over 100 cm deep by the time of stem elongation.

The roots are covered in microscopic hairs that greatly increase the surface area of the roots. When root hairs are factored in, the root system of a plant may have a total surface area of hundreds of square metres.

Canola has longer root hairs than many other crops. Under conditions of low phosphorus, canola root hairs will increase in both length and density, giving the crop better access to soil phosphorus.

Rate of growth

Root growth is the result of cell division and enlargement at the tip of the root. Root development is relatively constant, averaging nearly 2 cm a day as long as there is good soil moisture. Canola roots can elongate by 8 mm a day.

Early-sown crops are likely to have more extensive root systems than later sown crops, simply because of the extra time available for growth.

Roots do not grow in search of water or nutrients. They intercept water and nutrients that they come into contact with in the soil pore space.

Root density

About 25% to 50% of canola roots are found in the top 10 to 20 cm of soil. Canola root density is generally 65% of wheat root density in the top 60 cm of the soil profile. Less than 1% of canola roots are found below 1 m.

The surface area of the root system is very important for water and nutrient uptake from the soil.

Root depth

At emergence, the roots will be 3 to 5 cm deep. After establishment, the root system grows rapidly. The taproot extends down into the soil, the secondary roots extend laterally, and the taproot begins laying down reserves.

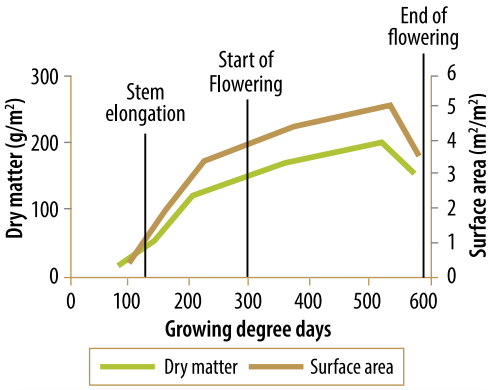
The depth of the root system will depend on factors such as soil type, moisture content, temperature, salinity and soil physical structure.

Early in the season the crop extracts water to a depth of 40 cm. At flowering the roots reach 80% of their maximum depth (see Figure 2–1). By maturity the roots have grown to a depth of around 110 cm. In some soils, the roots can extract water and nutrients from depths of up to 1.5 m.

Root radius increases rapidly initially and then plateaus just before stem elongation.

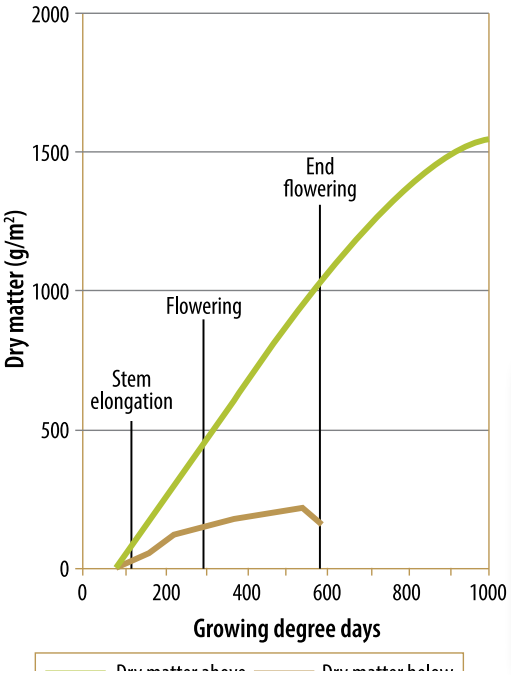
The maximum amounts of non-structural carbohydrates and nitrogen are stored at the end of the rosette stage. These root reserves are used mainly for regrowth of leaves in spring.

Below-ground dry matter levels start to decrease at the end of the flowering stage, well before the decline in above-ground dry matter levels (Figure 2–2).



- Roots increase in both dry matter and surface area until just before the end of flowering.

Figure 2–1. Change in root dry matter and surface area over the whole profile (0–100 cm) from emergence to the end of flowering. Source: modified from Kjellstrom (1991)



- Below-ground dry matter levels start to decrease at the end of the flowering stage, well before the decline in above-ground dry matter levels.
- Maximum root dry matter, surface area and length are reached late in the flowering phase.
- At the end of flowering, the shoot to root ratio is 8:1.

Figure 2–2. Comparison of dry matter accumulation above ground (leaves and stems) and below ground (roots). Source: modified from Kjellstrom (1991)

Factors limiting root penetration

Factors limiting root penetration through the soil include waterlogging, soil dryness, soil compaction, weed competition for moisture and nutrients, cool soil temperatures and the presence of a salt layer. Canola plant roots will not grow into waterlogged, dry or compacted soil.

As the roots grow, they use oxygen and release carbon dioxide. Restriction of soil aeration by excess water or soil compaction results in low oxygen and high carbon dioxide levels and, eventually, root death.

Moist topsoil with dry subsoil during the early stage of plant growth results in a shallow root system. Roots penetrate dry soil only slightly beyond the available moisture supplies.

Insects (such as false wireworms) and diseases (rhizoctonia or pythium) can damage the roots and restrict the uptake of water and nutrients.

Canola and mustard need to have deep roots to allow them to bring water to the leaves and to maintain water status.

Because brassica crops cannot grow roots through extended zones of dry soil, if the crop is limited to using water near the soil surface there is an increased risk of lower yields unless there are timely rains in spring.

Root glucosinolates

Modern varieties of canola have been selected for their low seed glucosinolate content. However, canola roots still contain glucosinolates. The glucosinolates are metabolised rapidly to sustain plant growth. They appear to be a form of storage for nitrogen, carbon, and especially sulfur.

Some glucosinolates suppress soil-borne pathogens; this is thought to be partly why canola is useful as a cereal break crop.

Canola lines with high root glucosinolate levels are less likely to host the cereal root lesion nematode *Pratylenchus neglectus*. When canola green manure crops are ploughed into the soil, isothiocyanates are released from the roots as the glucosinolates degrade. These isothiocyanates are toxic to a range of soil-borne fungal pathogens. This toxic action is referred to as **biofumigation**.

Biofumigation
Death of soil-borne fungal pathogens due to the isothiocyanates released from the roots as the glucosinolates degrade.

The main root glucosinolate is 2-phenylethyl glucosinolate. The total glucosinolate concentration varies markedly among varieties (from 5 to 20 µmol/g). Most of the glucosinolates that release isothiocyanates are contained in top and lateral roots greater than 2 mm in diameter.

The highest concentrations of glucosinolates in spring canola occur at flowering, which coincides with the highest root concentrations.

Leaf growth – Growth stages 1.0–2.0

Four to eight days after emergence the seedling develops its first true leaves (Figure 2–3). The growing point (apical meristem) is situated at the junction of the cotyledons. It initiates the leaf primordia (i.e. the leaves in their earliest stage of development).

Leaf emergence is initially rapid. Production and growth of the earliest leaves govern the rate at which solar radiation is maximised.

The plant continues to produce leaves (Figure 2–4) at a rate of about one every 7 to 10 days. There is no definite number of leaves produced by a canola plant. A canola plant under good growing conditions normally produces 10 to 15 leaves on the main stem, depending on variety, time of sowing, nutrition, plant population and available soil moisture.

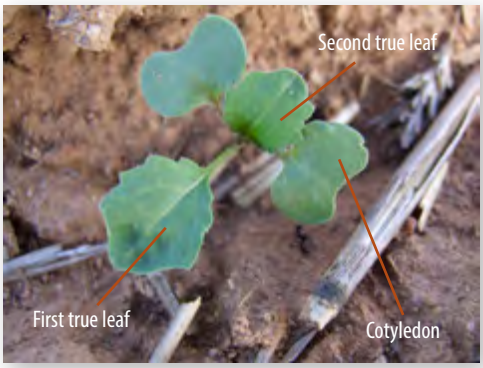


Figure 2–3. First and second true leaves (left). Photo: Jan Edwards

Figure 2–4. A canola leaf. Photo: Jan Edwards

A mature, unstressed leaf will have an area of between 200 and 250 cm^2.

Leaf formation is continuous between germination and the onset of reproductive development. Once reproductive development begins, the apex begins to initiate the first flower buds of the terminal **raceme**. Senescent (old) leaf material begins to drop from around the stage when the flower buds are visible.

Rosette formation – Growth stage 2.0

The plant quickly establishes a rosette (Figure 2–5), with older leaves at the base increasing in size and smaller, younger leaves developing in the centre. *Brassica napus* plants develop rosettes of up to six waxy, blue-green leaves. During this rosette growth stage the stem length remains essentially unchanged, although stem thickness increases. A single leaf is attached to the stem at each node.

Figure 2–5. Canola flower bud arising from leaf axil. Photo: Kathi Hertel, DPI

Stem branching

At, or just before stem elongation, branch initiation begins. Branches arise from buds in the axils of the leaves (mainly the upper leaves). Branches develop one to four leaves and a flower bud. These branches are important, as they allow compensation for poor stand establishment, disease or hail.

Raceme
An unbranched flowerhead, with flower buds connected by short stalks called *pedicels*. This creates a bunch of flowers at the end of the stem.

Stem elongation – Growth stages 2.0–2.9

Stem elongation (Figure 2–6) overlaps with leaf development and normally occurs just after eight or nine leaves have emerged.

Stages of stem extension are defined by how many detectable internodes are found on the stem. A well-grown plant produces 15 to 20 internodes, spaced at about 5 to 10 mm. A leaf is attached to the stem at each node.

During stem elongation, flower and branch initiation begins. Maximum stem length is reached at full flowering. Stem diameter and height are influenced by sowing date, moisture, variety, soil fertility and plant population. Modern Australian canola varieties range in height from 70 to 100 cm to 150 to 170 cm. As the stems elongate, the roots continue to grow deeper.

Figure 2–6. Canola (variety Skipton) at stem elongation/early flowering. Photo: Kathi Hertel, DPI

The mechanisms that regulate the duration of stem elongation are independent of those involved in flower initiation. The stem elongation phase occurs between flower initiation and flowering. It is not sensitive to vernalisation (see Chapter 3) but is highly responsive to variations in temperature and photoperiod.

The length of the stem elongation phase (from initiation to flowering) is more important to final yield than the length of the stage before flower initiation.

Stem growth stops at the end of flowering. Stem weight is directly related to total crop dry weight (including the weight of dead leaves).

Factors affecting vegetative growth

Before establishment, plant growth is fuelled by energy reserves stored in the endosperm of the seed. The canola seed can support seedling development for about 1 week (depending on seed size), after which time the plant depends on the soil for nutrients and the sun for photosynthesis.

Once the first two cotyledons have unfolded, growth relies on energy produced by the plant through photosynthesis. Plant functions such as **evapotranspiration**, photosynthesis, water and nutrient absorption and transport, enzyme activity, and other biological and chemical activities are regulated by temperature. Other factors such as moisture, light (day length), nutrition and variety also play a role, but they generally have less influence.

Evapotranspiration
The combined loss of moisture from the soil and the plant.

Photosynthesis

$6CO_2$	+	$6H_2O$	→	$C_6H_{12}O_6$	+	$6O_2$
carbon dioxide	+	water	→	carbohydrate	+	oxygen

Figure 2–7. Photosynthesis.

Respiration

$C_6H_{12}O_6$	+	$6O_2$	→	$6CO_2$	+	$6H_2O$	+	energy
carbohydrate	+	oxygen	→	carbon dioxide	+	water	+	energy

Figure 2–8. Respiration.

Photosynthesis and respiration

Plants get their energy to grow from sunlight captured by the leaves. **Photosynthesis** (Figure 2–7) is the process by which plants use this energy to convert carbon dioxide to sugars. The sugars are converted to cell-wall-forming substances that make up the leaves, stems, roots and other plant parts. Excess sugars are stored as water-soluble carbohydrates.

When energy cannot be obtained from sunlight, such as at night, the plant draws on its reserves of starch and sucrose. This process is called **respiration** (Figure 2–8).

The rate at which the plant grows is closely linked to the amount of sunlight being captured by the leaves. Temperature influences the rate at which the chemical reactions of photosynthesis occur. The ideal temperature range for canola plants is from 12°C to 30°C.

Optimum temperature for growth is 25°C. Above this temperature the growth rate slows, with heat stress occurring between about 25°C and 35°C, although this research is still inconclusive.

The effects are confounded by variety, moisture and light.

Factors that affect the rate of photosynthesis, and ultimately crop yield, include:

- available water
- fertiliser practices
- variety selection
- sowing rate
- sowing depth
- weeds
- pest control.

For example, crops deficient in nitrogen have yellow, stunted leaves. Yellow leaves have less chlorophyll, and small leaves simply catch less light.

Transpiration

Plant water use, or the evaporation of water from within a leaf, is called **transpiration**. During transpiration, water evaporated from the wet cell walls within the leaf moves into the leaf airspaces and out through the leaf **stomata**.

Transpiration and photosynthesis are related. Leaf stomata need to be open to allow water vapour to move out and to allow carbon dioxide for photosynthesis to move in. When plants are transpiring, water is drawn up from the soil all the way to the leaves to replace the water lost to the atmosphere.

Transpiration is slower in cool conditions with high humidity, and faster in windy, hot conditions with low humidity. Hot conditions are unfavourable when combined with low soil moisture, because as the soil moisture decreases the stomata close to conserve moisture, and photosynthetic productivity is lost.

Dry matter accumulation and leaf area

The number of leaves produced before flower initiation has a major influence on leaf area and hence the photosynthetic potential of the crop.

The growth rate of the crop is closely related to the amount of solar radiation captured by the leaves.

Leaf area index

The leaf area index (LAI) is a measure of the upper surface area of leaves per unit of ground surface. A leaf area index of 4 means that there is 4 m^2 of leaf surface area per m^2 of ground surface. Canola plants usually develop a leaf area index of between 3 and 6.

A leaf area index of about 4 is required for the crop canopy to intercept about 90% of the incoming solar radiation. The larger the leaf area the crop can expose to the sun, the more dry matter the crop can produce per day. The more dry matter, the higher the potential yield (Figure 2–9).

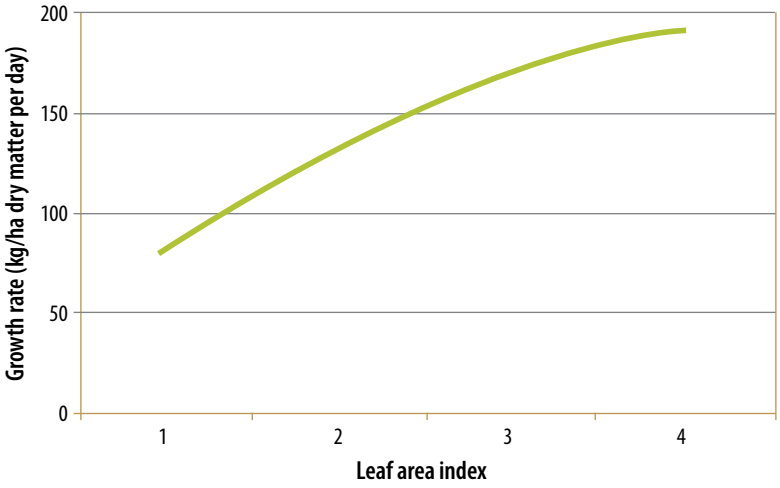
Canola leaves influence seed yield at early growth stages by influencing the development of the plant's overall sink capacity, pod set and early seed development.

Rapid leaf development also encourages root growth, reduces soil moisture evaporation and shades out weeds.

There is a positive correlation between seed yield and maximum leaf area index if there is good moisture. The leaf area index of a crop also determines water use. The higher the leaf area index, the more water is used during the vegetative growth stage. In dryland crops, a large area of leaf in early vegetative growth may use water needed for flowering and seed fill.

Transpiration
Evaporation of water from within the leaf.

Stomata
Tiny pores found on the leaves that allow gas exchange.



- As leaf area increases so too does plant growth rate.
- At a leaf area index of 1 the growth rate is about 75 kg dry matter/ha.
- At a leaf area index of 4 the growth rate is nearly 200 kg dry matter/ha.

Figure 2–9. Relationship between leaf area index and crop growth rate. Source: Canola Council of Canada (2003)

A balance needs to be reached between having a large leaf area to maximise photosynthesis and carbohydrate production and maintaining enough soil moisture for seed fill.

Early sowing and ideal growing conditions in the early development stage can produce a large plant with a high yield potential, but if the spring becomes dry the plant can fail to fill all the pods and can 'hay off', with low seed yields (Figure 2–10).

Leaves initially are the most important photosynthetic plant structures for fixing food for plant growth.

The leaf area index of canola starts to decrease shortly after first flower (Growth stage 4.2) (Figure 2–11). At full flower (Growth stage 4.8), the stems become the major photosynthetic structures, although the leaves are still important. At the beginning of ripening (Growth stage 6.4) the pod walls and stems account for the majority of photosynthesis (Figure 2–12).

Plants in low-population crops (e.g. 20 plants/m^2) have a higher leaf area index than do plants in high-population crops (e.g. 126 plants/m^2), as plants compete with each other for light, moisture and nutrients.

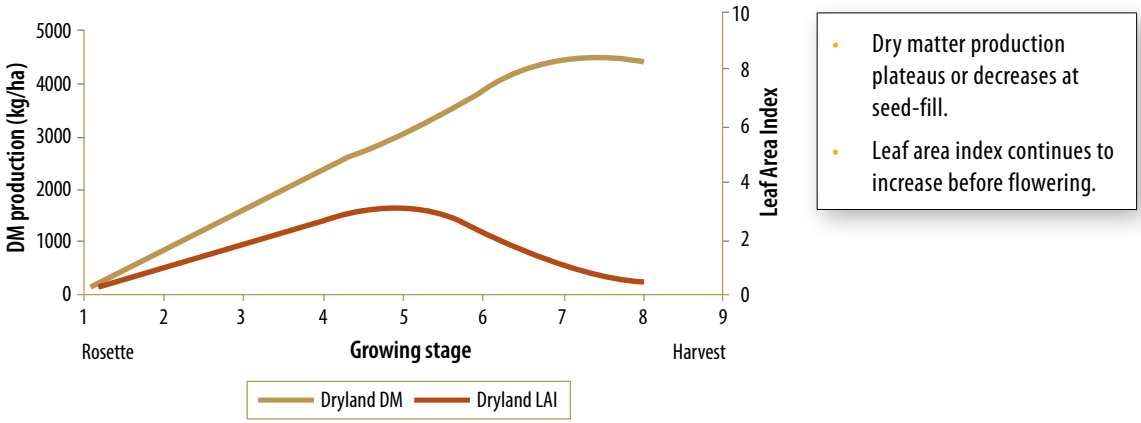


Figure 2–10. Dry matter (DM) production and leaf area index (LAI). Source: Modified from Canola Council of Canada (2003)

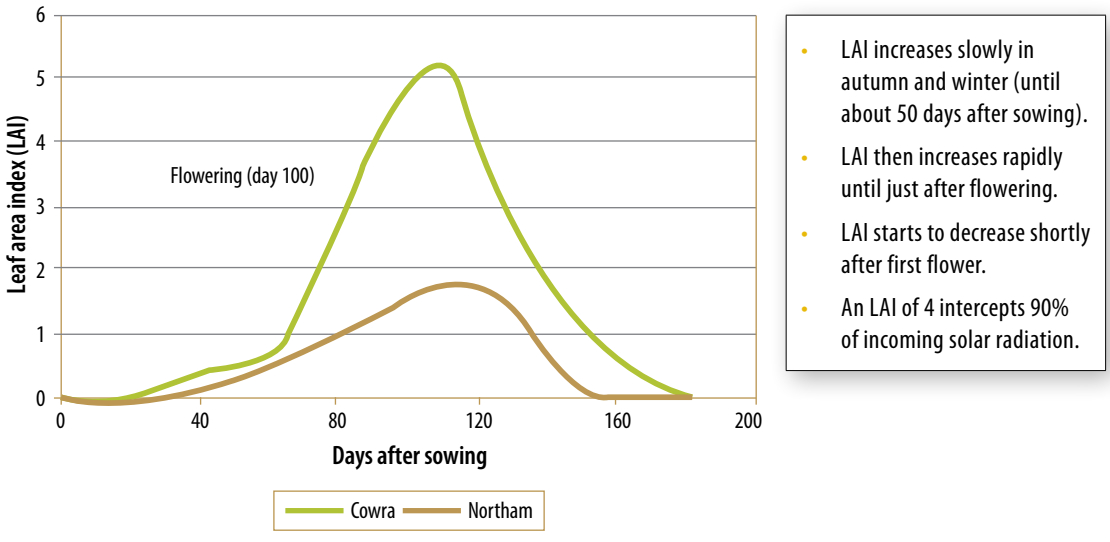


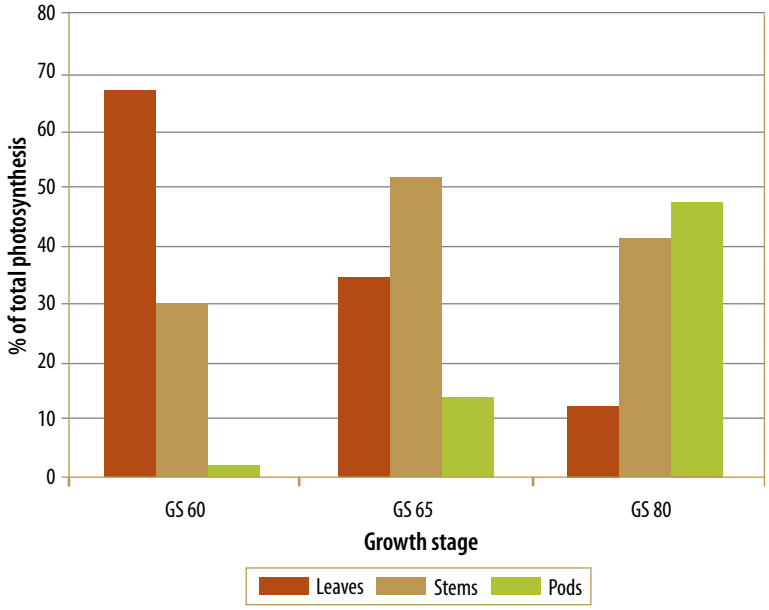
Figure 2–11. Leaf area indexes of crops at Northam, WA and Cowra, NSW. Source: Salisbury et al. (1999)

Dry matter accumulation

Dry matter accumulation in shoots is initially slow, but once canopy closure is reached a period of rapid growth ensues. Growth peaks and then slows as the leaves senesce during pod filling (Figure 2–13).

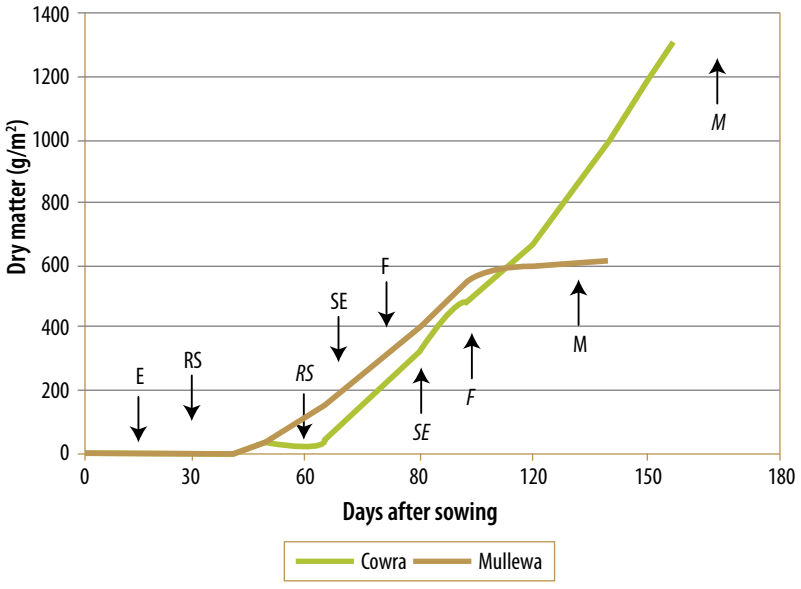
In the absence of major stress, a crop can accumulate about 1.2 g of dry matter in the shoots for each megajoule of solar radiation intercepted by the canopy.

At flowering, about 60% of the shoot dry matter is in the leaves and 40% in the stems. During pod filling, significant amounts of dry matter can be mobilised from the leaves (before being shed), stem and pod walls and used to fill the seed. The green pod walls and stems will photosynthesise actively but not as efficiently as the leaves, as their stomatal density is not as high.



- Before flowering, the leaves are contributing the most (65%) to photosynthesis.
- By mid flowering it is the stem that does most of the photosynthesising, and by grain fill it is the pods.

Figure 2–12. Photosynthetic contributions by canola plant structures. Source: Canola Council of Canada (2003)



- Dry matter accumulation is initially slow, but once canopy closure is reached there is a period of rapid growth.
- Dry matter accumulation slows as the leaves die during pod filling.
- The crop accumulates about 1.2 g of dry matter in the shoots for each megajoule of solar radiation intercepted by the canopy.
- At flowering about 60% of the dry matter is in the shoots and 40% in the roots.

Figure 2–13. Dry matter accumulation during canola crop growth at Cowra and Mullewa, Stages in roman type, Cowra. Stages in italics, Mullewa. E = seedling emergence, RS = rosette stage, SE = stem elongation commenced, F = flowering commenced, M = physiological maturity. Source: Modified from Salisbury et al. (1999)

Temperature

The optimum temperature range for leaf development of canola is between 13°C and 22°C. At higher temperatures growth is faster, so there is a shorter period of leaf development. Lower temperatures do not reduce yield in early growth, except when heavy frosts occur, but they slow the rate of development. As temperatures increase above 20°C in July and August, yields are reduced.

Frost

For frost injury there must be ice formation between or inside the cells. Water surrounding the plant cells will freeze at 0°C, but water inside the cells needs to be a few degrees cooler to freeze. The length of time for which the plant is exposed to cold is also an important factor. Plants can be cold hardened by repeated exposure over several days. They can survive –8°C to –12°C in Canada, but exposure to warm weather will reverse this hardening, making the plants susceptible to temperatures of –3°C to –4°C.

Moisture

Chloroplast
Organ in plant cells that conducts photosynthesis.

Water is essential for plant growth. Adequate soil moisture:

- promotes root growth
- promotes a large abundant leaf area
- helps plants retain their leaves longer
- lengthens the flowering period
- increases the number of branches per plant, number of flowers forming pods, seeds per pod, seed weight, and seed yield.

Moisture stress is more important during pod fill than at the vegetative stage. However, too much or too little water at any particular growth stage reduces yield potential. Factors that may limit yield include the:

- amount of summer soil-stored moisture
- rate and duration of rainfall during the growing season
- ability of the soil to absorb water, store it, and make it available for plants.

Modifying some of these factors can improve moisture availability and efficiency of water use.

When soil water and nutrients are abundant, the balance of root to stem and leaf growth typically shifts in favour of stem growth at the expense of roots. When water is limited, the opposite usually occurs. In moisture-stressed canola, roots account for about 25% of plant dry matter at stem elongation, compared with about 20% in unstressed plants.

Moisture stress during rosette formation and elongation

Canola has limited ability to withstand severe drought. To avoid dehydration the plant closes its stomata and rapidly sheds leaves.

Moisture stress during the early vegetative stages reduces the ability of stomata to conduct carbon dioxide and therefore slows down photosynthesis. This in turn reduces leaf area expansion and reduces dry matter production. It also limits root growth, which reduces nutrient uptake.

More severe water deficits inhibit photosynthesis as a result of cell and **chloroplast** shrinkage.

This is important in seasons with dry winters. It is also important in low rainfall areas where the period of crop growth is restricted at the start of the season by lack of rainfall and at the end of the season by water deficits and high temperatures.

Plants under early-season moisture stress will usually recover normal growth with subsequent rainfall or irrigation. Stressed plants are able to recover leaf area, form flowers, set pods and fill seeds when water becomes available, but with hastened development rates, early maturity and lower yields. The worst time for drought stress in canola is during stem elongation or flowering.

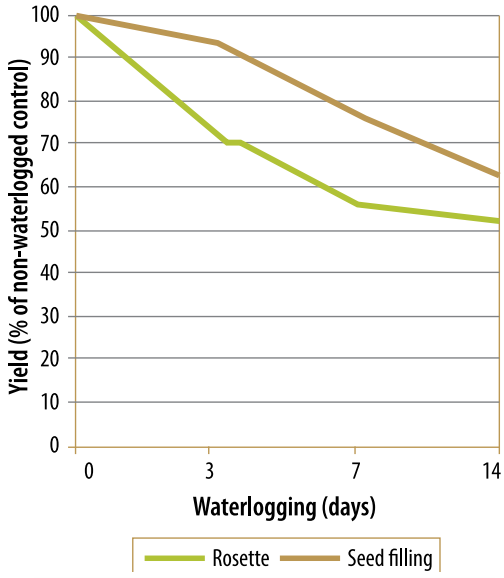
Long periods of drought will reduce yields more than frequent short periods of drought. The impact will be greatest on coarse-textured soils and shallow soils with low water-storage capacity.

Adequate soil moisture tends to lengthen the number of days to maturity by up to 10 days. Additional soil moisture will result in no further increase in yield and may cause yield reductions through poor soil aeration and/or increased lodging and diseases.

Waterlogging

Canola roots need a good mix of water and air in the soil. When the amount of water exceeds the soil's water-holding capacity, waterlogging may occur. Canola is quite susceptible to waterlogging and shows a yield reduction after only 3 days. Symptoms include older leaves turning purple and senescing (aging) more rapidly. In very poorly aerated soils plants can die.

The amount of yield loss depends on the growth stage at the time of waterlogging, the duration of waterlogging, and the temperature (Figure 2–14).



- The impact of waterlogging is greater if it occurs at the rosette stage rather than seed filling.
- The longer the period of waterlogging, the greater the impact.

Figure 2–14. Effect of waterlogging on yield. Source: Canola Council of Canada (2003)

Wet soils slow down, or prevent, gas exchange between the soil and atmosphere, causing an oxygen deficiency. High temperatures cause high respiration rates in roots and soil micro-organisms, so soil oxygen is consumed more quickly.

Soil texture also affects the time at which critical levels of soil oxygen are reached. This is due to the oxygen-carrying capacity of soils. Coarser textured soils can hold more oxygen, increasing the amount of time before oxygen levels are reduced to a critical point.

The other effects of waterlogging are to reduce root growth, plant growth, plant height, dry matter production and nutrient uptake.

During seed filling, waterlogging for more than 7 days decreases individual seed weight and oil content. High temperatures increase the detrimental effects of waterlogging on canola yield.

Nutrition

Nutrients need to be supplied within an optimum range from germination onwards to maximise plant growth.

Most nutrient uptake is through the root system. Intake of nutrients through the leaves is usually insignificant, except in the case of a few minor nutrients such as zinc, manganese and molybdenum.

Some nutrients, such as phosphorus and zinc, have low mobility in the soil solution. The roots need to grow through the soil in contact with the nutrients. This has implications for the placement of fertilisers in the soil.

Nutrients that are soluble, and therefore mobile, include nitrate, calcium, magnesium and sulfate.

Adequate soil moisture
Defined as maintaining 50% or more of the available soil moisture in the root zone.

Nitrogen

Nitrogen influences nearly all components of growth. During vegetative growth it is required for root growth, leaf growth and the production of chlorophyll.

Nitrogen increases canola leaf area index, leaf number per plant (Table 2–1) plant weight and growth rate. It therefore increases dry matter production (Figure 2–15). It also increases leaf duration, number of flowering branches, plant height, number of flowers, number and weight of pods and seed yield.

The majority of the nitrogen in green plant tissue is present in enzymes in the chloroplasts, where chlorophyll is located, but nitrogen is also part of many other critical plant components.

The relative nitrogen proportions in the plant change over time and with growth stage. The nitrogen proportioning closely resembles the dry matter partitioning.

The nitrogen level in canola plants is highest in the early seedling stage, when young leaves are the majority of the plant's dry matter. By maturity, canola straw contains just 0.5% to 1.5% nitrogen, whereas the seed contains from 3.4% to over 4% nitrogen.

Nitrogen deficiency symptoms. Healthy canola plants with adequate nitrogen have dark green leaves. Nitrogen is mobile within the plant and can be moved from older to younger leaves and pods. Therefore, nitrogen deficiency symptoms first show up in older leaves as pale green to yellow colouring, and sometimes purpling. These older leaves tend to die early, turn brown and drop off prematurely. Overall plant growth is slow, with short thin stems, small leaves, and few branches. The amount and time of flowering are also restricted, and pod numbers are low. In healthy canola, plant tissue tests of above-ground material at flowering should show more than 2.5% nitrogen.

Sulfur

Sulfur is a key component of two essential amino acids and is needed for protein synthesis. Synthesis of chlorophyll and of the volatile oils that accumulate as glucosinolates also requires sulfur.

Canola is more sensitive than cereals to sulfur deficiency and frequently responds to sulfur fertiliser. Canola requires about 1.5 kg sulfur to grow 100 kg seed, i.e. a 2 t/ha crop of canola requires 30 kg S/ha.

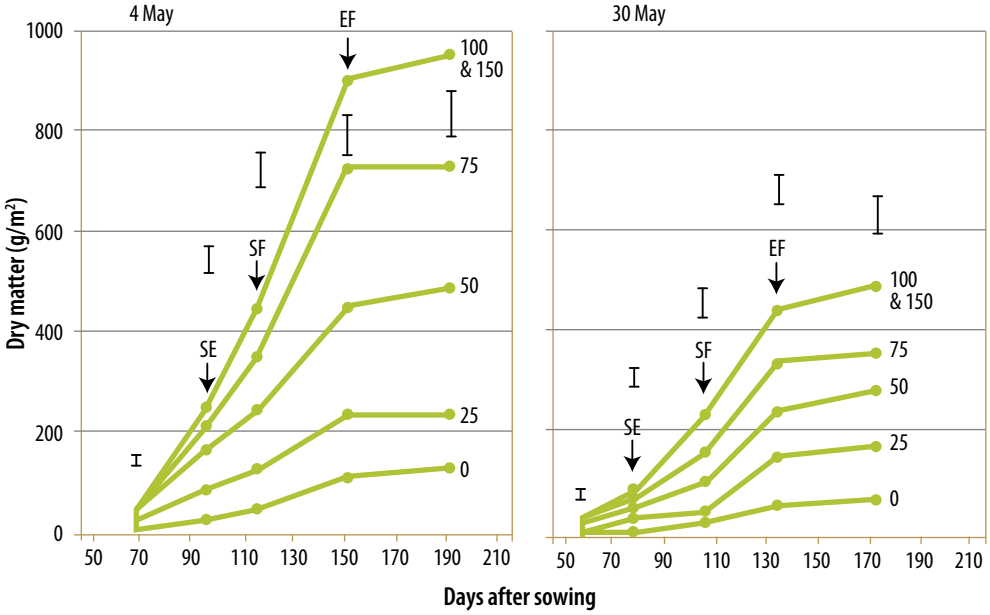
Nitrogen and sulfur requirements are closely related, because both elements are required for protein synthesis.

Table 2–1. Effects of sowing time and nitrogen fertiliser rate (kg N/ha) on leaf number per plant in canola at Cowra.

CROP AND SOWING DATE – COWRA	N FERTILISER RATE (KG/HA)					
	0	25	50	75	100	150
At rosette stage						
Canola 4 May	5.6	6.2	6.8	6.8	6.8	6.8
Canola 30 May	3.1	4.2	5.3	5.8	6.2	6.1
At stem elongation						
Canola 4 May	8.2	11.4	13.9	14.5	14.4	14.9
Canola 30 May	7.2	10.1	10.8	11.0	12.1	12.5
At start of flowering						
Canola 4 May	9.8	12.7	14.5	15.3	15.2	15.5
Canola 30 May	8.1	10.7	11.4	12.1	12.9	13.0

Source: Modified from Hocking et al. (1996)

- Leaf number per plant increases with nitrogen application.
- Leaf number is higher with earlier sowing.



- Increasing nitrogen application rates increased dry matter production up until the end of flowering.
- A later sowing date gave a lower dry matter production.

Figure 2–15. Effects of sowing time and nitrogen fertiliser on dry matter production by canola at Cowra. SE = stem elongation; SF = start of flowering; EF = end of flowering. Rates of N/ha are shown on the right of the graph lines. Sowing time is shown at the top of each graph. Source: Modified from Hocking et al. (1996)

Sulfur deficiency. Sulfur deficiency symptoms vary depending upon the severity and timing of the deficiency. In the vegetative stage, foliar symptoms show up under severe sulfur deficiency.

Symptoms of sulfur deficiency in canola crops are often not apparent until the period of rapid growth during stem elongation. Stem elongation coincides with the most rapid growth phase of the crop, occurring during late winter when soil temperatures are low.

Mild sulfur deficiency often does not result in noticeable symptoms, but it can still reduce yield. By stem elongation, sulfur deficiency begins to affect yield parameters such as branches per plant, fertile flowers per plant, seeds per pod and individual seed weight.

Since sulfur has low mobility within the plant, symptoms are observed more readily on the youngest leaves, which are greenish-yellow compared with the normal bluish-green of *B. napus*. The yellowing (chlorosis) starts from the leaf edges, and the tissue around the leaf veins remains green. Subsequently, the leaf edges and bottoms may turn purple.

By the bolting stage, new leaves of sulfur-deficient plants show chlorosis, purpling and spoon-like leaf cupping. The purpling is caused by enhanced pigment (anthocyanin) synthesis due to sugar accumulation resulting from a reduction in amino acid and protein synthesis. The degree of leaf cupping is highly dependent on the timing of the sulfur deficiency. There is significant cupping when sulfur deficiency occurs before half of the leaf weight is attained.

Critical sulfur values are: at the rosette stage (5 or 6 leaf) 0.58%; visible bud stage: 0.40%; and stem elongation stage, 0.24%.

Potassium

Potassium plays a major role in the physiological processes of plants. Therefore, it is required in large amounts for adequate crop production. Most soils are generally well supplied with K.

A healthy, high-yielding crop contains potassium at between 150 and 300 kg/ha. The concentration in the seed is low relative to that in other plant parts. Thus, only low levels are removed in the seed at harvest. The rate of uptake increases as the concentration in the soil solution increases.

Potassium deficiency in canola reduces growth, but not as severely as nitrogen and phosphorus deficiency. Deficiency symptoms tend to begin in the older leaves, with marginal and interveinal chlorosis of successive leaves accompanied by withering and development of a dark green appearance. Chlorosis occurs first in the middle leaves and later in the older leaves. Small necrotic spots develop, and plants may wilt. In severe cases, leaves die but may remain attached to the stem. The plant may be more susceptible to lodging, which shows little response to applied potassium.

Potassium has no significant effect on oil or protein content. It is less mobile in the soil than nitrogen, but somewhat more mobile than phosphorus. Banding will increase application efficiency.

Canola requires approximately 20% more applied potassium than does wheat to produce 90% of the maximum shoot or seed yield, but canola is more efficient than wheat at taking up applied potassium.

Calcium

Calcium is an important component of the membrane of the cell wall. It is also critical in membrane stability and cell integrity and so influences the uptake of other ions. It is required for cell elongation and cell division and is involved as an activator of a few enzymes. Uptake of calcium is mainly passive.

Because of the immobility of calcium in the plant, symptoms of deficiency occur first in the younger tissue and include chlorosis of the leaf margins and production of stunted, discoloured leaves.

Micronutrients

Micronutrient deficiencies are not common, but on specific soils they can severely restrict crop yields. Symptoms are often hard to diagnose and need tissue testing to confirm. With any of these nutrients there is a fine line between adequate nutrition and toxicity.

Manganese. Manganese is required for many metabolic processes, including chlorophyll production. It is relatively immobile in the plant.

Manganese toxicity is more common than deficiency. It appears as yellowing of the leaf margins, rolling of the leaf edges, and crinkling of the leaf surface. It appears in older leaves first. The leaves become lighter in colour and turn brown as they die.

After extended dry periods, readily available manganese can build up to toxic levels in the soil. Four to six weeks after the autumn break, the readily available manganese is converted to unavailable forms. Young plants are susceptible to manganese toxicity, so early sown crops are at greater risk.

Zinc. Zinc is involved in the enzyme systems of plants. It is also required for protein synthesis, hormones and carbohydrate metabolism. Membrane stability also relies on zinc.

Symptoms of deficiency range from brown spots on cotyledons, purpling on new emerging leaves, interveinal chlorosis and cupping of leaves, and reduction in leaf size. A deficiency can also inhibit stem elongation.

Zinc deficiency is more common on high-pH soils and those with a high carbonate content.

Boron. Boron plays a role in root elongation and pollen tube growth. Its link to root elongation is not well understood.

Deficiency symptoms are more likely to be seen during flowering (see Chapter 4), as demand is higher during the reproductive stage than in the vegetative stage. Symptoms of deficiency (low flower and pod numbers) are more likely during periods of low moisture or where liming has reduced the availability of boron.

Grazing

In mixed farming systems, it is common practice to graze cereal crops to fill the winter feed gap. Winter cereal varieties can be grazed without a major grain yield penalty, provided they are not grazed after their vernalisation (cold requirement) period has been met. The same principles can be used for canola.

Canola is a high-quality forage (See Chapter 3).

March-sown canola can produce between 2.5 and 4.0 t/ha of biomass in 8 weeks of growth. By comparison, early-May-sown canola can produce up to 1 t/ha of forage (Table 2–2). Winter and long-season varieties are more suitable for grazing. Spring cultivars produce comparable biomass, but the early onset of flowering make them less suited to early sowing.

Grazing before stem elongation will remove between 60% and 70% of the biomass. Biomass removal may reduce crop water use early in the season because there is less dry matter to transpire. The water conserved in the profile may then be available for use during seed fill.

Provided grazing occurs before stem elongation, there is little impact on the time of flowering and yield [see Chapters 3 (delay in flowering) and 4]. Grazing after stem elongation will remove the stem and its flower buds. The plant then has to regrow the main stem.

The other impact of grazing is to make plants more susceptible to blackleg (see Chapter 4).

Table 2–2. Biomass (t/ha) of canola varieties before grazing and 1 week after grazing. Field trial at Ginninderra Experiment Station, Canberra, 2004–2005.

VARIETY	UNGRAZED		GRAZED		BIOMASS GRAZED	
	2004	2005	2004	2005	2004	2005
Spring type – Hyola 60	4.6	–	2.1	–	2.5	–
Winter type 1	4.7	8.11	2.1	3.80	2.6	4.30
Winter type 2	2.8	6.56	2.5	3.51	0.3	3.05
Winter type 3	–	6.42	–	3.08	–	3.36

Source: Kirkegaard (2007)

- Canola produced between 0.3 and 4.3 t/ha grazeable biomass after 8 weeks of growth.
- The amount depended on rainfall.

References and further reading

Brennan RF, Bollard MDA 2007, Comparing the potassium requirements of canola and wheat. *Australian Journal of Agricultural Research* 58, 359–366

Canola Council of Canada 2003. *Canola Growers Manual.* www.canola-council.org/chapter3.aspx

Hocking PJ, Pinkerton A, Good A 1996, Recovery of field-grown canola from sulphur deficiency. *Australian Journal of Experimental Agriculture* 36, 79–85

I&I NSW 2011, *Winter crop variety sowing guide.* Industry & Investment NSW, Orange. Available at www.dpi.nsw.gov.au/agriculture/field/field-crops/winter-cereals/winter-crop-variety-sowing-guide

Kahlon R, Pang E, Salisbury P, Kadkol G, Taylor P 2001, Synthetic *Brassica napus* lines for blackleg resistance and manganese toxicity. In *Proceedings of the 12th Australian Research Assembly on Brassicas*, 2–5 October, Geelong

Kirkegaard JA 2007, *Evaluating the Potential for Dual-purpose Canola in the Mixed Farming System of Southern Australia.* Report to Grains Research and Development Corporation on Project CSP00085. CSIRO, Canberra

Kirkegaard JA, Sarwar M 1999, Glucosinolate profiles of Australian canola (*Brassica napus annua* L.) and Indian mustard (*Brassica juncea* L.) cultivars: implications for biofumigation. *Australian Journal of Agricultural Research* 50, 315–324

Kirkegaard JA, Rebetzke GJ, Richards RA 2001, Inheritance of root glucosinolate content in in canola. *Australian Journal of Agricultural Research* 52, 745–753

Kjellstrom CJ 1991, Growth and distribution of the root system in *Brassica napus*. In McGregor DI (ed), *Proceedings of the 8th International Rapeseed Congress, Rapeseed in a Changing World,* 9–11 July, Saskatoon, Saskatchewan, Canada, pp. 722–726.

Robertson MJ, Kirkegaard JA 2003, Crop modelling for the Australian canola industry: a review. In *Proceedings of the 13th Biennial Australian Research Assembly on Brassicas,* 8–12 September 2003, Tamworth, NSW. NSW Agriculture, Orange

Robertson MJ, Watkinson AR, Kirkegaard JA, Holland JF, Potter TD, Burton W, Walton GH, Moot DJ, Wratten N, Farre I, Asseng S 2002, Environmental and genotypic control of time to flowering in canola and Indian mustard. *Australian Journal of Agricultural Research* 53, 793–809

Rumberger A, Marschner P 2004, 2-Phenylethylisothiocyanate concentration and bacterial community composition in the rhizosphere of field grown canola. *Functional Plant Biology* 31, 623–631

Salisbury PA, Green AG 1991, Developmental responses in spring canola cultivars. In *Rapeseed in a Changing World* (D I McGregor, ed). *Proceedings of the GCIRC Rapeseed Congress 1991*, July 9–11, Saskatoon, Saskatchewan Canada. Vol. 6 of 6, pp. 1769–1774

Salisbury PA, Potter TD, McDonald G, Green AG 1999, Canola in Australia: the First 30 Years. In *Proceedings of the 10th International Rapeseed Congress,* Canberra.

IN THE PADDOCK

The following are some examples of activities that can be done in the paddock to illustrate the stages of growth and development that have just been discussed. These are practical exercises to help farmers assess the progress of their crop at this stage.

Examining the root system

Aim: to check the root system of the crop for signs of disease.

1. Carefully dig up 10 plants.
2. Wash out the roots.
3. Find the taproot and identify secondary roots.
4. Examine the roots. Check the root growth pattern. Is the taproot straight or bent? Are there signs of damage caused by a hardpan? Examine the secondary roots for signs of herbicide residues or mineral toxicities.
5. Do the roots look healthy? Are they white or are they discoloured? Is there any sign of disease (such as rhizoctonia or pythium) and/or insect damage (e.g. wireworm)?

Assessing plant growth stage

Aim: to accurately assess the current crop growth stage.

1. Dig up a plant
2. Identify the cotyledons (if present) and true leaves.
3. Count the number of true leaves present.
4. Compare with the decimal growth scale in the Introduction and record the growth stage
5. Examine leaves for signs of discolouration and presence of disease or insect damage.
6. Look closely for signs of buds development 6 to 8 weeks after sowing.
7. Assess percentage ground cover at budding stage. Make a circle with the forefinger and thumb of your hand. Position your hand 10 cm in front of your eye and look at the crop 2 m in front of you and make an estimate of the amount of covered ground.
8. Perform this procedure 10 times and record the results.
9. Repeat this exercise with different varieties and different sowing dates.

PLANT	GROWTH STAGE	
	PADDOCK 1	PADDOCK 2
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		

IN THE PADDOCK

Dry matter assessment

Aim: to determine what quantity is available to graze. Assess the crop when its root system is firmly anchored.

1. Using a quadrat, cut a known area to ground level. Perform this process at five locations.
2. Mix the sample and weigh it. This is the wet weight.
3. Weigh out a subsample of 150 g and very carefully dry it in a microwave. Place a cup of cold water in the microwave with the sample to prevent the material from burning. Replace the water when it begins to boil.
4. Calculate the dry matter percentage by using the following formula:

$$\text{Dry matter \%} = \frac{\text{weight of dry sample (g)} \times 100}{\text{weight of wet sample (g)}}$$

5. To calculate herbage mass, you need to convert your quadrat cut to kg DM/ha.

$$\text{Herbage mass (kg DM/ha)} = \frac{\text{average wet weight} \times \text{dry matter \%} \times \text{conversion}}{100}$$

6. The quadrat conversion factor is:
   – for a 30 cm × 50 cm quadrat: 0.15 m^2 or 1/66 666 of a hectare, so multiply by 67 to get kg DM/ha
   – for a 50 cm × 50 cm quadrat: 0.25 m^2 or 1/40 000 of a hectare, so multiply by 40 to get kg DM/ha.

CUT	WET WEIGHT (g)	DRY MATTER %	HERBAGE MASS (kg DM/ha
1			
2			
3			
4			
5			
Average			

Nitrogen and sulfur topdressing

Aim: to assess growth stage, soil moisture, crop colour and deficiency symptoms to determine yield potential and whether a topdressing response is likely.

1. Identify the growth stage of the crop. Topdressing decisions should be made by the bud formation stage.
2. Measure the depth of wet soil. Every 10 cm of wet soil will hold approximately 18 mm for a heavy clay; medium clay 15 mm; clay loam 10 mm; sandy loam 8 mm.
3. Is sufficient soil moisture available for the crop to reach maturity? Is the crop moisture-stressed?
4. Examine the crop for signs of yellowing, paleness and/or stunting.
5. Examine individual plants for symptoms of nitrogen and/or sulfur deficiency. Distinguish between deficiency symptoms and frost injury.
6. Check the short-term rainfall outlook and seasonal prognosis. At least 5 mm of rainfall within 3 days of application is required to move fertiliser into the soil. Preferably apply fertiliser to moist soil to minimise volatilisation losses.

Monitoring for pests, diseases and injury

1. At regular intervals through the growth cycle, walk a transect across the paddock.
2. Stop at five locations and check the plants for signs of insect damage or disease.
3. Look for nutrient deficiency symptoms and herbicide effects.

3. Reproductive development

by Jan Edwards and Kathi Hertel

Chapter Snapshot

Introduction

The reproductive phase of the canola plant continues the process of determining final yield. Vegetative growth prepared the plant to form the main stem, flower head and yield components. Management and environmental conditions during vegetative growth, and environmental stresses during the reproductive phase, determine the maximum yield that can be set by the plant within its genetic potential.

The aim of this chapter is to provide knowledge of the stages of reproductive development to help farmers manage the canola plant during this phase, minimising the effects of various stresses and maximise yield.

Learning Outcomes

At the end of this chapter, you will be able to:

- recognise the change from vegetative growth to reproductive development
- identify stem elongation, floret formation and flowering
- describe the effects of frost, heat and moisture on reproductive development
- determine the flowering window for a variety, taking into account location
- assess the growth stage of a crop, particularly for the purposes of nitrogen topdressing, herbicide application and removal of grazing stock.

Reproductive development

The reproductive phase begins when the shoot stops forming leaves and begins forming flower buds. This is a complex phase, with a number of developments happening at the same time (see Figure v in the *Introduction* chapter).

The two key stages in the reproductive development of canola are floral initiation and flowering.

Floral initiation

Once the vernalisation or photoperiod requirement has been met, the plant stops producing leaves and begins to develop flowers. This period is called floral initiation.

The axil of one of the upper leaves begins to swell. These swellings will become the flowers of the main stem. Once the main stem flower buds are formed, the buds develop sequentially in the auxiliary branches.

The main controls of floral initiation are:

- the presence of a minimum number of leaf initials before initiation can take place. This varies among varieties.
- the basic temperature response or leaf production rate
- vernalisation response
- daylength response.

Floret
One of the small individual flowers within a dense cluster of flowers.

If the vernalisation or daylength response is not fully satisfied, initiation of flowering will be delayed past the minimum number of leaf initials. This will delay flowering until either the winter has passed or the days are lengthening.

There is a large variation in the number of nodes that appear before initiation. This is the major reason for the variation in time to flowering. The number of days per node is 6 to 10 at low temperatures and 4 to 5 at high temperatures.

Temperature has a large impact on leaf development, so the number of nodes on a plant is a better indication of a plant's 'physiological' age than calendar days.

Flower bud development – Growth stages 3.0–3.9

Flower buds start to develop at flower initiation, just before or during winter or early in spring. At first the flower buds remain enclosed in the leaves during early stem elongation.

Green bud stage – Growth stage 3.3

The green bud stage (Figure 3–1) is when the buds become visible to the naked eye. As the stem elongates, the flowers emerge above the leaves but are not free from them. At this stage the stamens and petals are developing. Pollen is also beginning to form.

Yellow bud stage – Growth stage 3.7

The stem continues to elongate until the flowers are free from the leaves and the lowest flower buds assume a flattened shape. The lower buds are the first to become yellow (yellow bud stage), and progressively more buds become yellow as the stem grows. Flower bud development is completed during the yellow bud stage (Figure 3–2).

Flowering – Growth stages 4.0–4.9

The flowering (or 'anthesis') period (Figures 3–3 to 3–6) begins with the opening of the first floret on the main stem. Flowering starts at the base of the branch and continues upwards to the tip of the flower head (the raceme).

The basic floral structure consists of a large number of small flowers or **florets**. These continue to develop, and by the time the flowers open the maximum number of potential ovules within each pod will have been determined.

Figure 3–1. Lower bud development: green bud stage.
Photo: Julie White, DPI

Figure 3–2. Flower bud development: yellow bud stage.
Photo: Lowan Turton, DPI

The first floret to be initiated flowers first, and it ultimately forms the first pod at the base of the terminal flower head.

The length of the flowering period varies with variety and soil moisture. Generally, flowers open over 26 to 30 days. About 75% of the pods retained to maturity are formed from flowers that opened within 11 days of flowering (Figure 3–7). Most of these flowers are from the terminal raceme and on the basal and middle regions of nodes 1, 2 and 3. Although many hundreds of flowers are usually produced by an individual plant, relatively few reach maturity.

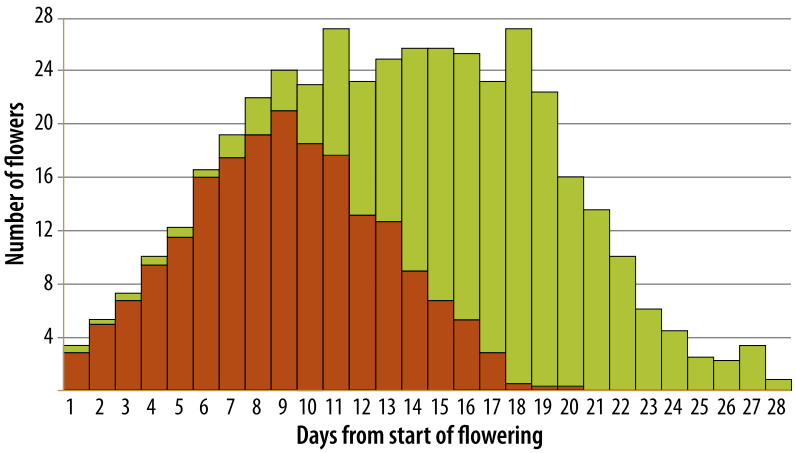
At flowering, the yield potential is set by the balance between vegetative growth and the potential number of flowers, pods and seeds.

Figure 3–3. Start of flowering (growth stage 4.1).
Photo: Julie White, DPI

Figure 3–5. Flowering. Photo: Julie White, DPI

Figure 3–4. Start of flowering (growth stage 4.2).
Photo: Lowan Turton, DPI

Figure 3–6. Flowering (growth stage 4.8).
Photo: Jan Edwards



- Most of the pods that were retained developed from the first flowers to open.
- About 75% of the pods present at maturity developed from flowers that opened in the first 11 days (from the opening of the first flower).

Figure 3–7. Total number of flowers that opened at daily intervals over the whole plant and the number of these that formed pods retained to maturity (shaded bars). Source: Tayo and Morgan (1975).

Environmental conditions during the 2 to 3 week period after flowering are critical to yield.

Flowering stops because of competition for resources with the pods. Active seed growth, beginning on the lower pods, has a strong demand for assimilates. This demand appears to switch off the supply of assimilates to the apex of the plant about 16 days after flowering (depending on temperature). This causes the end of flowering.

Extending the flowering period extends the period of competition for assimilates between the growing stem, the newly formed flowers, and the developing seeds, resulting in an overall reduction in yield.

Pollination

Canola pollen can remain viable for 1 to 7 days, depending on the temperature and humidity. Under natural conditions pollen viability gradually decreases over 4 or 5 days. In Australia, canola crops flower in spring, as temperatures are increasing and the humidity declining. Under these conditions pollen viability can be reduced to 24 to 48 hours.

Canola plants are mainly self-pollinating, but cross-pollination does occur. Fertilisation of the ovules usually results from self-pollination, because each flower produces a large amount of pollen, which out-competes the pollen from adjacent flowers.

Outcrossing can occur between adjacent plants at levels of about 30%. Canola can cross-pollinate through physical contact between neighbouring plants, or it can be insect pollinated. The level of outcrossing depends on the presence of insect pollinators, the variety, and the weather conditions.

Canola pollen can become airborne and travel several kilometres downwind. Wind-borne pollen plays a minor role in long-distance pollination. The vast majority of pollen travels less than 10 m, and the amount of pollen decreases as the distance from the pollen source increases.

Canola pollen is heavy and sticky. The flowers also produce nectar with high concentrations of sugars and a colour and structure that are attractive to insects (particularly bees) (Figure 3–8).

Figure 3–8. Canola pollination. Photo: Jan Edwards

The germinability of pollen grains is close to 80% for flowers at flowering.

Pollen tube growth is optimal at 10°C to 25°C, but the amount of growth does not affect the germinability or viability of the pollen.

Each ovule needs to be fertilised by a pollen grain. Pollen grains produced by the anthers that surround the immature pod may land on the stigma (pollen receptor) of the same flower (self-pollination). Pollen from other flowers may also reach the receptive stigma via pollinating insects that are attracted to the flower. Nectar is produced in the nectaries at the outer base of the female reproductive organ.

Successful pollination of all the ovules within each pod is influenced by the potency and maturity of the pollen grains produced by its floret. Temperature changes at the time of maturation of the male and female reproductive organs can greatly reduce the chance of fertilisation.

Factors affecting reproductive development

Plant development is primarily altered by photoperiod and temperature, with a general shortening of phases as the day length and/or temperature increases. After vernalisation, development is hastened. These factors interact to determine the number of days between sowing, flowering and maturity.

Yield potential is determined by the time of flowering.

Competition

Because pod and then seed development begins immediately after flowering, a canola plant is at multiple growth stages—that is, the pods and seeds are at different stages of development. There is therefore competition for assimilates between flowers on the same raceme and between racemes on different branches. Earlier-developed pods have a competitive advantage over the later-formed ones.

Assimilate supply before flowering is a major determinant of the number of flowers and seed-bearing pods.

Limitation of assimilate supply is the main cause of cessation of flowering and seed abortion.

Stresses in the supply of assimilates around the time of flowering reduce the number of pods that develop and the capacity for compensatory growth when the supply returns to normal.

The supply of carbohydrates at or after flowering therefore regulates the yield of seeds and pods. Prolonged stress results in smaller pods and fewer, lighter seeds.

Moisture stress

Water supply is critical during flowering and early pod development, when the numbers of pods and seeds are being determined. These stages of growth usually coincide with increasing temperatures and decreasing soil water supply.

Moisture stress during the flowering or ripening stages results in large yield losses, especially if combined with high temperatures.

Moisture stress causes a dramatic decrease in leaf photosynthesis. Leaves wilt and die sooner, thus reducing branching, pods per plant, pod length, seed size and seeds per pod. Seed oil content drops and protein content increases. If moisture stress is severe, recently formed pods may abort.

Moisture stress may greatly slow or stop root growth, affecting soil water uptake. Some varieties are better able to adjust total seed and pod number and **harvest index** to reduce the impact on yield.

Osmotic adjustment

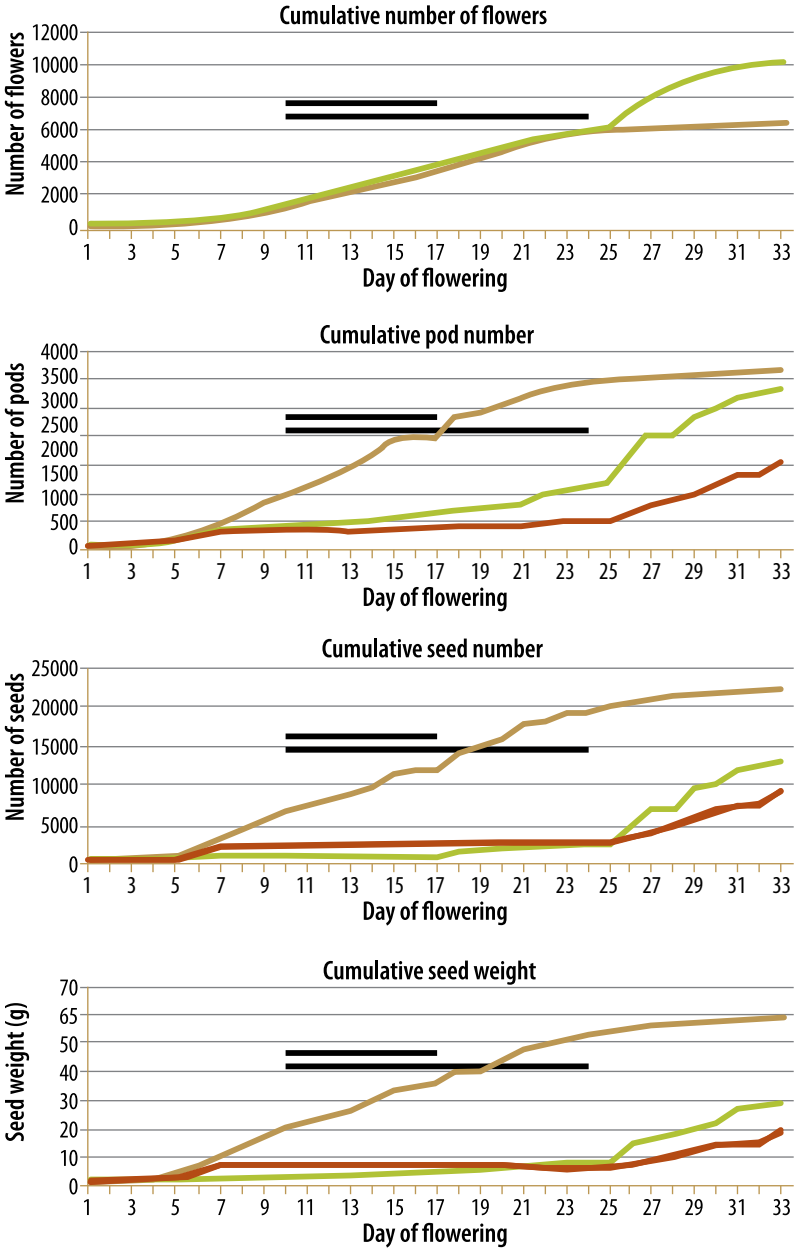
Plants have a number of mechanisms for adapting to drought. For example, the plant can accumulate solutes (substances dissolved in solution) to help maintain cell turgor (the force exerted outwards on a plant cell wall by the water contained in the cell). This osmotic adjustment allows the plants to function for a few days longer under drought stress.

Pollen tube
Tube that acts as a conduit to transport male gametes in the pollen grain from the stigma (pollen receptor) to the ovules at the base of the pistil (female reproductive organ).

Harvest index
The ratio of yield biomass (i.e. seed weight) to the total cumulative biomass (i.e. plant weight) at harvest.

The capacity of a plant to express osmotic adjustment is influenced by the stage of plant development. The cultivar Monty adjusts at the juvenile and elongation vegetative phases, but not at flowering or later. Monty also consistently showed much lower osmotic adjustment than Karoo at flowering.

Mustard shows osmotic adjustment in the juvenile, elongation and flowering stages. It is therefore better adapted to low-rainfall growing regions.



Temperature

Temperature can affect the reproductive development of canola in three ways: heat stress, cold/freezing, and effects on the rate of development (through thermal time).

Heat stress

High temperatures (above 25°C), particularly when accompanied by warm night temperatures, can:

- induce both male and female sterility
- reduce the length of time for which flowers are receptive to pollen
- reduce the length of the pollen release period
- reduce pollen viability.

Plants under moisture stress have decreased maximum temperature thresholds.

Brassica napus is the least able of the canolas to recover from severe stress at flowering.

In a growth chamber, pollen sterility can be induced by a temperature regime of 32°C/26°C day/night. *Brassica napus* plants grown throughout their life cycle at 27°C/17°C light/dark were found to be almost totally sterile (Figure 3–9).

- The greatest reduction in seed production occurred when plants were exposed to a 7-day heat-shock treatment of 35°C/15°C light/dark during early flowering.
- Half the control flowers developed pods, compared with between 6.7% and 14.6% for the heat treatments.
- Pod and seed abortion occurred in flowers fertilised up to 4 days before the heat stress, so developing fruits (i.e. pods and seeds) up to 4 days old were sensitive to heat stress.
- Reduced seed production continued for between 2 and 8 days after the removal of the heat stress.
- Decreased pollen viability persisted for up to 7 days after removal of heat stress.

Figure 3–9. Effect of heat stress treatment on the fertility of *B. napus* plants. ▬ = stress period; ▬ = control (16 hours of daylight at 23°C and 8 hours of darkness at 18°C); ▬ = 1 week of temperature stress (increase in daytime temperature from 23°C to 35°C over 6 hours, maintained at 35°C for 4 hours, then down to 23°C for the remaining 6 hours and a night-time temperature of 18°C for 8 hours); ▬ = same for 2 weeks. Source: Young et al. (2004)

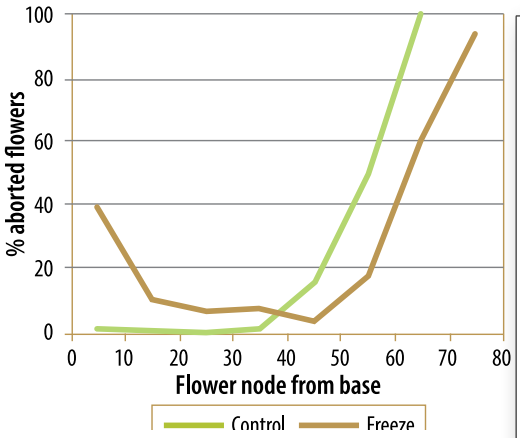
The effect of high temperature on seed set is greater when the pollen donor plants are heat stressed than when the receptor plants are heat stressed. The heat stress-induced changes in *B. napus* pollen development are irreversible, but in one study *B. juncea* pollen grains were still able to germinate after 4 or 24 hours at 45°C or 60°C.

Cold and freezing

Canola plants become acclimatised ('hardened') to cold during the vegetative phase. Once the plant has elongated and experienced warmer temperatures, it becomes less tolerant to low temperatures and more susceptible to frost.

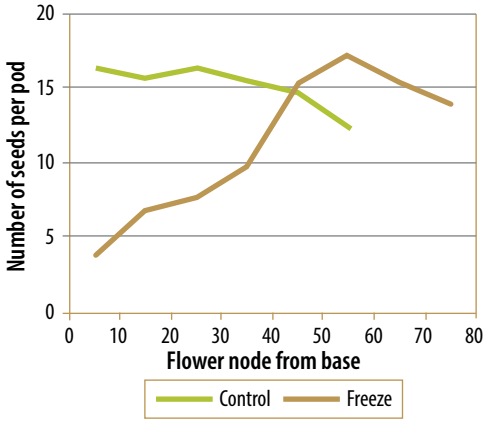
Cold temperatures, although they may not injure tissues, can slow down or alter processes such as pollination and fertilisation and development (Figures 3–10 to 3–12).

Low temperatures can also cause flower abortion, but because canola has a long flowering period, the plants can compensate. The first flowers to open, at the base of the raceme, are the most sensitive to frost.



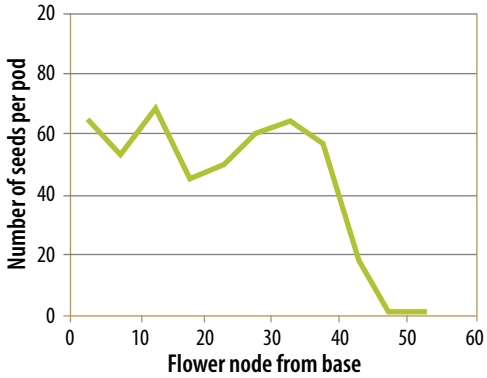
- Flowers at the base of the main raceme are more sensitive to freezing than those in the middle.
- Even unstressed plants abort flowers. This is due to the competition for assimilates with the lower flowers.
- 40% of the first 10 flowers did not develop into pods.

Figure 3–10. Freeze sensitivity of flowers on the main raceme (flower 1 was the first to open).
Source: Lardon and Triboi-Blondel (1995)



- Number of seeds per pod depended little on the pod position on the raceme of the unstressed plants.
- Number of seeds per pod was lower in plants subjected to freezing than the control, particularly at the base of the main raceme.
- The number of seeds per pod decreased dramatically on the lower half of the main raceme of the stressed plants, and many pods aborted.

Figure 3–11. Effect of freezing on number of seeds per pod on the main raceme (flower 1 was the first to open).
Source: Lardon and Triboi-Blondel (1995)



- Flower 1 was the first to open.
- Many ovules on the lower halves of the stressed plants were injured.

Figure 3–12. Freeze sensitivity of ovules on the main raceme. Source: Lardon and Triboi-Blondel (1995)

Thermal time

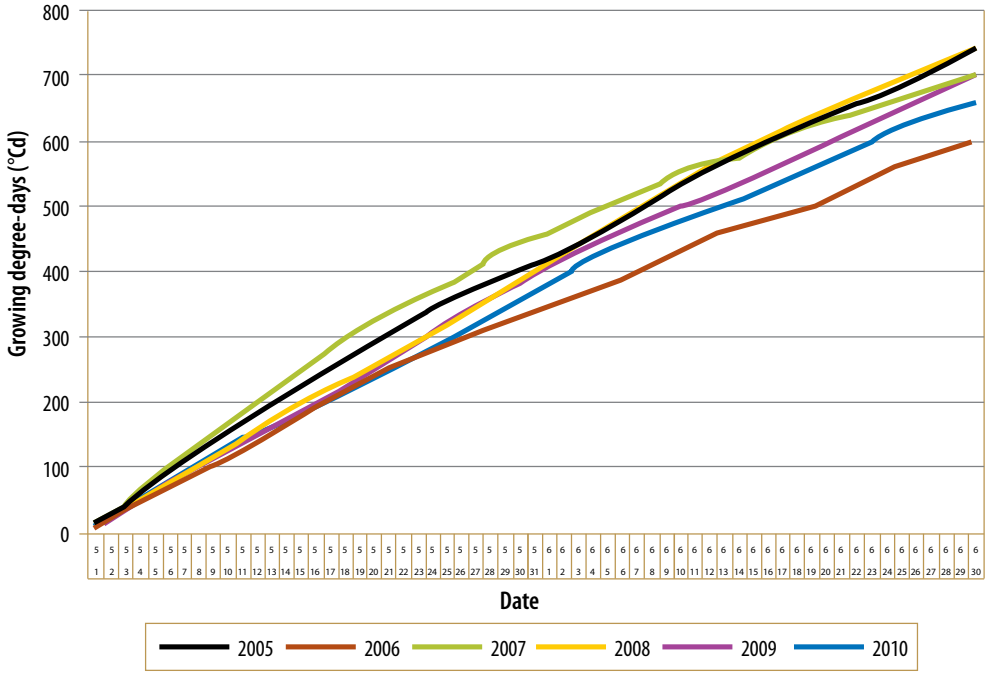
The underlying variable driving development rate is temperature.

Thermal time is a way of expressing accumulated temperature. It helps to explain the relationship between plant development and temperature. It is calculated as the mean daily temperature minus a base temperature and is recorded as degree-days (°Cd). The base temperature is the minimum temperature at which the plant grows, and this varies for each crop. For canola, the base temperature is 0°C during vegetative growth and 5°C in the reproductive phase.

Thermal time varies from year to year in the same location (Figure 3–13).

Table 3–1 lists the approximate number of degree-days required for each growth stage in canola. For example, the length of time from sowing to emergence (fully open cotyledons) in terms of thermal time is about 120 degree-days.

Although not commonly used at the farm level, thermal time is very important for predicting the growth stages of some crops. It is a better indicator in canola than in wheat. In general, high temperatures accelerate development, reducing the number of days between germination and flowering.



- Thermal time varies among years.
- In 2007, nearly 450°Cd accumulated during May, compared with less than 350°Cd in 2004 and less than 300°Cd in 2006.

Figure 3–13. Variation in thermal time across 5 years for the month of May at Cowra.
Source: Jan Edwards

Table 3–1. Growing degree-days required for each growth stage in canola.

GROWTH STAGE	CUMULATIVE DEGREE-DAYS FOR *B. NAPUS*
Emergence – cotyledons completely unfolded	120–150
Two leaves unfolded	282–324
Four leaves unfolded	411–463
Flowering begins – at least one open floret on 50% or more of plants	533–621
Flowering 50% complete	759–852
Seed-fill begins – 10% of seeds have reached final size	972–1074
Maturity: seed begins to mature –10% of seed has changed colour	1326–1445
Windrowing: 40% of seed on main stem has changed colour	1432–1557

Source: Modified from Canola Council of Canada (2003)

Photoperiod

Photoperiod or day length is the duration (number of hours) of exposure to daylight. Canola, like wheat, is a long-day plant, meaning that the length of time from sowing to flowering is shortened under long days.

This sensitivity determines whether the plant continues to produce leaves or switches to reproductive development.

Photoperiod can affect the development of canola by:

- causing changes in the rate of leaf area expansion and dry matter production
- providing a cue for the start of reproductive development
- changing the rate of reproductive development.

Long days reduce the length of the phase from initiation to stem elongation and the length of the stem elongation phase.

Under typical Australian field conditions the average photoperiod between sowing and flowering varies between 11 and 13 hours. Spring varieties of *B. napus* from Canada, Europe and Australia have been found to respond to day lengths beyond 10 to 12 hours: the European varieties were most responsive to increased day length, followed by the Australian varieties, with a Canadian cultivar the least responsive.

Vernalisation

Vernalisation is a cold requirement that needs to be met before a plant switches from vegetative growth to reproductive growth.

Sowing date has less effect on flowering date in varieties that require vernalisation. Varieties that require vernalisation can be sown earlier, as they will not flower until their cold requirement is met. This reduces the risk of frost damage during flowering and seed-fill.

In canola, the effect of vernalisation is to lengthen the period between the germination and bud-visible stages, not the phase between bud visible and flowering. Vernalisation also results in smaller numbers of leaves on the main stem at flowering.

Canola varieties can be categorised as spring (requiring little or no vernalisation) and winter (requiring vernalisation). The majority of varieties sown in Australia are spring varieties that do not respond to vernalisation.

Winter canola has a vernalisation requirement that needs to be met before it switches to the reproductive phase. The length of the vernalisation requirement varies with the variety. There is currently one commercial winter canola variety available.

High temperatures can cause 'devernalisation', in which plants lose some of their accumulated temperature response.

Earliness

As well as the influence of temperature, photoperiod and vernalisation on maturity there is earliness. Some varieties are genetically quicker than others. In the range of current commercial varieties there are minor differences in earliness.

Plant maturity

The maturity of varieties available in NSW varies. Varieties fall into one of five categories: early, early-mid, mid, mid-late or late. The relative maturity of varieties does change with location. Up-to-date tables on the sowing windows for various varieties are published each year in DPI's *Winter crop variety sowing guide*. See also 'Sowing time' in Chapter 1.

The time to maturity, or length of time taken for a variety to reach flowering, depends on the vernalisation, photoperiod and thermal time requirements, as discussed above. Recommended sowing times are arrived at by assessing the maturity of varieties in different environments and with different sowing times. Table 3–2 shows the wide range in developmental stages that occurs with different sowing times.

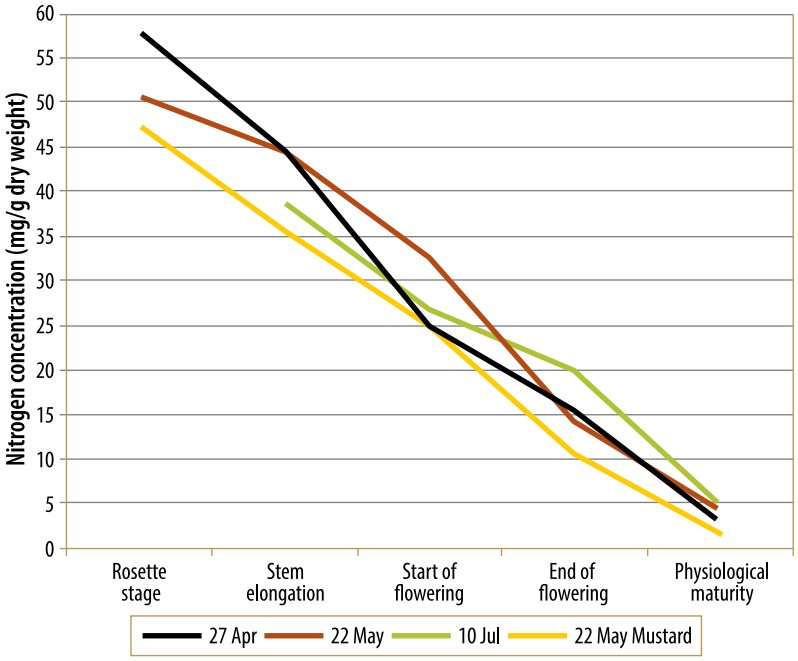
Table 3–2. A wide range in developmental stages occurs with different sowing times.

SOWING TIME	VEGETATIVE PERIOD (DAYS FROM SOWING TO FLOWERING)	DURATION OF FLOWERING (DAYS)	DURATION OF SEED FILL (DAYS)	DAYS FROM SOWING TO PHYSIOLOGICAL MATURITY
27 April	130	30	69	199
22 May	119	28	62	181
10 July	91	21	63	154
Mustard 22 May	105	42	77	182

Source: Hocking and Stapper (2001)

- Later sowing decreased the number of vegetative days and the duration of flowering.
- Later sowing has less impact on the duration of seed-filling.
- Later sowing shortens the number of days to flowering in both canola and mustard.

Delayed sowing can result in lower nitrogen concentrations in the shoots, despite the plants being smaller, because plants from early sowings have more time to take up and store nitrogen (Figure 3–14).



- Nitrogen was added to the soil at 53 mg/kg to a depth of 10 cm at sowing.
- Delaying sowing reduces the nitrogen concentration of shoots.
- Plants from early sowings have more time to take up nitrogen.

Figure 3–14. Effect of sowing time on concentration of nitrogen in the shoots of canola and mustard at Ariah Park, NSW. (Does not include seed at physiological maturity.)
Source: Hocking and Stapper (2001)

Nutrients

Nitrogen

In canola the concentration of nitrogen in shoots declines through the growing season. The major decline occurs between the start of flowering and maturity, a time when dry matter production is proportionally greater than nitrogen uptake. Much of the dry matter produced in this period is associated with branch, rather than leaf, production. Most of the leaves have senesced by the end of flowering.

The most rapid uptake of nitrogen is during stem elongation, and maximum plant levels are found around the start of flowering. By then the crop may hold 150 kg/ha of nitrogen (Figure 3–15).

Sulfur

By flowering, sulfur deficiency symptoms can show up in the petals. If severe sulfur deficiency has occurred in the vegetative stage, symptoms can be found in both foliage and flowers: petals can be smaller and lighter yellow. However, if sulfur deficiency occurs at around flowering, leaf symptoms may not be obvious, but flower petals may become paler. Yellow and white petals may even exist side by side on a single flower.

The lifespan of sulfur-deficient petals is shortened to 1 day from the normal 2 or 3, and pollen production is greatly reduced. In addition, sulfur-deficient petals are egg-shaped, compared with the more rounded petals on plants that have enough sulfur. Flowering is often delayed and prolonged. Sulfur deficiency can delay plant maturity. Reports suggest that sulfur-deficient plants do not attract honey bees, perhaps because of a lack of pollen.

Topdressing with adequate amounts of sulfur as late as the stem elongation stage can cause a recovery of seed yield and oil concentration in severely sulfur-deficient canola.

Increasing the sulfur supply increases the glucosinolate concentration in canola seed.

Boron

Boron deficiency can restrict pollen tube growth. This is why boron demand is higher during the reproductive stage than the vegetative stage. Boron also affects fertilisation and seed set by increasing pollen production of the anthers and pollen viability. It may influence the sugar composition of the nectar.

Boron deficiency symptoms are most likely to be seen during flowering. Symptoms include:

- deformed, curled and rough skinned leaves with torn margins
- yellow to brown spots in the interveinal areas of leaves
- red to brown-purple margins on the new leaves
- interveinal mottling
- early leaf drop
- shortened stems
- cracked stems
- prolonged flowering
- flower sterility
- poor pod set and yield.

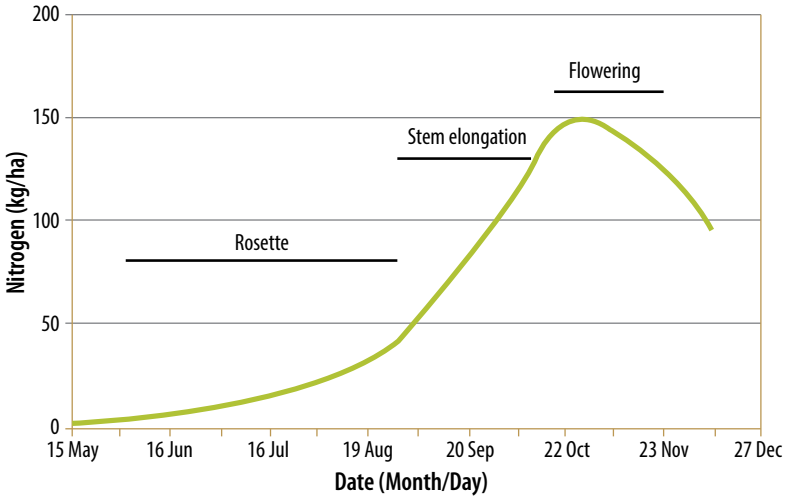


Figure 3–15. Nitrogen uptake in a well-fertilised crop. Source: Norton (1992)

Grazing

Grazing delays flowering and reduces plant height. The extent of these changes depends on the intensity of grazing and the stage of the plant at the time of grazing. One study found that the delay in flowering was 0 to 3 days if grazing occurred before buds were visible, 4 days if buds were visible but not elongated, 7 to 15 days if buds were elongated to 25 cm but no flowers were open, and 26 to 30 days if plants were in flower (Table 3–3).

Table 3–3. Impact of grazing on the timing of flowering.

PLANT STAGE WHEN GRAZED	DELAY IN FLOWERING	HEIGHT REDUCTION (CM)
Vegetative stage before buds visible	0 to 3 days	None
Plants with buds visible but not elongated	4 days	Up to 10 cm
Buds elongated (20–30 cm) but no flowers	7 to 15 days	10 to 20 cm
First flowers open	26 to 30 days	30 to 40 cm

Source: Kirkegaard (2007)

Cutting a failed crop

Table 3–4 shows the changes in plant quantity and quality. By flowering, the stem makes up 60% to 80% of available feed. The high-nutrient-value leaf material retains its energy and protein well over the period, but quantity declines rapidly. Although pod quantity and energy values increase during seed fill, the pod represents less than 14% of the available feed.

Figure 3–16 shows the decline in both yield and digestibility of a standing canola crop in 2006. Dry matter yield increased slightly after mid October, but the dry matter was all poorly digestible stem, seed and pod material. Leaf mass continued to decline after flowering. This is reflected in the digestibility fall after flowering, which was substantial.

Table 3–4. Changes in partitioning of dry matter, crude protein and metabolisable energy with time of cutting of canola.

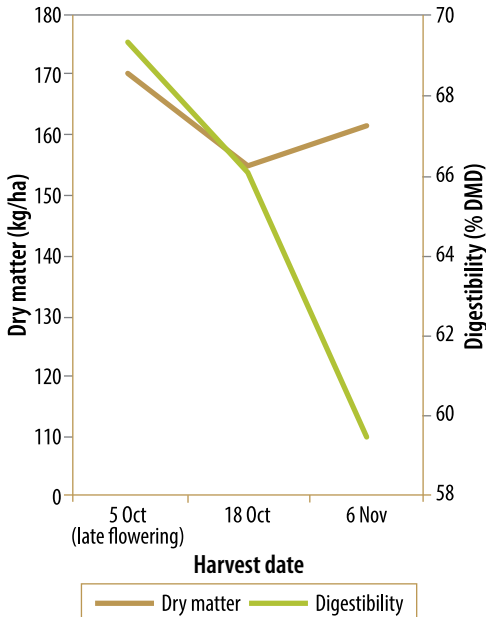
DATE	DRY MATTER (KG/HA)		
	LEAF	STEM	POD
5 October 2006	505	1178	135
18 October 2006	376	861	189
6 November 2006	292	1100	209
17 November 2006	126	934	121
	CRUDE PROTEIN (%)		
	LEAF	STEM	POD
5 October 2006	31.4	17.4	22.8
18 October 2006	32.2	17.5	28.6
6 November 2006	32.2	15.2	17.3
17 November 2006	33.4	11.9	12.6
	METABOLISABLE ENERGY (MJ/KG DM)		
	LEAF	STEM	POD
5 October 2006	11.1	9.2	10.8
18 October 2006	11.1	8.5	10.6
6 November 2006	10.7	8.1	13.6
17 November 2006	11.5	7.2	11.7

Source: Phillips and McMullen (2007)

- The stem makes up 60% to 80% of available feed.
- The protein and energy values of the leaf material do not fall.
- Quantity (dry matter) declines rapidly.
- At seed filling, the pods represent less than 14% of the available feed.

Dry matter digestibility

Dry matter digestibility (DMD) is the proportion of dry matter in the feed that can be digested by an animal. It is expressed as a percentage of dry matter. Dry matter is everything remaining after the water in the sample has been removed.



- Dry matter is highest at flowering, decreasing in mid October then increasing slightly as the pods and seeds mature.
- Digestibility falls quickly after flowering.

Figure 3–16. Effect of forage harvest date on the yield and digestibility of canola. DMD = dry matter digestibility. Source: Phillips and McMullen (2007)

As a general rule, a drop of 1% in digestibility will result in a 3% to 5% drop in animal performance. Cutting canola crops after flowering will usually result in an increased cost per megajoule of the final product.

The feed quality of the canola crops harvested in southern NSW for forage in 2006 was generally high (Table 3–5). Feed quality testing of canola crops in 2007 gave similar results.

Table 3–5. Feed quality of 2006 canola at cutting, southern NSW

	AVERAGE	RANGE
Digestibility (DMD %)	69.4	57–76
Metabolisable energy (MJ/kg DM)	10.0	7.9–11.6
Crude protein (%)	21.0	12–31
Nitrate (mg/kg)	2540	52–8394

Source: Phillips and McMullen (2007)

References and further reading

Hocking PJ, Stapper M 2001, Effect of sowing time and nitrogen fertiliser rate on growth, yield and nitrogen accumulation of canola, mustard and wheat. *Australian Journal of Agricultural Research* 52, 623–634

Kirkegaard JA 2007, *Evaluating the Potential for Dual-purpose Canola in the Mixed Farming System of Southern Australia.* Report to Grains and Research Development Corporation on Project CSP00085. CSIRO, Canberra

Lardon A, Triboi-Blondel AM 1995, Cold and freeze stress at flowering, effects on seed yields in winter rapeseed'. *Field Crops Research* 44, 95–101

Ma Q, Niknam SR, Turner DW 2006, Responses of osmotic adjustment and seed yield of *Brassica napus* and *Brassica juncea* to soil water deficit at different growth stages. *Australian Journal of Agricultural Research* 57, 221–226

McGregor DI (ed.) 1991, Rapeseed in a Changing World. In *Proceedings of the GCIRC Rapeseed Congress* 1991, July 9–11, Saskatoon, Saskatchewan Canada. Vol. 3 of 6

Norton R 1992, Nitrogen is the key canola nutrient. In Casey M, Cooke P (eds) *Canola Cache. The Farmer's Handbook for Growing Canola*. Kondinin Group, Perth

Phillips N, McMullen G 2007, *Drought affected canola and wheat – feed quantity and quality decline in standing crops.* NSW DPI, Orange

Robertson MJ 2001, Understanding how environment and genotype determine time to flowering in canola and Indian mustard. In *Proceedings of the 12th Australian Research Assembly on Brassicas,* Geelong Vic., NSW Agriculture, Orange NSW. pp. 96–100

Robertson MJ, Watkinson AR, Kirkegaard JA, Holland JF, Potter TD, Burton W, Walton GH, Moot DJ, Wratten N, Farre I, Asseng S 2002, Environment and genotypic control of time to flowering in canola and Indian Mustard. *Australian Journal of Agricultural Research* 53, 793–809

Robertson MJ, Kirkegaard JA 2003, Crop modelling for the Australian canola industry: a review. In Proceedings of the *13th Biennial Australian Research Assembly on Brassicas,* 8–12 September 2003, Tamworth, NSW. NSW Agriculture, Orange NSW

Tayo TO, Morgan DG 1975, Quantitative analysis of the growth, development and distribution of flowers and pods in oil seed rape. *Journal of Agricultural Science*, Cambridge. 85, 103–110

Young LW, Wilen RW, Bonham-Smith PC 2004, High temperature stress of *Brassica napus* during flowering reduces micro- and megagametophyte fertility, induces fruit abortion, and disrupts seed production. *Journal of Experimental Botany* 55(396), 485–495.

IN THE PADDOCK

The following are some examples of the activities that can be done in the paddock to illustrate the stages of reproductive development that have just been discussed. These are practical exercises to help farmers assess the progress of their crop at this stage.

Flowering

Aim: to assess the stages of flowering.

1. **Start of flowering.** Examine the crop. Determine when 10% of plants have an open flower. Record this as the start of flowering.
2. **Examine the crop.** Determine when 50% of plants have an open flower. Record this as mid-flowering.
3. **End of flowering.** Examine the crop. Determine when only 10% of remaining plants have an open flower. Record this as the end of flowering.
4. Identify whether frosted flowers are present (they will look brownish yellow). Compare the appearance of the frosted flowers with that of healthy flowers.
5. Identify parts of the paddock that may have advanced/delayed flowering; identify the different topography etc. that may have made these patches more or less susceptible to frost.
6. Once the above have been identified, discuss options such as sowing time, variety and other agronomic factors.

Thermal time

Aim: to calculate the accumulated thermal time for a location.

1. Using the records from the nearest meteorological station, calculate the mean temperature for each day:

$$\frac{\text{Daily maximum temperature + daily minimum temperature}}{2}$$

IN THE PADDOCK

2. Add the mean temperatures together to give the accumulated degree-days.

DAY	MAXIMUM	MINIMUM	MEAN	ACCUMULATED DEGREE-DAYS
1				
2				
3				
4				
5				
6				
7				
8				
9				
10				
11				
12				
13				
14				
15				
16				
17				
18				
19				
20				
21				
22				
23				
24				
25				
26				
27				
28				
29				
30				
31				
Total				

4. Pod and seed development

by Kathi Hertel and Jan Edwards

Chapter Snapshot

Introduction

Seed development is the period from flowering to physiological maturity and is the final stage in the life cycle of the canola plant. Carbohydrates and oil are deposited in the seed as it grows and ripens. Final seed yield is determined during this phase and is influenced not only by conditions at the time, and management decisions, but by everything that has preceded it. Seed quality is greatly affected by the conditions during seed development. Chapter 4 explains how the seed develops and reaches physiological maturity, and the environmental conditions that influence its progression.

Learning Outcomes

At the end of this chapter, you will be able to:

- describe the stages of seed development
- list the sources of oil and carbohydrate for seed fill
- understand the impact of conditions during seed development on seed yield and quality
- identify the different components of yield, when they are set, and what influences final yield
- assess variations in seed size in the pod
- estimate seed yield
- calculate harvest index and water use efficiency.

Pod and seed development

Pod and seed development is the period from flowering to physiological maturity when the fertilised flowers develop into pods. This phase overlaps with the flowering stage, as the pods begin to grow immediately after each flower is fertilised. Because canola is indeterminate, there will be multiple growth stages on each plant.

Pod development – Growth stages 5.1–5.9

Fruit
The pods and seeds of the canola plant.

Once a flower is fertilised, the pod begins to grow (Figure 4–1). Pod development starts on the lowest third of the branches on the main stem. Pods grow rapidly in length, to between 6 and 9 cm. They reach their maximum fresh weight and length by the end of the first half of **fruit** development. By this stage, the seeds have accumulated only 35% of their mature dry weight.

Pods reach maturity about 82 days after flowering of the parent flowers. A mature pod contains between 15 and 25 seeds, which account for about 60% of the total pod dry weight.

Figure 4–1. Pod development begins immediately after a flower is fertilised. It starts on the lower branches before the upper flowers are fertilised. Photo: Jan Edwards

The stages of pod development are defined by the proportion of pods that have extended to more than 2 cm. See Table ii in the *Introduction* for these stages.

Not all the flowers will develop into pods, even in unstressed plants. Most of the lower (basal) half of the main raceme will develop pods, but towards the upper part of the raceme the number of pods decreases because of competition for assimilate. These pods have flowered and developed later than the lower ones.

Pod development coincides with a reduction in the number of leaves on the plant. After flowering, the pods are the major photosynthetic organs on the plant. The pod also functions as a temporary storage organ for key nutrients such as nitrogen and phosphorus, which are then redistributed to the developing seeds. This is particularly valuable when limited soil moisture reduces nutrient uptake during pod filling.

Pod walls are also an important source of zinc, magnesium and copper for seeds, but there is little redistribution of potassium and sulfur to the seeds. The lack of redistribution of sulfur from the pod walls may be related to the low levels of glucosinolates in canola.

Side branches compete with the main stem for nutrients and assimilates.

Seed development – Growth stages 6.1–6.7

Seed development begins before the pods have expanded to their full length.

Stage 1: Seed enlargement – Growth stage 6.1

The first process is expansion of the seed coat to it full size. The seed is initially translucent and watery (Figure 4–2). The seed expansion process begins about 15 days after fertilisation and takes about 12 days. The seed embryo then grows rapidly to fill the space occupied by the fluid.

Seeds have accumulated only 35% of their mature dry weight by the time the pod walls have fully elongated.

Figure 4–2. Seed development stage 1: seed enlargement. Photo: Lowan Turton, DPI

Stage 2: Seed fill – Growth stages 6.1–6.3

When the pods are nearly full length, about 20 days after flowering, rapid seed fill begins. It lasts between 35 and 55 days and comprises two processes: oil deposition and protein deposition. Oil and protein accumulation in the seed start and finish at the same time, but the rate of accumulation differs.

Seeds grow at about 0.08 to 0.12 mg/day. The rate of development depends on temperature. At a mean temperature of 15°C, rapid seed growth begins about 20 days after flowering. At 20°C, rapid seed growth begins 10 days after flowering.

Protein accumulates rapidly in the early stages of seed development. Storage proteins begin to accumulate when the embryo starts to grow rapidly. Most of the protein in the mature seed is found in the cotyledons: about 76% is in the cotyledons, 17% in the rest of the embryo, and 7% in the seed coat.

Oil is synthesised from the carbohydrates stored in the pods, stems and remaining leaves. Most of the oil is synthesised during the period from 35 to 55 days after flowering. Oil is deposited into the seed until 40% of the seeds have changed to their mature black appearance, at around 60 days after flowering. At this point the seed oil concentration plateaus.

At the completion of this stage the seed has enough oil and protein reserves to support future germination and seedling growth.

The developing seeds are green because of their chlorophyll content.

Stage 3: Physiological maturity – Growth stages 6.4–6.8

By 42 days after flowering, seed weight has almost doubled, and seed development is complete. During the next 2 weeks (from 50 to 72 days after flowering), the seeds dehydrate and change from green to black (Figure 4–3). The oil concentration of the seed is maximised by the time 40% of the seeds have changed colour. When 60% of the seeds have changed colour, maximum dry weight has been reached. Seeds are fully mature (physiological maturity) about 80 days after flowering.

The growth pattern of the pod wall and seeds is summarised in Figure 4–4. The pod walls reach their maximum weight about 50 days after flowering. A significant loss (20%) of dry matter from the pod walls occurs during the last half of seed development as carbohydrate and nutrients are translocated into the seed. The seeds reach their maximum weight about 70 days after flowering.

Windrowing
Cutting the crop and placing it in rows on the cut stubble. Windrowing hastens the drying rate, ensures even ripening and reduces shattering.

Figure 4–3. Seed colour change in canola.
Photos: Michel Dignand

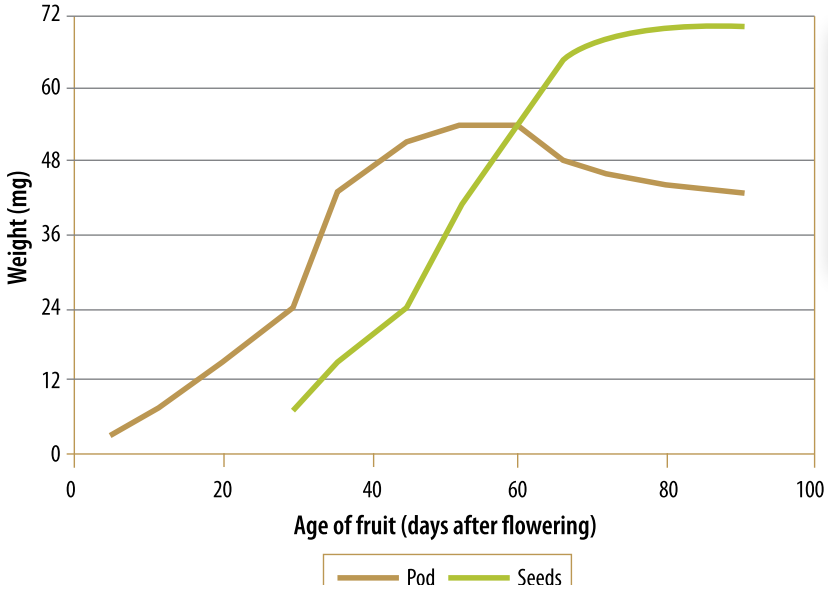
Harvest maturity – Growth stage 6.8

The most common method for harvesting canola is to **windrow** the crop, allow it to dry down, and then harvest the windrows (Figure 4–5). The timing of physiological maturity will determine the correct time for windrowing.

Windrowing can start when 40% of the seeds have changed to their mature colour and the seed moisture content is 9% (Figure 4–6). Windrowing before 40% of the seeds have changed to their mature colour can result in substantial reductions in both seed weight per pod and seed oil concentration.

By the time of the 40% colour change, seed oil concentration has reached its maximum. However, there is a penalty in terms of the dry weight of seeds per pod. Windrowing at this time can mean a 10% reduction in 1000-seed weight (Figure 4–7). Waiting until 60% of the seeds have changed colour allows the seeds to reach maximum dry weight. The change from 40% to 60% usually takes 3 or 4 days. However, there is an increased risk of shattering if windrowing is done at this later stage.

Harvesting of the windrows can begin when the moisture content is 8%. This is usually 5 to 10 days after windrowing.



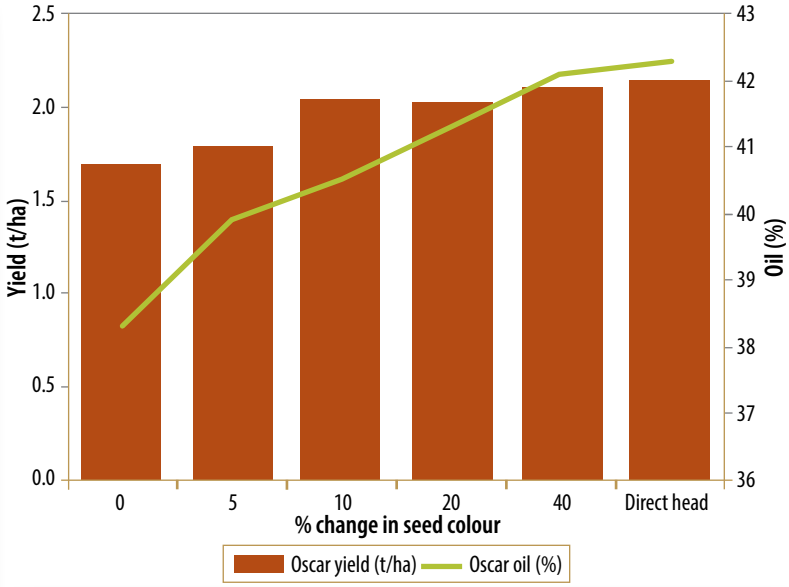
- Pod development peaks about 50 days after flowering.
- At this stage seed development has already begun.
- Seeds reach their maximum weight about 70 days after flowering

Figure 4–4. Patterns of growth of pod walls and seed of canola.
Source: Hocking and Mason (1993)

Figure 4–5. A windrowed canola crop.
Photo: Jan Edwards, DPI

Figure 4–6. Seed colour change: 40% of the seeds have changed colour from green to mature. Source: R Colton and L Banks, DPI

Canola (and more commonly mustard) can be **direct headed** when all the pods are dry and rattle when shaken. The bottom pods on the plant will ripen first. Harvest should begin when the seed moisture content has reached 8% or less. Above 9% moisture the seed can heat up rapidly, making storage difficult.



- Yield and oil content are optimal at 40% seed colour change.

Figure 4–7. Effect of the timing of windrowing (measured by the percentage change in seed colour) on yield and oil. Source: Anon. Increased yield and oil contents in canola with optimum timing in the windrowing/swathing operation. *Dovuro Agronomic Update* 1998

Sources of assimilates for pod and seed growth

Protein

The protein content of the developing seed is established before the oil content. Protein is built from simple amino acids. Nitrogen is an essential component of amino acids.

Storage proteins accumulate when the embryo starts to grow rapidly, to replace the endosperm and fill the fully expanded seed coat.

Seed protein accumulation coincides with rapid cell expansion and a rapid increase in embryo weight; about 52% of the nitrogen content of mature canola plants is accumulated before flowering.

Nitrogen can be mobilised from stems and upper leaves and moved to the developing pods. The pods provide 23% to 33% of the nitrogen content of the seeds.

Most of the protein in the mature seed is found in the cotyledons: about 76% is in the cotyledons, 17% in the rest of the embryo, and 7% in the seed coat.

Direct heading
Harvesting the standing crop without windrowing.

Not all the nitrogen in the plant is relocated to the seed. Some is lost when leaves are shed. In one study the amount of dry matter lost in shed leaves ranged from 1 to 1.75 t/ha, equivalent to 10 to 30 kg/ha nitrogen.

In one study, late sowing resulted in a higher seed nitrogen concentration (Table 4–1). Nitrogen removal in the seed of April-sown canola is about 35% higher than in the grain of wheat sown at the same time. From May and June, the amounts are similar.

Table 4–1. Effect of sowing time on concentrations of nitrogen in the seed of canola at Ariah Park, NSW. Nitrogen was applied to the soil at sowing at 53 mg/kg at a depth of 10 cm.

SOWING DATE	CONCENTRATION (%) OF N IN THE SEED	N REMOVED IN THE SEED (KG/HA)
27 Apr	3.1	73
22 May	3.8	49
10 Jul	4.0	34

Source: Modified from Hocking and Stapper (2001)

Carbohydrates

There are two sources of carbohydrates. The first is from photosynthesis during seed fill. Photosynthesis during seed fill comes mainly from the pods, but also the stem. At this time, most of the leaves have died. The canola pod is capable of photosynthesis and is an important source of assimilates for the developing seeds, especially after the onset of rapid pod growth, as most of the leaf canopy has died off by this time.

Assimilates
Products of photosynthesis.

The second is the redistribution of **assimilates** from dying plant parts (mainly leaves) and from the pods. Leaves are the main source of these redistributed assimilates. Redistributed dry matter and assimilates from the pod walls can provide 11% (dry matter) to 25% (nitrogen) of the amount in mature seeds.

During photosynthesis, carbon dioxide is converted to sugars. Most of these sugars are used to form the cell walls of the plant. Excess sugars are stored in the plant as water-soluble carbohydrates.

Pods intercept about 70% of the incoming light; therefore, the seeds grow in a light-limited environment. The amount of light reaching the pods is strongly influenced by canopy structure.

When light is excluded from 30 days after flowering, there is an increase in the dry weight of the pod walls, with a decrease in the proportion of the dry matter appearing in the seeds.

Water-soluble carbohydrates contribute only a small amount to seed fill (about 12% to 18% of final seed yield). Decreased dry matter production (e.g. from shading) decreases the assimilate supply and causes increased reliance on reserves.

Oil

Oil is synthesised in the seed from the carbon molecules in simple sugars. Sucrose (from the leaves, stems and pods walls) is converted to fatty acids and then synthesised into oil. This process uses energy derived from light by chloroplasts in the embryos. Light stimulates the chlorophyll in the embryo of the canola seed to produce the energy for synthesising oil.

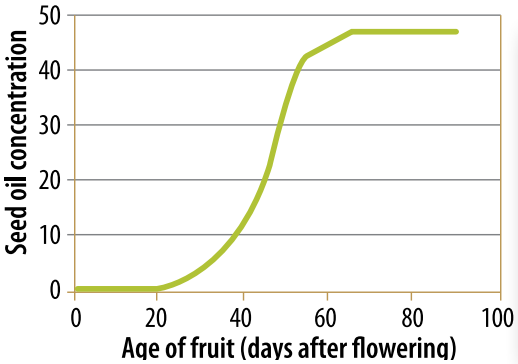
Most of the oil is synthesised during the period from 35 to 55 days after flowering. Cotyledons are the main oil storage organs. The stored oil droplets increase in size and number between 20 and 30 days after pollination. Oil is deposited in the seed until 40% of the seeds have changed to their mature black colour, at around 60 days after flowering. At this point the seed oil concentration plateaus at about 30% to 50% (Figures 4–8 and 4–9).

Oil content is not directly linked to seed size.

Oil composition is more strongly related to genetics than to seed yield.

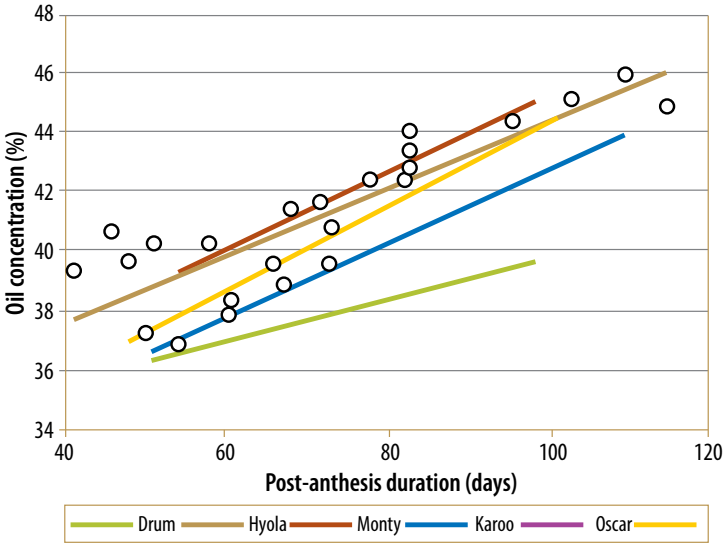
In general, high temperatures, moisture stress and high nitrogen levels decrease oil concentration. Variety is also a factor. Generally, triazine-tolerant varieties have lower oil concentrations because of their less efficient photosynthetic systems.

Oil synthesis is more sensitive than protein synthesis to high temperature. Oil concentration is negatively correlated with protein content. Every 1% increase in seed



- Oil concentration in the seeds increases rapidly from 20 days after flowering
- Oil percentage plateaus about 60 days after flowering when the seed is 70% of its final dry weight.
- Final oil concentration will be between 30% and 50% at delivery, depending on heat, moisture and nitrogen supply during seed filling.

Figure 4–8. Pattern of oil concentration in seeds. Source: Hocking and Mason (1993).



- Seed oil concentration increases after flowering.
- Different varieties accumulate oil at different rates.
- The amount of oil is more strongly related to genetics than seed yield.
- Early-flowering cultivars produce high oil concentrations.

Figure 4–9. Canola seed oil concentration (%) after flowering in five canola varieties – Drum, Hyola, Monty, Karoo and Oscar – grown in Western Australia in 1993 and 1997. Source: Si and Walton (2004)

protein percentage (above 22%) due to heat stress is accompanied by a reduction in oil percentage of around 1.6%.

There is also a negative correlation between oil content and glucosinolate content.

Oil and protein accumulation in the seed start and finish at the same time, but the rates of accumulation differ. Environment has a larger effect on oil and protein than does variety, and the effect of the year is larger than the effect of the region. Among the environmental factors that regulate oil content, temperature is probably the most important (Table 4–2). Wetter and colder springs favour higher oil contents and low protein contents. High temperatures or water stress will stop oil biosynthesis, making the protein contents higher than under unstressed conditions. A mean spring temperature rise of 1°C would reduce oil contents by 0.5%.

Table 4–2. Correlation coefficients between environmental variables in the spring and seed quality.

	SEED OIL %	SEED PROTEIN %
Spring rainfall	+ 0.30 **	– 0.27 *
Mean maximum temperature	– 0.40 ***	0.29 *

Significance: * $P < 0.05$; ** $P < 0.01$; *** $P < 0.001$
Source: Pritchard et al. (2000)

Chlorophyll

Chlorophyll accumulates in the seed embryo during the filling process (20 to 30 days after flowering). Levels are highest at the start of the filling process but decline as the seed approaches physiological maturity. Enzymes within the seed break down the chlorophyll.

Retention of chlorophyll in canola seed can reduce the value of the crop. When canola seed is crushed, chlorophyll is extracted with the oil and is then difficult to remove by conventional bleaching processes. Expensive processes are required to purify the oil. Chlorophyll discolours the oil and increases the rate of oxidation, reducing the oil's shelf-life. It inhibits the hydrogenation catalyst used for hardening in the manufacture of margarine.

Frost damage or other stress during seed-filling is a major cause of high chlorophyll levels. Time of sowing is critical for managing this risk.

Competition between plant parts

Flowering, pod development and seed development occur in sequence. Because individual canola flowers open over a long period (up to 55 days), there will be seeds and pods at different stages of development on one plant. There is therefore competition for assimilates. Earlier-developed pods have a competitive advantage over the later-formed ones.

Limitation of the supply of assimilates made by the plant is the main cause of cessation of flowering and seed abortion.

Green area index
The ratio of leaf green area to the area of ground on which the crop is growing

Reduction in the supply of assimilates around the time of flowering is particularly harmful, since in addition to reducing the number of pods that develop it appears to restrict the pods' capacity for compensatory growth when the supply returns to normal.

The supply of carbohydrates at, or after, flowering therefore regulates the yield of seeds and pods. Prolonged stress results in smaller pods and fewer, lighter seeds.

Seed filling places large demands on moisture and assimilates, and there is competition between the pods on each branch, as well as between branches.

Solar radiation

At flowering, incoming solar radiation is intercepted by the flowers. Only 40% of the incoming solar radiation gets past the flowers to the leaves. With less radiation available to the leaves, the rate of photosynthesis decreases and leaf death increases; the **green area index** (Figure 4–10) then drops. Consequently, crop growth over the flowering period can be reduced by up to 27%. This is significant, as it is the time when seed numbers are being determined in the earlier-formed pods and these pods are therefore at risk of seed abortion.

- The total green area of the plant peaks at late flowering.
- The proportion of green area made up by leaves falls rapidly after the vegetative growth stage.
- By pod development there are very few leaves left.
- The area of the pod increases after flowering.

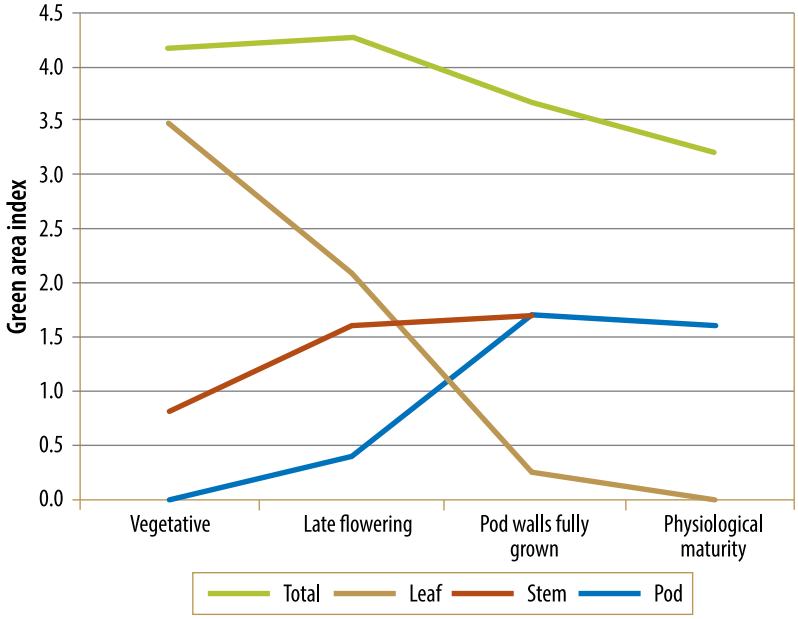


Figure 4–10. Green area index at different stages of development in the canola variety Rafal.
Source: Norton et al. (1991)

Pods at this stage of decreased radiation availability have not established their photosynthetic capacity; the pod walls are growing rapidly and the assimilate supply from the leaves is greatly reduced, leading to risk of seed abortion.

The final seed number per pod is primarily a result of the amount of radiation intercepted by each pod. Pods and stems are less efficient than leaves in terms of photosynthetic capacity on an area basis, because they have fewer stomata.

The lower-most pods have the advantage of being closer to carbon sources and tend to be more productive than the upper sections of the terminal flower head.

In dense crops, self-pollination is important, because as the pod canopy develops the earliest pods may be shaded by later ones. Only 20% to 30% of radiation reaches the lower-most layer of the canopy. The amount of solar radiation hitting a particular pod is one of the main factors determining the photosynthetic rate.

Factors affecting seed development

The period 2 to 3 weeks after flowering is a critical time for seed abortion. From a constant number of 30 ovules per pod at flowering, the number of surviving seeds declines over a 3-week period. It is stable in the last 3 to 4 weeks before maturity. In dense canopies, seed losses are larger and occur mainly in the lower canopy. In late-sown crops there is less competition between the pods and little difference in seed abortion between levels within the canopy. The period of most seed loss coincides with the main growth of pod walls, before the seeds start their rapid weight increase.

Moisture

Moisture stress

Table 4–3 shows the effects of moisture stress on the yield components in Monty canola. Water stress at flowering has the greatest impact on the number of pods per plant. Moisture stress also increases seed glucosinolate concentrations.

Table 4–3. Seed yield and yield components of Monty canola either well watered or subjected to a single water deficit at different stages under glasshouse conditions.

TREATMENT	PODS/PLANT	SEEDS PER POD	100-SEED WEIGHT (G)	SEED WEIGHT (G/PLANT)	TOTAL PLANT WEIGHT (G)	HARVEST INDEX
Well watered	390	15.3	0.35	20.7	70.1	0.3
Water stress at early vegetative stage	457	15.1	0.34	23.4	76.6	0.3
Water stress at elongation	377	13.9	0.36	18.3	68.6	0.27
Water stress at flowering	329	12.7	0.35	14.7	64.5	0.23
Water stress at seed fill	384	11.3	0.36	15.0	67.6	0.22
l.s.d. ($P = 0.05$)	93	n.s.	n.s	3.0	8.6	0.04

Source: Qifu et al. (2006)

- Seed number per pod was unaffected by water stress.
- Water stress at elongation, flowering and seed fill decreased the number of pods per plant.
- Late water stress decreased seed weight.
- Harvest index also fell with late stress.

Temperature

Heat stress

Canola is susceptible to heat stress, even if it occurs only for short periods during seed development. High temperatures accelerate the time to seed maturity, lower yield, and lower oil content (Table 4–4). High temperatures can also change oil quality.

Seed coat development is complete 25 days after pollination. High temperatures during this time are thought to result in thinner seed coats. Yield is reduced in response to heat shock, owing to a reduction in the number of pods per plant (up to 31%) and in individual seed weight (by up to 72%).

Oil and protein content are inversely related and are influenced by temperature through its effect on nitrogen uptake. Increasing temperature increases nitrogen uptake; this in turn favours an increase in protein and a decrease in oil synthesis.

Higher temperatures of up to 26.5°C inhibit the build-up of oleic acid and therefore alter the fatty acid composition of the oil. Conversely, when seed filling occurs at low temperatures more polyunsaturated fatty acids are found. Similarly, linolenic acid content increases during exposure to low temperatures during seed filling (Table 4–5).

Decreasing rainfall (moisture stress) after flowering exacerbates the effects of high temperature on oil production.

Table 4–4. Effects of temperature and water stress applied from the end of flowering until maturity on yield, yield components and oil content in *B. napus*.

	DAY/NIGHT TEMPERATURE EFFECT (% RELATIVE TO THE IRRIGATED LOW TEMPERATURE RESULTS)			
	18°C/10°C		26°C/18°C	
	IRRIGATED	WATER STRESS	IRRIGATED	WATER STRESS
Seed yield	100	63	67	56
Pods/m^2	100	72	82	65
Seeds/m^2	100	84	83	72
Seed weight	100	76	81	77
Oil content	100	88	89	83

Source: Jensen et al. (1996)

- Even under well-watered conditions, heat decreased canola quality and yield.
- Increasing temperature decreased seed yield under well-watered conditions by 33%.
- With moisture stress, the reduction in yield was an additional 7%.

Table 4–5. Effect of temperature on oil content of the canola variety Westar.

TEMPERATURE (°C) NIGHT/DAY	12°C/17°C	17°C/22°C	22°C/27°C
Oil percentage	45	41	Not tested
Fatty acid content*			
C 16:0	3.7	4.0	5.2
C 18:1	65.2	62.9	57.2
C 18:2	17.4	18.6	25.0
C 18:3	9.2	9.6	6.8
C 22:1	0	0	0

* Data expressed as % of total. Source: Modified from Yaniv et al. (1991)

Frost

A frost during seed development can cause major losses. Even if the number of pods is already set, the developing seeds are very susceptible.

At a moisture content of 20% or more, seeds will suffer injury. The higher the moisture content, the greater the injury. At 50% to 60% moisture content a temperature of –3°C will kill a developing seed. Mature, dry seeds are safe from frost.

Frost stops seed development, reducing individual seed weight.

If all the seeds of a young pod are injured, the pod aborts. Damage from frost can be to a few seeds in the pod or the whole pod.

The other affect of frost is on the level of chlorophyll in the seed. Sub-lethal frosts (0°C to 1°C) disrupt the biological enzyme system that breaks down chlorophyll in the seed nearing maturity. Very cold temperatures can result in sudden plant death before the seed can dry down, resulting in green seeds with a high chlorophyll content.

Nutrition

Nitrogen

Increasing nitrogen supply increases the formation of protein, and hence growth. Too much nitrogen assimilation can result in overproduction of protein, causing a drop in oil content. High nitrogen applications can also cause lodging and delayed maturity. Lodging reduces seed yield by reducing movement of assimilates and moisture to the seed. It also decreases dry matter production post-lodging and reduces all major yield components.

In one study of the effects of nitrogen fertiliser on seed yield and oil concentration at Wellington and Parkes in NSW, nitrogen application increased seed yield at both sites (Figure 4–11) but had little effect on dry matter per pod, pod length, 1000-seed weight, seed number per pod, seed oil concentration and concentrations of mineral nutrients in the pod walls and seeds.

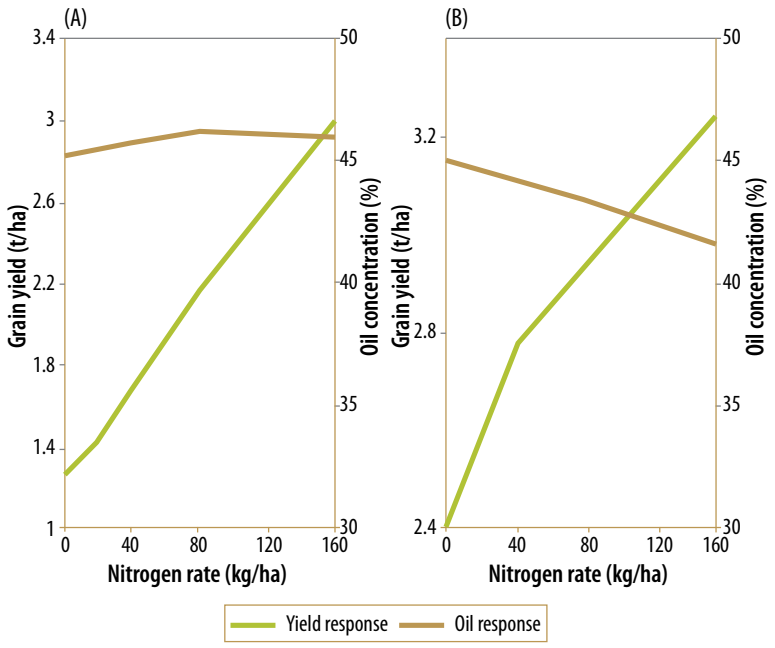
Sulfur

Studies in the late 1980s in south-east Australia showed yield losses of 80% due to sulfur deficiency, plus a reduction of 20% in seed oil concentration.

By podding, sulfur deficiency becomes distinctive. Pod number and size and seed number per pod are reduced significantly. Pods may be pale green, often with purpling, and can be compressed or flattened.

By maturity, canola straw contains approximately 0.3% to 0.4% sulfur. The pods contain slightly more (0.5% to 0.6%). Canola seed contains about 0.4% to 0.6% sulfur.

At harvest, canola straw and pods contain around twice as much sulfur per hectare as the seeds.



- Increasing rates of nitrogen have a greater impact on yield than on oil concentration.
- In (A), N fertiliser increased yield but had no effect on oil concentration.
- In (B), N increased yield, but at higher rates it decreased oil concentration.

Figure 4–11. Effects of nitrogen fertiliser on seed yield and oil concentration at (A) Wellington and (B) Parkes in NSW. Source: Salisbury et al. (1999)

Addition of sulfur can increase yields (Figure 4–12).

Sowing time

Delaying sowing of canola past the optimum sowing date for a variety reduces both seed yield and oil concentration.

Yield is strongly related to biomass at flowering. Later sowing shortens the vegetative phase of growth, reducing biomass at flowering. It also results in a shorter flowering period, particularly in southern NSW, where hot dry winds in October frequently end the flowering period.

When canola is sown later than optimum, the pods and seeds will develop under hotter and drier conditions, resulting in a shorter pod and seed development period.

Reported rates of yield decline per week attributed to delayed sowing have ranged from 3% to 11%. This equates to about 25 kg/ha for each day's delay in sowing.

In addition, canola oil content has been found to decline with later sowing. The rate of oil concentration decline will be between 0.5% and 0.75% per week of sowing delay. The reduction is due to increased temperature and water stress during grain filling.

Variety differences are greater for oil and protein content than for yield components. There is a strong genetic control of oil concentration. This means that a variety will have a similar ranking in a range of environments.

The extent of the reduction in oil concentration from delayed sowing is greater in low-rainfall areas. Oil concentrations tend to be lower from later-sown crops because the seed development stages occur during periods of high temperatures. In a study at Condobolin, the oil concentration from varieties sown on 14 June was significantly less than that from the earlier sowings (Table 4–6).

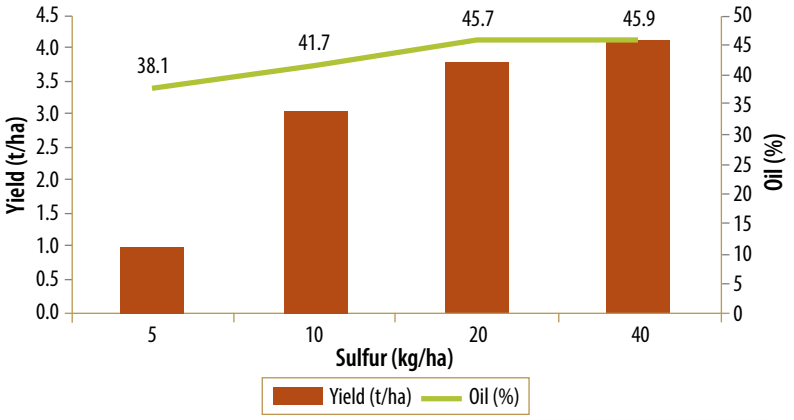
However, the date of flowering needs to be optimised between the risk of frost during flowering and of water and heat stress during seed filling.

Disease

The two main diseases of canola are blackleg and sclerotinia.

Blackleg

Blackleg is caused by the fungus *Leptosphaeria maculans*. It is the most common and serious disease of canola in NSW. The disease can be carried over from one season to the next by the



- Addition of 20 kg/ha of sulfur increased yield by 74% and oil percentage by 17%.

Figure 4–12. Canola response to sulfur applied at Wellington NSW in 1992. Source: J Sykes, NSW Agriculture and Fisheries extension brochure

Table 4–6. Main effects of sowing time on seed yield and oil concentration at Condobolin in 2002.

SOWING DATE	YIELD (T/HA)	OIL CONCENTRATION (%)
22 April	0.951	41.0
17 May	0.598	41.0
14 June	0.137	39.3

Source: MacKinnon and Fettell (2003).

- The oil concentration from a 14 June sowing was significantly less than for the earlier sowings.
- Yield fell by 0.8 t/ha with later sowing at Condobolin.
- Oil percentage also decreased.
- Higher temperatures also gave a smaller seed size from the June sowing.
- The ranking of different varieties for oil concentration, and seed yield to a lesser extent, remained constant across sowing dates and locations.

survival of the fungus on canola stubble. Blackleg can attack plants at all growth stages. Heavy infection can kill young seedlings or plants in the rosette stage. In older plants the infection can occur on the leaves, stems, flowers and developing pods. Older plants frequently become infected at the base of the stem, leading to the decay of the stem at ground level. This cuts off sap movement and either kills the plant or causes shrivelling of the seed. Damage is often not apparent until early spring. Growing resistant varieties is the best strategy.

Sclerotinia

Sclerotinia stem rot is caused by fungi of the *Sclerotinia* species. It causes stem rotting, with a white fungal growth appearing on the stem. Control is difficult, as virtually all broadleaf crops are affected and it survives for long periods in the soil. Cereals in a rotation aid in its control. The disease may affect the plant at the base of the stem or higher up. Wilting occurs above the point of the attack. In a severe attack the plant lodges.

Lodging aggravates the problem of uneven pod maturity and creates an ideal micro-environment for the spread of diseases such as sclerotinia and alternaria.

Disease reduces the photosynthetic capacity of the stems and pods, reducing yields.

Insects

Aphids (Figure 4–13) can attack during flowering or seed fill. Aphids suck the sap from the plant. High densities may prevent pod set or even kill plants. Aphids can also reduce the accumulation and movement of assimilates.

Grazing

Yield

The effect of grazing on yield depends on whether there has been a significant delay in flowering that has in turn delayed pod and seed fill to a hotter, drier time (see Chapter 3). Preliminary experiments (see Table 4–7) have shown no significant difference in yield or oil content between grazed and ungrazed canola.

Figure 4–13. Aphids on a canola flower head.
Photo: Jan Edwards

Table 4–7. Impact of grazing on yield and oil. Field trial at Ginninderra Experiment Station, Canberra, 2004–2005.

VARIETY	2004				2005			
	YIELD (T/HA)		OIL (%)		YIELD (T/HA)		OIL (%)	
	UNGRAZED	GRAZED	UNGRAZED	GRAZED	UNGRAZED	GRAZED	UNGRAZED	GRAZED
Spring type: Hyola 60	4.8	4.6	50.5	50.5	–	–	–	–
Winter type 1	4.1	4.3	47.9	46.1	2.63	2.10	45.9	48.8
Winter type 2	4.1	4.0	48.4	47.6	2.67	2.07	47.6	46.8
Winter type 3	–	–	–	–	2.82	2.27	43.5	45.3

Source: Kirkegaard (2007)

- In 2004 the spring-type canola yielded significantly more than the winter types.
- In 2005, yield and oil results were not significantly different between the ungrazed and grazed canola in either year.

Disease

Grazing canola increases the risk of blackleg (Tables 4–8 and 4–9). This risk is increased with varieties that have lower blackleg resistance ratings.

Table 4–8. Effect of the timing of grazing on yield and blackleg severity (% of stem cross-section infected) for cultivars sown at Young, NSW.

TREATMENT	ATR-BEACON		SKIPTON		46Y78		SURPASS501TT		COLUMBUS	
	YIELD (T/HA)	BLACKLEG (%)	YIELD (T/HA)	BLACKLEG (%)	YIELD (T/HA)	BLACKLEG (%)	YIELD (T/HA)	BLACKLEG (%)	YIELD (T/HA)	BLACKLEG (%)
Ungrazed	2.03	16	1.80	40	2.47	16	1.89	11	0.65	2
21/6–6/7	1.50	40	2.08	56	2.23	17	1.76	9	0.37	3
6/7–20/7	1.32	38	1.52	54	1.85	22	1.88	11	0.31	3
20/7–3/8	1.31	41	1.32	56	1.97	26	1.75	9	0.88	2
3/8–17/8	1.04	48	1.33	49	1.48	27	2.01	13	0.99	2
17/8–5/9	1.17	51	1.67	35	1.52	15	2.04	15	0.58	2
5/9–20/9	0.70	47	0.53	40	0.65	26	0.78	16	0.47	2

UG = ungrazed; other dates represent the 2-week periods in which the crop was grazed heavily by sheep. Columbus is a resistant winter canola.
Source: Kirkegaard (2007)

Table 4–9. Effect of the timing and duration of grazing on yield and blackleg in Thunder TT canola sown on 23 March 2007 at Galong, NSW

TREATMENT	GRAZING TIME	YIELD (T/HA)	HARVEST INDEX	BLACKLEG (% STEM)
Ungrazed	no grazing	1.12*	0.17	1.6
G1F1	28/5–1/8	1.06	0.30	20.3
G1F2	28/5–17/8	0.91	0.31	16.6
G2F1	26/6–1/8	0.88	0.29	24.7
G2F2	26/6–17/8	0.67	0.25	34.1

Start grazing times: G1, 6 to 8 leaves; G2, full cover
Finish grazing times: F1, 1/8; F2, 17/8; * Some bird damage
Source: Kirkegaard (2007)

Measuring crop performance

Yield

Yield is determined by three components:

- number of pods/m² (pod density)
- number of seeds per pod
- seed weight.

The yield components develop sequentially, although there is some overlap. The potential number of seeds is set well before flowering, the pod number at around flowering, and the seed weight between flowering and maturity. Seed weight is the least variable of the yield components, because it is largely determined by the genetic potential of the variety.

Pod density

Potential pod number is largely determined at the onset of flowering. Final pod density is controlled by assimilate availability both before and during flowering.

Total dry matter at the onset of flowering and end of flowering is related to the potential and actual pod density, respectively.

Growth before pod initiation influences the potential number of pods through its effect on the number of leaves, which influences the number of potential sites for flowering branches in the leaf axils.

Losses of pods and seeds per pod are higher in the more shaded, lower parts of the canopy, especially in dense crops. As branch order increases up the plant, fewer flowers form pods and the seed number per pod decreases. A dense canopy means more competition between pods. Pod and seed survival is greatest at the top of the main stem and upper branches, where there is better distribution of solar radiation.

Pod density and seed density reach their final values shortly after flowering. However, pods can also be aborted up to maturity in very dense crops because of shading and lack of assimilates from photosynthesis. This is because low light levels in the lower part of the canopy mean that there are not enough assimilates available to maintain the lower pods and seeds.

Pod numbers on all branches are increased by nitrogen application.

Number of seeds per pod

Position on the plant is an important determinant of pod and seed survival (Figure 4–14). The developmentally superior florets at the base of the terminal flower head and on the first four primary branches have a 2 to 3 week competitive advantage in obtaining water and assimilates over those that open during the latter part of the flowering period.

The potential number of seeds per pod ranges from 15–25, depending on the variety. Seed set is determined mainly by assimilate availability during flowering. Pollination and fertilisation generally do not limit the potential number of seeds per pod.

Seed growth in earlier pods competes with seeds developing in the later pods.

There is a correlation between seed density and total dry matter production during flowering and the length of the flowering period.

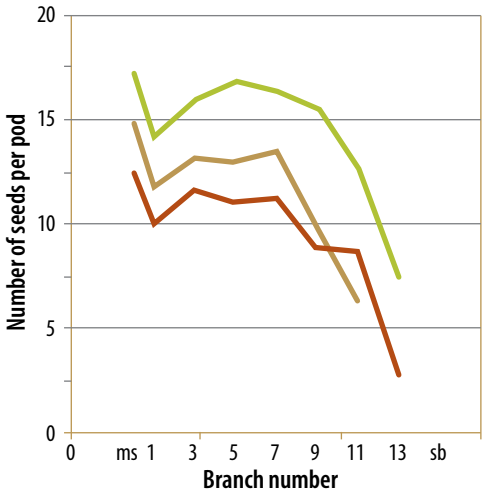


Figure 4–14. Number of seeds per pod as a function of the position of the pods on different branches. Treatments are: low plant density; high plant density; and shading from flowering to harvest. Source: Habekotte (1993)

- Seed set per pod varies with primary branch number.
- Only a small proportion of the pods (<10%) was situated on branches with below-average seed numbers.
- The smaller number of seeds per pod on branches of a higher order may be due to competition with the stem and earlier-formed pods for assimilates.

Seed weight

Seed growth rate is determined by assimilate availability during seed filling and the number of competing seeds. Remobilisation of reserve carbohydrates accumulated during earlier growth phases in the roots, stems, leaves and pod hulls may contribute to seed filling, but the remobilisation rate is only about 12% to 17.5%.

Seed size varies with the location of the seed in the pod and the pod on the plant.

Yield compensation

The ability of canola to compensate for low plant density is achieved mainly through an increase in pod numbers per plant. Conventional canola varieties can produce similar seed yields across a range of plant densities. Triazine-tolerant varieties are less able to compensate, because they have lower photosynthetic rates and slower growth.

Compensation depends on temperature and soil moisture conditions and the time the stress was applied. Restricted flowering followed by good conditions creates a crop with few pods but very large numbers of seeds per pod.

There is an inverse relationship between number of pods and seeds per pod. There is also a relationship between both of these factors and the size of the crop (i.e. the ability to support both factors). The number of seeds per pod increases with increased crop dry weight at flowering.

Harvest index

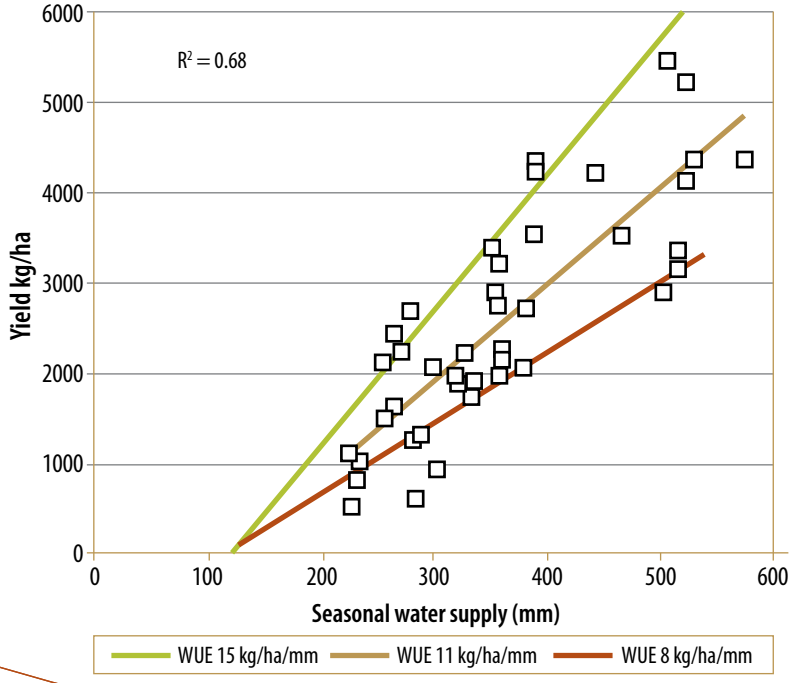
The harvest index is the proportion of above-ground dry matter that is seed. It is the ratio of seed dry weight to above-ground dry weight at harvest maturity. Canola harvest index is generally between 0.25 and 0.35. This is lower than in a wheat crop, but the canola seed also contains between 32% and 45% oil, which has 2.5 times the energy of the carbohydrate in cereal grains.

Moisture or temperature stress can substantially reduce the harvest index because of poor pod set.

Water use efficiency

Water use efficiency (WUE) is a measure of how efficiently crops have used available moisture (i.e. stored moisture as well as in-crop rainfall). It is also used to set a target yield based on available soil moisture and expected in-crop rainfall. See *In the paddock* in this chapter.

Canola plants require a threshold amount of water before any yield is obtained. Beyond that threshold, increasing amounts of water will result in higher yields. WUE is measured in kg seed/mm water. The WUE for canola is between 8 and 15 kg/ha/mm, depending on rainfall distribution (Figure 4–15). Early-sown crops will have a higher WUE than later sown crops.



- The water use efficiency (WUE) for potential canola seed yield is between 8 and 15 kg/ha/mm.
- The average WUE is 11 kg/ha/mm.
- The WUE of triazine-tolerant canola is lower (10 kg/ha/mm).
- WUE is estimated to fall by 10% for every month's delay in sowing after late April.

Figure 4–15. Range of WUE for canola, modified from the French and Shultz equation (see *In the Paddock: Calculating water use efficiency* in this chapter. Source: Robertson and Kirkegaard (2005)

References and further reading

Habekotte B 1993, Quantitative analysis of pod formation, seed set and seed filling in winter oilseed rape (*Brassica napus* L.) under field conditions. *Field Crops Research* 35, 21–33.

Hocking PH, Mason L 1993, Accumulation, distribution and redistribution of dry matter and mineral nutrients in fruits of canola (oilseed rape) and the effects of nitrogen fertiliser and windrowing. *Australian Journal Agricultural Research* 44, 1377–1388.

Hocking PJ, Stapper M 2001, Effect of sowing time and nitrogen fertiliser rate on growth, yield and nitrogen accumulation of canola, mustard and wheat. *Australian Journal of Agricultural Research* 52, 623–634.

Hocking PJ, Randall PJ, DeMarco D 1997, The response of dryland canola to nitrogen fertiliser: partitioning and mobilisation of dry matter and nitrogen, and nitrogen effects on yield components. *Field Crop Research* 54, 201–220.

Jensen CR, Mogensen VO, Mortensen G, Andersen MN, Schjoerring JK, Thage JH, Koribidis J 1996, Leaf photosynthesis and drought adaptation in field grown oilseed rape (*Brassica napus* L.). *Australian Journal of Plant Physiology* 23, 631–644.

King SP, Badger MR, Furbank RT 1998, CO_2 refixation characteristics of developing canola seeds and silique walls. *Australian Journal of Plant Physiology* 25, 377–386

Kirkegaard JA 2007, *Evaluating the Potential for Dual-purpose Canola in the Mixed Farming System of Southern Australia.* Report to Grains Research and Development Corporation on Project CSP00085. CSIRO, Canberra.

MacKinnon GC, Fettell NA 2003, The effect of sowing time, supplementary water and variety on yield and oil concentration of canola (*Brassica napus*). In *Proceedings of the 13th Biennial Australian Research Assembly on Brassicas*, 8–12 September 2003, Tamworth, NSW. NSW Agriculture, Orange NSW.

Norton G, Bilsborrow PE, Shipway PA 1991, Comparative physiology of divergent types of winter rapeseed. In McGregor DI (ed)), *Proceedings of the 8th International Rapeseed Congress,* Saskatoon, Canada, pp. 578–582.

Office of the Gene Technology Regulator 2008, *The Biology of Brassica napus L. (Canola)*. Version 2: February. Australian Government Department of Health and Ageing, Canberra.

Pritchard FM, Eagles HA, Norton RM, Salisbury PA, Nicolas M 2000, Environmental effects on seed composition of Victorian canola. *Australian Journal of Experimental Agriculture* 40, 679–685.

Robertson MJ, Kirkegaard JA 2005, Water use efficiency of dryland canola in an equi-seasonal rainfall environment. *Australian Journal of Agricultural Research* 56, 1373–1386.

Salisbury PA, Potter TD, McDonald G, Green AG 1999, Canola in Australia: the first 30 years. In *Proceedings of the 10th International Rapeseed Congress,* Canberra.

Si P, Walton GH 2004, Determinants of oil concentration and seed yield in canola and Indian mustard in the lower rainfall areas of Western Australia. *Australian Journal of Agricultural Research* 55, 367–377.

Walton G, Mendham N, Robertson M, Potter T 1999, Phenology, physiology and agronomy. In Salisbury PA, Potter TD, McDonald G, Green AG (eds) In *Canola in Australia: the First 30 Years.* Proceedings of the 10th International Rapeseed Congress, Canberra.

Yaniv Z, Elber Y, Schafferman D, Zus M 1991, The effect of temperature on the fatty acid composition of high and low erucic acid rape cultivars. In McGregor DI (ed), *Rapeseed in a Changing World.* Proceedings of the GCIRC Rapeseed Congress 1991, July 9–11, Saskatoon, Saskatchewan Canada. Vol. 6 of 6, pp. 1821–1825.

IN THE PADDOCK

The following are some examples of activities that can be done in the paddock to illustrate the stages of seed and pod development that have just been discussed. These are practical exercises to help farmers assess the progress of their crops at this stage.

Pods

Aim: to assess the stages of pod development. This stage overlaps with flowering.

1. **Start of pod development.** Pods start to develop at the bottom of the flowering spike following fertilisation.
2. Identify whether **frosted pods** are present: they will be a different colour or may have not developed. Compare frosted with healthy pods.
3. **End of pod development.** Pod development is finished when the seeds begin to show a change in colour from a darker to a paler green. Seeds are considered to have reached physiological maturity at this stage.
4. **Seed development.** As seeds mature, their colour changes from green to light bronze to black. Moisture content slowly decreases during these changes.

Determining the timing of windrowing

Aim: to determine the amount of colour change in the seeds

1. Collect 10 pods randomly from the top, middle and bottom podded branches of the canopy.
2. Keep the pods from each section of the canopy separate.
3. Carefully open pods and empty the seeds into a white container. Look for the change in colour of the seeds.
4. The crop is ready to windrow when about 50% of the seeds are brown/black.
5. Compare the seeds from each location in the canopy.
6. Windrowing can be calculated from the date when 10% of plants have flowers remaining.

Estimating yield

Aim: to estimate yield in the paddock.

1. Count the number of pods per 0.5 m of row.
2. Do this at 10 locations across the paddock.
3. Add the 10 counts together and divide by 5 to get the average number of pods per metre of row.
4. Multiply the pod counts by the row-spacing factor:

17.5 cm = 5.71	25.0 cm = 4.00	33.0 cm = 3.03
20.0 cm = 5.00	27.5 cm = 3.36	36.0 cm = 2.77
22.5 cm = 4.44	30.0 cm = 3.33	40.0 cm = 2.50

5. Count the seeds in at least 10 pods and calculate the average number of seeds per pods. Count a range of seed sizes to get an accurate estimate.
6. Use an estimate of seed weight of between 0.030 and 0.045 g. Seed weights will vary between seasons. Use 0.030 g if you think the seeds are small or 0.045 g if the season has been good and the seeds are larger.
7. Estimate the yield using the calculation below:

$$\text{Yield (t/ha)} = \frac{\text{Pods/m}^2 \times \text{number of seeds/pod} \times \text{estimated seed weight (g)}}{100}$$

IN THE PADDOCK

COUNT	NUMBER OF PODS/M²	NUMBER OF SEEDS PER POD	ESTIMATED SEED WEIGHT	YIELD (T/HA)
1				
2				
3				
4				
5				
Average				

Assessing seed-size variation

Aim: to assess seed-size variation from pods in different parts of the canopy.

1. Collect 10 canola plants across a paddock.
2. Collect 10 pods from the top branches.
3. Pull the pods apart and place the seeds from each into a dish.
4. Then repeat the process with 10 pods each from the middle and lower branches.
5. Weigh the different dishes to compare the variation in seed size.
6. Repeat the exercise, this time collecting pods from the bottom branches.

Calculating harvest index

Aim: To calculate harvest index

Harvest index is the ratio of grain dry weight to above-ground dry weight at harvest maturity.

$$\text{Harvest index} = \frac{\text{seed dry weight (kg/ha)}}{\text{above-ground dry weight (kg/ha)}}$$

	PADDOCK 1	PADDOCK 2	PADDOCK 3
Seed dry weight (kg/ha)			
Above-ground dry weight (kg/ha)			
Harvest index			

Calculating water use efficiency

Aim: to use rainfall records to estimate the water use efficiency (WUE) of a crop.

This method is a modification of the French and Shultz method used for cereals. It was developed by Robertson and Kirkegaard in 2005.

1. Using your rainfall records, record the amount of fallow rainfall. This is used to calculate the soil water at sowing. It is assumed that 50% of the rainfall during the fallow will be stored in the soil and available to the crop. This will vary depending on stubble cover, weed growth and rainfall pattern.

   Soil water at sowing = (fallow rainfall – 80 mm) × 0.5

2. Record the amount of in-crop rainfall up until the date of windrowing (or estimated date of windrowing for direct-headed crops). For the equation to work, this should be less than 450 mm.

IN THE PADDOCK

3. Calculate the amount of soil water left at harvest.

   Soil water at harvest = (post-flowering rain – 50) × 0.5

4. The seasonal water supply (the amount of water that was available to the crop) can be calculated. One hundred and twenty millimetres is subtracted from the total available water to account for the water required by the crop to grow the biomass and to account for losses from run-off and evaporation. The formula is:

   Seasonal water supply = (in-crop rainfall) + (soil water at sowing) – (soil water at harvest) – 120

5. Using the seasonal water supply and the yield, calculate WUE.

   $$\text{WUE (kg/mm/ha)} = \frac{\text{crop yield (kg/ha)}}{\text{seasonal water supply (mm)}}$$

	PADDOCK 1	PADDOCK 2	PADDOCK 3
Fallow rainfall			
In-crop rainfall			
Total water supply			
Yield (t/ha)			
WUE (kg//ha/mm)			

Glossary

Adequate soil moisture

Defined as maintaining 50% or more of the available soil moisture in the root zone.

Anthesis

Flowering; the moment when pollen is released from the anthers (the pollen-producing top parts of the male reproductive organ).

Apical meristem

Growing point; a zone of cell division at the tip of the stem or root.

Assimilates

Products of photosynthesis.

Chlorophyll

Pigment responsible for capturing sunlight energy.

Embryo

The part of the seed that contains the main plant structures. It is made up of the scutellum, plumule and radicle.

Endosperm

A nutritive tissue within the seed that surrounds the embryo and provides energy for germination.

Fertilisation

The union of the male genetic material from the pollen with the female material in the ovule.

Floret

One of the small individual flowers within a dense cluster of flowers.

Glucosinolates

The main glucosinolate in canola roots is gluconasturtiin, whereas shoots contain mostly aliphatic glucosinolates. As gluconasturtiin degrades, a compound referred to as PEITC is released. This compound is toxic to a range of soil-borne fungal pathogens. This is referred to as biofumigation.

Indeterminate growth

A plant has indeterminate growth if it grows and produces flowers and fruit until killed by frost or some other external factor.

Internode

Area of stalk between two nodes.

Leaf area index (LAI)

A measure of the upper surface area of leaves of a crop in comparison with the ground surface area.

Node

Stalk region slightly larger than that of the adjacent stalk internodes. Vascular bundles cross-connect at nodes, but they are separate within internodes.

Ovule

The part of the plant that, after fertilisation, develops into the seed.

Osmosis

The diffusion of water through a semi-permeable membrane, from a solution of low solute concentration (high water potential) to a solution with high solute concentration (low water potential).

Pericarp

The outer covering of the seed. Pericarp thickness varies across hybrids and contributes to differences in field dry-down rates.

Plant physiology

A botanical term describing the functioning (physiology) of plants.

Plumule

Growing point of the seed that develops into the shoot bearing the first true leaves.

Pollen

The male gametes, which are produced in the stamens.

Pollen tube

A tube that acts as a conduit to transport sperm cells in the pollen grain from the stigma (pollen receptor) to the ovules at the base of the pistil (female reproductive organ).

Pollination

The transfer of pollen from an anther (the male reproductive organ) to a stigma (the receptive part of the female reproductive organ).

Primordia

Organs in their earliest stage of development.

Raceme

An unbranched flowerhead, with flower buds connected by short stalks called pedicels. This creates a bunch of flowers at the end of the stem.

Radicle

The part of a plant embryo that develops into the primary root.

Relative growth rate

The rate of dry matter accumulation per unit of existing dry matter.

Scutellum

A shield-shaped structure in a seed that absorbs the soluble sugars from the breakdown of starch in the endosperm.

Seed meal

The part of the seed remaining after the oil is removed. It contains proteins, carbohydrates, minerals and fibre.

Secondary dormancy

Dormancy induced by exposure to temperature fluctuations, low moisture conditions or low oxygen.

Soil water potential

A measure of the energy needed to move water from the soil.

Stamen

The structure in a flower that produces pollen grains; consists of a stalk (filament) and an anther.

Turgor

The force exerted outwards on a plant cell wall by the water contained in the cell.

Vernalisation

A cold requirement that triggers the switch from vegetative growth to reproductive growth.

Winter feed gap

The period in early winter before pasture growth can match animal needs.