Wheat Special Report No. 32

Guide to Plant and Crop Sampling: Measurements and Observations for Agronomic and Physiological Research in Small Grain Cereals

M.A. Bell, Experiment Stations, and
R.A. Fischer, Wheat Program, CIMMYT

September 1994

Correct citation: Bell, M.A., and R.A. Fischer. 1994. Guide to Plant and Crop Sampling: Measurements and Observations for Agronomic and Physiological Research in Small Grain Cereals. Wheat Special Report No. 32. Mexico, D.F.: CIMMYT.

ISSN: 0187-7787
ISBN: 968-6923-25-X
AGROVOC descriptors: Cereal crops, plant developmental stages, plant physiology, agronomic characters, yield components, measurement, sampling
AGRIS subject codes: F60; F62
Dewey decimal classification: 633.1

Contents

Preface

This wheat special report is a guide that outlines the procedures used for measuring many components of the crop in small grains research. The actual observations taken, however, will depend on the objectives of the work in question.

To assist with assessing the relevance of an observation, a small discussion, where applicable, is presented under each section outlining the value or application of that particular observation. Some key references are included in each section to help with interpretation or application of the data collected.

This guide is a companion piece to Wheat Special Report No. 18, Guide to Soil Measurements for Agronomic and Physiological Research in Small Grain Cereals.

Acknowledgments

We would like to give special thanks to Dr. P. Wall for his thorough revision of the manual, and G.P. Hettel for editorial assistance. In addition, we wish to thank E. Acevedo, P. Hobbs, C. Meisner, I. Ortiz-Monasterio, M. Reynolds, K. Sayre, and D. Tanner for their input.

1 Bias and Sampling Techniques

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1.1 Bias

Cause: Bias refers to the systematic (i.e., nonrandom) error in results (e.g., always sampling from the best part of the plot).

Sampling bias can arise if there are consistent gradients along plots (e.g., due to direction of seeding or irrigation) and if samples are drawn from a given fixed location in each plot (i.e., a location that does not represent the whole plot). As a consequence of differences within a plot or field, any gradient must be noted. A degree of informed judgement is required to assess whether the differences that occur within a plot are a part of natural variation arising independently of the treatment (e.g., rodent damage in a fertility trial is not due to the treatment).

Avoidance: A common source of observer bias can, in part, be avoided in experiments if observation is conducted without reference to the treatments (i.e., no reference to the treatments while taking notes). For example, when evaluating a herbicide trial, a field plan that shows plot number, but not treatment should be used. By avoiding reference to treatments, the researcher can avoid biasing scores or sample selection (i.e., avoid giving a better score to a "favorite" treatment).

The guidelines outlined below provide further information for avoiding bias.

1.2 Plant selection

When you wish to select plants at random, you should select plants from the base and not from the top or spike. This will ensure less bias against selection of larger or taller plants or shoots. Even better control against bias is provided by counting along the row and selecting the plant occupying a given predetermined position (e.g., 10th).

If whole plants are to be sampled, the whole plant should be pulled or dug out of the ground and checked to see that it is only one plant and that it has all of its tillers.

1.3 Selecting areas of crops for sampling

Selection of quadrats or rows for sampling should be at random. For example, a predetermined number of steps should be taken into the field or plot. The quadrat should then be placed or the row

selected without visual assessment (close your eyes before selecting the sampling spot). Once selected, however, the sample area may at times be rejected if it is *very* obviously not representative of the field or plot.

1.3.1 Eliminating borders—It is usually worthwhile to avoid edge effects in any plot measurement (i.e., the extra time is worth the precision gained in estimating the population mean). Border effects are often very obvious under lower fertility or water supply, but are also present under optimal conditions because border plants receive extra light. Under the latter conditions, discarding the outside row (for 15-cm spacing or wider) and the end 50 cm of the plot is adequate. When there is soil stress, however, more border (minimum of 50 cm on all sides) should be discarded, especially with barley. Sometimes plot end bias can arise because plant density is greater or lesser then in the rest of the plot due to a faulty seeding technique.

1.3.2 Destructive sampling—Where consecutive destructive sampling is to be made during the life of the crop, the most correct and least biased way to do this is to choose the sampling position at random for each plot on each sampling date. This is, however, unnecessarily tedious and most workers use a systematic location of sampling combined with use of the same sampling position in all plots of a given block or replicate on a given date.

The simplest systematic system is to begin at one end of the plot on the first date and move steadily down the plot date by date, leaving an adequate buffer area between adjacent positions (40-60 cm). To avoid bias, the sampling positions should at least be different in different blocks or replicates, something most easily achieved by reversing the direction of sampling along the plot in adjacent blocks. This has the advantage of helping to balance out any linear trends there may be within the experimental area. The buffer area between consecutive quadrat cuts should be 40-60 cm depending on time between samples and sun angles (e.g., when plants are young, less distance between samples may suffice), and sun angle needs to be considered so that the next sampling area does not receive more light than the rest of the plot due to the removal of the previous sample.

1.3.3 Quadrat design—For row crops, a straight rod of wood or metal of known length can be used (e.g., 2 or 3 m). More accurate is an open square or rectangular metal quadrat (i.e., a "⊔" shape with one end open), which can be pushed at ground level perpendicular to the direction of the rows in the area to be sampled; the arms of the open quadrat must be rigid. (See sections 1.4.1-1.4.2 for sampling area recommendations.)

1.4 Representative sampling

For reasonably accurate work, measurements in a plot should be made across all those rows, which at maturity will be harvested for yield. This is again to avoid bias because there are often systematic differences in plant density between rows, again due to inefficiencies in seeding machinery (e.g., if the machine is not level in all directions, some rows will be seeded more deeply than others and may have less plants or tillers). In typical plots of six or eight rows in width, in which four or six inner rows will be harvested, plant counting and growth sampling should include all these inner rows. Thus, destructive growth sampling is usually most efficient if it runs across the plot occupying a given length of the four to six rows (e.g., 50 cm x 4 or 6 rows, giving an area of 0.4-0.6 m^2/plot). Plant

counting, which is usually nondestructive, can comprise 50 or 100 cm of each of the inner rows, selecting the counting area at random along each row. Easier still, row position can be selected approximately along the plot diagonal.

1.4.1 Recommended sampling sizes—A common concept is that as the yield potential increases, the variability decreases, and thus the required number of samples to estimate yield decreases. However, this is actually untrue (Barreto, pers. comm.; Bell, unpublished data).

1.4.1.1 Plot sampling. The best sample size depends on the number of replicates, the variable studied, variability within the plot, and the degree of accuracy desired. Generally, however, if four replications are sampled, then 0.5 m^2/plot for growth measurements, and 1 m^2/plot for yield would be a minimum for reasonable accuracy.

1.4.1.2 Field sampling. When sampling a field (up to 5 ha), the field should first be divided into sampling units (i.e., those areas of the field considered relatively uniform). Within each sampling unit, five x 1 m^2 samples taken at random should be adequate to estimate yield.

When obvious variation is apparent across a field, then these sampling units should be stratified in order to equally represent the variation (i.e., if 75% of a field or plot is apparently poorer then the other 25%, then 75% of the final yield estimate should be based on yield of that poorer area).

A further factor to consider in sample size is whether the field or plot is broadcast or drill-seeded. Drill-seeded fields or plots are generally more uniform, have less variation, and therefore may require fewer subsamples than a broadcast field.

1.4.2 Sampling broadcast versus row-planted plots or fields—Placement of a quadrat in a broadcast field can be done at random without concern for orientation of the sample. However, in row-planted crops, a source of error can sometimes be introduced by sampling within the quadrat area without reference to the row spacing of the crop. For example in **Figure 1**, the quadrat sample would give an artificially high (biased) estimate of say plant population.

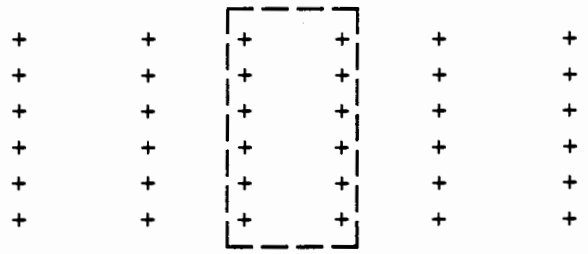

Figure 1. If plant population in this row planted plot is calculated using the quadrat area basis, then the results would be an overestimate of true population. The sampling width should be an exact multiple of the distance between rows. Row spacing is best ascertained by measuring the distance from a row to the nth (say 11th) row and then dividing by (n-1), i.e., in this case 10.

Quadrat sampling in both broadcast and row-planted fields may have problems especially if the crop is leaning or lodged. When closed quadrats are used, it is almost impossible to avoid bias. A straight line opening needs to be made at random in the crop at ground level and this is then used as the starting point for the area to be sampled. An open ended quadrat is then pushed forward a known distance at 90° to the straight line. Only plants with crowns in the quadrat area are sampled.

For row-planted crops, the row spacing of the quadrat area should always be determined. To do this, measure the distance between the two closest unsampled rows on each side of the sampled area (x). See example below. Next, count the number of rows in the sampled area (y). The effective width of the sampled area is then:

$$y^*[x/(y+1)].$$

Example. A plot consists of 8 rows planted uniformly at a 30-cm spacing and the four inner rows are to be sampled. Then, x = 1.5 m (distance from row 2 to row 7), and y = 4. The width of the sample is thus 4*[1.5/(4+1)] = 4*0.30 = 1.2 m.

1.5 Accuracy

The accuracy of the mean value of any measurement (including those for plant and soil) is a function of:

- The accuracy of each individual measurement, and
- The number of replications—in a plot trial particularly the number of blocks, but the number of samples or measurements per plot as well.

Simple statistics reveal the size of the two errors involved in experimentation (i.e., between plots and within plots combined with the measurement per se) and the optimum sampling strategy, which is a function of variability and sampling cost between and within plots (See Snedecor 1956, pp. 512-519).

Usually, accuracy for growth and yield measurements is optimized by maximizing blocks or replications, with only one measurement or quadrat per plot, provided an area of 1 m^2 or more is sampled (less may suffice for growth measurements; see section 1.4).

The level of accuracy required is a function of the differences one wishes to detect; resources are wasted with both too little accuracy and too much.

Research agronomists should strive for standard errors (SE; i.e., $(S^2/n)^{1/2}$, where S^2 is the error mean square (or variance) and n the number of replications) in growth and yield parameters of less than 5% of the trial mean; this ensures that at $P<0.05$, differences of 15% or more of the mean will be significant since the relevant LSD is approximately 3 times the SE of the mean (See example).

Example

1. Using coefficient of variation:

If CV is the coefficient of variation in % (CV = (S/trial mean) x 100), then the standard error $(S^2/n)^{1/2}$ expressed in % terms is given by $(CV^2/n)^{1/2}$. Thus, for the precision we need (i.e., SE<5% trial mean ($\bar{x}$)) (note: trial mean terms cancel out).

$(CV^2/n)^{1/2} < 5$

If CV = 10%, a typical value for well managed trials, then

$(10^2/n)^{1/2} < 5$

$<=> n > 10^2/5^2$

$<=> n > 100/25$

$<=> n > 4$

However, if CV = 7.5, then

$n > 7.5^2/5^2$

$<=> n > 2.25$

2. Using trial mean:

From experience, a mean trial yield of 3 t/ha is expected, and you wish to detect significant differences of as low as 15% of site mean yield (i.e., 0.45 t/ha).

Then, 5% of 3 t = 0.15 t.

This means that we want the standard error less than 0.15 t

or $(s^2/n)^{1/2} < 0.15$ t

$<=> s^2/n < (0.15)^2 t^2$

$<=> s^2/n < 0.0225. t^2$

If an error variance (s^2) of 0.06 is expected (s = 0.245),

then $n > 0.06/0.0225$

$<=> n > 2.67$, or

a minimum of 3 replications is required.

Note: When considering variance between reps (S_1) and variance within plots (S_2), the total variance (S^2) can be described as:

$S^2 = (S_1)^2/n_1 + (S_2)^2/(n_1n_2)$, where:

1 refers to the variance of n samples between replicates, and

2 refers to the variance of n subsamples within plots.

Standard error of the mean (SE) is therefore equal to:

$SE = [(S_1)^2/n_1 + (S_2)^2/(n_1n_2)]^{1/2}$

Thus, if $(S_1)^2 >$ or = $(S_2)^2$, which is common in field experiments and n_1 and n_2 constant, the SE gets smaller if n_1 increased at the expense of n_2. However, often a sample within plot costs less than a plot, so the optimal solution is not so straight forward as maximizing n_1 (See Snecdor, 1956 5th edition - sampling in two stages, pp. 512-519).

For detecting differences at a given site in on-farm research, usually from 12-20 degrees of freedom for the error represents the most efficient use of resources (this is because the change in the tabulated F value is little improved by further increases). Therefore, in a trial with 10 treatments, two replications would give only 10 degrees of freedom for the error, whereas three replicates would give 19. Therefore, further replication would be unnecessary for improving the estimate of S^2, although more replications may improve the estimate of the standard error of the mean. For on-station trials, an increase by one in replication, in comparison to on-farm trials, may further be justified, as a noticeable decrease in the SE may still be attained.

2 Crop Development Observations and Measurements

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2.1 Development stages (sometimes loosely called growth stages)

Development is sometimes referred to as growth stages, although this is a misnomer. Development refers to the timing of key events in the morphogenesis of the crop. Allocation of dry matter to different organs is closely related to these events. For example, the spike commences growing (albeit very slowly) with the onset of floral initiation at the vegetative apex. Another example, grain growth

begins soon after flowering (or anthesis). The response of the crop to external environmental factors, such as management events or stresses can be more easily understood if the timing of such events is related to crop development; indeed certain management activities are more efficient if carried out at given stages of development.

2.1.1 Growth versus development—Growth is the enlargement of an existing organ (e.g., the expansion of a leaf), whereas development involves a change in the meristem of the plant, the timing of such events, and often is only subsequently revealed by the changes in appearance of an organ.

2.1.2 Development scales—Many different scales exist for the evaluation of plant development stages. The decimal code and Feekes growth scales, which allow the development stage to be determined both quickly and nondestructively are described in **Appendices 1 and 2**. The decimal code (DC), also known as Zadoks scale (Z) or development stage (DS), is more commonly used to assess development (Zadoks et al. 1974, Bauer et al. 1983).

2.1.3 Plants to sample—In general, two to three plants per plot are sufficient to assess the development of a crop within a plot (for a given treatment), assuming that there are three or four replicate blocks (thus 6-12 plants for a given treatment). When assessing grain stage, spike-bearing culms should be sampled at random. The spikelets in the middle of the spike mature first. The key point is that sampling is conducted in a consistent (hence repeatable) manner preferably by the same person.

2.1.4 Sampling with time—Fortunately, development within an experiment is fairly consistent for a given treatment (measuring 8-10 plants is enough to give an accurate mean, i.e., two plants in each of four replicates).

For greatest accuracy with respect to changes with time, random but typical plants can be marked with small stakes or flags, and the observer can score nondestructively these same plants at each date the plots are visited. Marking leaves of the main shoot (e.g., leaf 5, leaf 8) with paint or a marking pen enables rapid assessment of leaf stage. Determination of the stage of the apical development on the main shoot (e.g., Kirby and Appleyard 1981) is destructive and requires sampling of representative plants (e.g., Klepper et al. 1982), as does determination of the exact pattern of tillering (See section 3.4 and Appendix 2 for discussion on tillering).

2.2 Key development stages

The key externally-visible development stages are:

- Seedling emergence,
- First node,
- Flag leaf emergence,
- Spike emergence,
- Anthesis,
- Physiological maturity.

Counting emerged leaves on the main stem is another useful guide to development (although in older plants, caution is required as early leaves may be lost or later ones confused with tiller leaves).

The accurate determination of growth stages requires frequent visits (1-2 times weekly depending on temperature). In remote sites or at busy times when this may not be possible, careful determination of development stage whenever visits can be made can permit determination of key stages by interpolation. Methods of assessing growth stages are outlined in sections. 2.2.1-2.2.8

Information gained:
Correct evaluation of the development stage is vital to understand both the internal and external developmental changes of the plant. For example, environmental conditions at the time of flowering can affect seed set. Irregularities in development across a field or plot should be noted, as this may provide information on management problems. For example, differences in development that follow a linear gradient across a field may be due to differences in depth of sowing. In this respect, tillering pattern (more a growth measure than a developmental one) is especially useful as a record of the stress history of the crop (Klepper et al. 1982). If for example, the first tiller (T1) (see section 3.4) is absent, then it implies stress during the 1-3 leaf stage.

Development is primarily driven by heat units (Heat unit = day degree), photoperiod, and amount of vernalizing cold (especially in winter wheats). The response of rate of development to heat units is approximately linear above a minimum base temperature (T_b) (usually 0°C) up to a maximum mean daily temperature of about 25°C. This means that, other things being equal, the day degree sum to complete a given period of development is constant (i.e., Day degree = k ($T-T_b$), where k = a constant for a given variety). The effect of photoperiod is such that a longer photoperiod accelerates development up until flowering, thus the day degree sum is less than otherwise expected. In vernalization-sensitive cultivars, early development (up to terminal spikelet) is accelerated by periods of temperature below about 10°C (therefore daily minimum temperature is important). After vernalization or in vernalization-insensitive varieties, even small differences in the heat units received lead to changes in development rates. For example, N deficiency results in increased light penetration into the crop, and thus being warmer, development is quicker; this is a common observation in check plots of fertility trials (Seligman et al. 1983).

2.2.1 Date of seedling emergence (Zadoks scale DC10)—Date of emergence is the date when 50% of the seedlings have emerged—emergence being the appearance of the first leaf lamina breaking through the soil surface. A visual estimate is usually adequate, as seedling emergence is usually fairly uniform. However, when more accurate data are required, such as in a depth of seeding trial, at least 2 x 1-m lengths of rows (or two quadrats) should be marked out in each plot. Daily counts of emerged plants are made until the number of emerged plants is constant. The date of 50% emergence is then the date at which half of the final number of plants had emerged. Use the mean of the two sample areas, and record as days after seeding.

Information gained:
When related to the date of seeding, date of emergence can be used to help interpret the effects on emergence of seed vigor, depth of planting, moisture, and/or temperature.

2.2.2 Date of first node at 1 cm (Zadoks scale DC 31)—This is estimated as the date at which the first node can be detected at approximately 1 cm above ground level. This stage is not to be confused with the stage "epi 1 cm" used by French researchers at which the top of the spike is 1 cm above the base of the crown (approximately DC30 = onset of stem elongation). Measure on 8-10 plants (or 2 plants/plot across 4 replications).

Information gained:
Spike at 1 cm above the crown roughly corresponds to the end of tiller initiation and the formation of the terminal spikelet. With production of the terminal spikelet, the maximum potential number of spikelets has been determined. In addition, for a given variety, this stage can be used to assess the effect of heat units affecting development.

2.2.3 Leaf emergence—The number of leaves usually refers to the number of leaves on the main stem only (leaves on tillers are normally not counted). Those leaves showing their ligule are considered fully emerged. The fraction emerged of the next leaf (relative to its final lamina length) is then recorded. Therefore, if a plant has three fully expanded leaves and one other that has half emerged, it would be recorded as 3.5. The Zadoks scale (Appendix 1) classifies development based on 50% of a leaf being visible, but originally did not consider fractions of leaves. When two emerging leaves are visible, the rating given for development will therefore depend on the extent of emergence of the earlier leaf. Instead of counting fully emerged leaves as in the Zadoks scale, some workers count visible tips.

The number must be determined with care as the early leaves usually senesce early (the first leaf can be recognized by its unique boat-shaped tip). Consequently, it is best to put some form of permanent mark on a reference leaf (e.g., leaf number 5) to facilitate leaf number determination, if repeated observations are being made on the same leaf. Measure on 8-10 plants (or two plants/plot across four replications).

Information gained:
Although not truly a development stage, leaf number, particularly when recorded over time, can be a useful indicator of the rate of development. Generally, spring wheats have 7-9 leaves on the main stem and the first tiller will usually emerge when the plant has three leaves. For a given variety and sowing date, leaf number is a good guide for the occurrence of key events in the developing shoot apex—such information is often used in extension bulletins to help farmers better program field activities (e.g., optimum time for herbicide applications for a given variety).

2.2.4 Flag leaf emergence (Zadoks scale DC39)—Flag leaf emergence is defined when 50% of those culms expected to produce spikes have fully emerged flag leaves (flag leaf ligule is visible). Measure on 8-10 plants (or two plants across four replications).

Information gained:
This stage coincides approximately to the onset of rapid accumulation of spike dry matter, the period when grain or kernel number/m^2 is largely defined. Also, it comes just after the period of meiosis in florets, a period of special sensitivity to stress (Gusta and Chen 1987).

2.2.5 Date of heading or ear emergence (Zadoks scale DC 55)—Heading is defined in the Zadoks scale as that stage at which 50% of the spike is emerged (i.e., middle of spike at flag leaf ligule on 50% of the culms). However, we prefer to define heading as the date or number of days from sowing when the base of 50% of the ears have emerged from the flag leaf (i.e., the base of the ears is above the ligule of the flag leaf) - this is equivalent to DC 60 (full heading). Although often recorded, date of heading has less application physiologically than date of anthesis which usually follows heading, but under drought or high temperature may occur at almost the same time.

Measure as follows:

- Visual assessment has a certain amount of variation. It is therefore suggested that the same person judge heading for all treatments within a trial.

- More accurate assessments can be made by counting—for a total of 50 culms selected in several random clumps within a plot—the number with the base spikelet above the ligule of the flag leaf.

Information gained:
For a given variety can be used to assess the effect of heat units affecting development.

By heading, approximately fifty percent or more of the potential number of florets will have been aborted and the number of fully formed (competent) florets will have almost been determined.

2.2.6 Date of anthesis (Zadoks scale DC65)—Anthesis is the date from sowing when 50% of the spikes have extruded at least one anther; it can be assessed by either of the methods outlined above for estimating heading.

Note: Anthers first appear from florets in the middle of the ear, and are then extruded both up and down from the center. Anthesis is usually complete for an individual spike within 2 or 3 days after its initiation.

Durum wheats, barleys, and heat-stressed bread wheats, in particular, may not show extruded anthers—especially under water stress when pollination can occur with the spike still in the boot. Basal florets of the central spikelet must be opened in order to reveal anthesis. In these circumstances, anthesis has occurred if the anthers have a yellow color and are no longer below the stigma. Another guide for the occurrence of anthesis is evidence of fertilization (withering of the stigma and growth of the carpel).

Pollination of the ovule has already occurred by the time dehisced anthers are extruded. Pollination is followed by obvious withering of the stigma. The date of anthesis can be estimated retrospectively with reasonable accuracy by assuming that the developing grain takes from 7-10 days after pollination to reach its full final length.

Information gained:
For a given variety, date of anthesis can be used to assess the effect of heat units affecting development. For adaptation purposes, there is usually an optimum date of anthesis at any location. See Fischer (1985b) and Information gained section in 11.4.2. Thus, when anthesis occurs relative to sowing date, it is an important aspect of varietal suitability. Anthesis marks the initiation of grain growth. Grains attain their maximum length within 7-10 days after anthesis; this process is essentially independent of environmental temperature conditions. Growth is essentially linear, so if grains are at approximately half their final length, then anthesis would have occurred about 5 days previously under normal mean temperatures (15-20°C). Attainment of full length is followed by the period of linear dry weight increase.

2.2.7 Grain development (Zadoks scale DC70-DC87)—Grain development passes through water, milk, soft, and hard dough stages. See Appendix 1 for method of determination. Hard dough (DC87) corresponds to the attainment of maximum dry weight.

2.2.8 Physiological maturity (Zadoks scale DC86)—Physiological maturity (PM) is measured as the date when 50% of the peduncles (i.e., the part of the stem immediately below the spike) are ripe (yellow); at this stage, glumes (which are the last part of the plant to lose their green color) will be losing their color as well. It can be assessed by either of the sampling methods outlined above for estimating heading. Data, as for other stages, are usually presented as the number of days from sowing or from seedling emergence.

Information gained:
For a given variety, PM can be used to assess the effect of heat units affecting development and, like anthesis date, can be related to meteorological data and/or cropping system patterns to assess the suitability of a variety for a region. As defined, PM is usually reached within a few days of the cessation of dry matter accumulation in grains; it also corresponds to DC86 falling between soft dough (DC85) and hard dough (DC87).

Once PM is attained, hand-harvested samples can be taken to estimate yield. Grain moisture will be about 30%—as moisture levels drop, shattering loses with hand harvesting generally increase. At times, mechanical harvesting may cause grain damage (primarily crushed grain) when grain is harvested at high moisture contents.

3 Crop Growth Observations and Measurements

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3.1 Germination

The following discussion on germination draws from Cooper (1981).

3.1.1 Sampling—When testing seed germination, seed should be sampled from the entire seed store. Problems due to unrepresentative samples are sometimes encountered when seed is sampled only from the top of the seed bag or where a number of bags are to be used, only from the bag that is easily accessible. Sampling should be done close to sowing time.

3.1.2 Measuring—Germination (expressed as a percentage) is the number of seed germinated divided by the total number of seed in the incubated sample. Only seeds possessing an embryo should be tested for germination, however, the number of cracked or partial seed not possessing an embryo should be expressed as a percentage (by weight) of the total seed stock.

A germinated plant includes a coleoptile (with enclosed leaf) and seminal roots (see Appendix 2 for an explanation of plant parts). An abnormal plant should be noted, but not counted as a germinated seed.

Germination can be measured by various methods, including 1) petri dish, 2) rag doll, or 3) soil box.

Note for 1) and 2): Many germination tests are inadequately performed as seed is either kept too moist (and thus rots) or too dry (and thus the seed can not imbibe). As a general rule, there should be no free moisture apparent, but the medium should be moist to the touch. This can be achieved by freely wetting the paper in both petri dish and rag doll tests and then allowing to drain under gravity for 10-20 seconds.

- Petri dish. Place a known number of seeds (best to test a minimum of 100 seeds) on moist paper in a petri dish and count the number of germinated seeds after incubating for 4-5 days (room temperature is usually adequate).

- Rag doll. Place 100 seeds on a moistened piece of paper toweling, place another piece of paper on top of the seed, moisten and roll the paper up; place in a plastic bag. Count the number of germinated seed after 4-5 days (room temperature is usually adequate).

- Soil box. Count the number of seeds that germinate from seed planted in a soil box. Seed can be planted at the expected seeding depth (e.g., 3-5 cm). The soil box method has the added advantage of giving some indication of seed vigor as well as percent seed germination (room temperature is usually adequate.)

Information gained:
Germination can be used to assess the quality of the seed. Germination can be adversely affected by many factors, including storage conditions (especially heat, humidity, and exposure to chemicals; see Section 4.2), and conditions during grain-fill. If germination is low, then adjustments are required to the seeding rate to ensure a satisfactory plant stand. The adjustment is calculated by:

$$\text{Required seed rate} = \frac{\text{Desired seeding rate} * 100}{\text{Percent germination}}$$

A germination of 85% or more is usually considered adequate.

3.2 Seed viability

Seed viability refers to whether the seed is alive or not. A simple test is to take a number of seeds, soak them in water for 24 hours, cut then in half and then soak them in tetrazolium (10% solution) for another 10 minutes. Seed showing a darkening are viable. The number of viable seed should then be compared with the number of germinating seed to check for dormancy.

Information gained:
Seed may at times show low germination due to dormancy. This test is used to check seed viability when germination is low.

3.3 Depth of seeding

Seeding depth is assessed by digging up 10 plants at random and measuring the distance from the seed or seed remnant (check for the origin of the seminal roots) to the soil surface or to the point of color change (white to green) on the culm. However, if sowing is so deep or soil characteristics so poor (e.g., crusting) that percent emergence is substantially lowered, the depth of those seedlings that do not emerge may be more than that of the total population of emerged seeds. Searching for seedlings, which failed to emerge, is time consuming but may have to be carried out on occasions; such seeds are characterized by the leaves that are generally yellow and bent (due to first leaf emergence and growth under the ground).

Note: The seeding depth after planting and before rain (in a conventional-till situation) will appear to be 20-40% more than the depth after substantial rain, which causes the soil to settle.

Information gained:
The emergence of seed planted at a depth that is too shallow may be reduced by both temperature and moisture stress. In addition, the development of crown roots may be poor. When seed is planted too deeply, the emergence and general vigor of the plants are generally reduced. Seedlings may fail to emerge even though they germinate. Other causes of poor emergence of apparently high germination % seed can be poor seedling vigor, waterlogging, seedbed crusting and compaction, and fungus, insect, bird, or rodent attack.

3.4 Tiller emergence

Tillers emerge from within the leaf sheath of each leaf. The first tiller (T_1) will normally emerge from the axil of leaf 1 at the three-leaf stage. Tillers are normally counted when they are visible above the ligule of the leaf in which they are formed, and are numbered according to the numbering of these leaves (see Appendix 2).

Information gained:
Tillering pattern is especially useful as a record of the stress history of the crop (Klepper et al. 1982). If for example, T_1 is absent then it implies stress during the 1-3 leaf stage. A well managed crop may have 50% T_0 (the tiller that emerges from the seed or coleoptile tiller) and 100% T_1, whereas a crop experiencing early stress may have only 15% and 50% respectively, but a greater number of T_2 tillers (see Appendix 2 for a description of the tiller numbering system).

3.5 Lodging

3.5.1 Area—Estimate the area (**percent of area to be harvested**) lodged and the angle of the stem in relation to the vertical, or more accurately the angle made by a direct line from the spike to the base of the stem (see 3.5.2 for measurement)

Note: Many people estimate lodging on an entire plot basis rather than on a harvested area basis; this can cause problems when attempting to interpret yield data using lodging percentages as a covariate, particularly since borders, which usually lodge less, are generally not included in the harvested sample.

3.5.2 Timing and estimate—It is very important to note the growth stage or at least the date at which lodging occurs. The timing and cause of the lodging can usually be determined by reference to the occurrence of a rain storm or irrigation event, and this should be done when the lodging is first noted, not months later when memories are less reliable. Sometimes especially with early lodging, the angle of lodging may change with time; both the initial and final angle of lodging should be noted (**Figure 2**). For example, plots lodged flat just before flowering will often right themselves by bending of upper stem nodes in a few days, but the bent stems will always be evident and they remain shorter than nonlodged plots and yield processes are likely to be adversely affected.

Information gained:

The timing of lodging greatly influences the extent of yield loss (Stapper and Fischer 1990). For example, lodging at early grain-fill results in much greater loss than lodging during hard dough. Once lodged, yield losses probably arise due to shading effects and/or due to increased disease incidence on the spike and upper leaves. Additional yield losses may occur because of lodging during mechanical harvesting. In variety trials, if there is sufficient variation in lodging score between replicates (within varieties), then the lodging score may be used as a covariate to adjust yields.

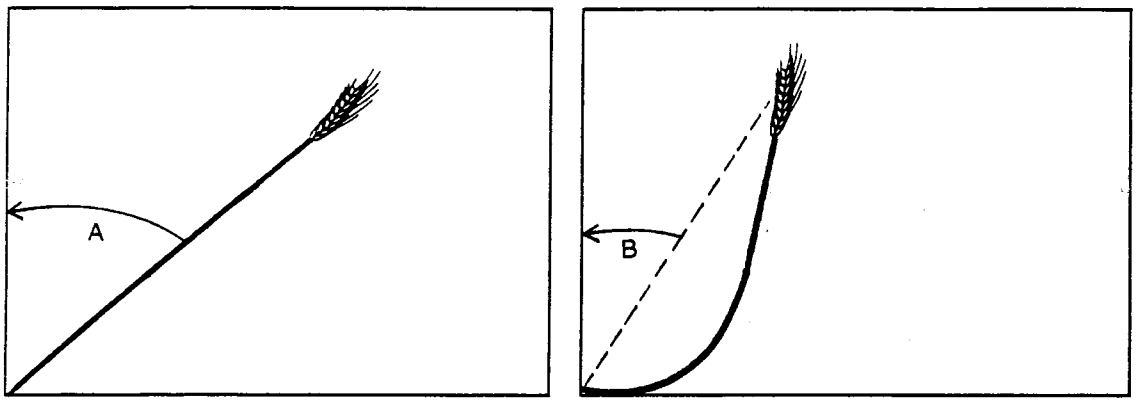

Figure 2. Angles to record when a crop lodges and algorithm to calculate the lodging score. (The angle is calculated relative to the vertical and a line from the base of the stem to the base of the spike.)

3.6 Plant height

Measure from the ground to the average top of the terminal spikelet (do not include the awns). At least three handfuls of spike-bearing culms should be measured per plot, considering all productive spikes and an average estimate. Culms must be pulled up to the vertical position if they are lodged before measurement.

Information gained:

Plant height gives an indication of dwarfing due to both genes present and environmental conditions affecting growth. Variation of height between sites for the same variety may therefore be potentially used as an indication of stress or fertility. Although often measured, however, plant height is seldom constructively interpreted. Plant height may also give an indication of competitive ability against certain types of weeds, although the rate of early ground cover seems to be a better mechanism than height for competing with weeds (K. Sayre, pers. comm.).

4 Biomass or Total Above Ground Dry Weight at Various Growth Stages

Biomass sampling can serve for many purposes (crop growth rate, dry matter distribution, leaf area, organ size, nutrient content). In most cases, such determinations are made on representative

subsamples (see section 4.1) to reduce processing time. In such studies, dry weight normally refers to the constant weight reached after drying at 70°C for 24 hours in a well ventilated oven.

Processing samples fresh and no later than physiological maturity is recommended to avoid loss of plant parts (leaf laminas, grains) due to brittleness and shattering, and is essential for leaf area determination.

4.1 Subsampling

If the samples are large and/or bulky, subsamples may be taken for drying and the fresh weight of the sample and subsample used to relate the latter to the former (see example). It is always easier and more accurate to subsample, and usually to process samples, before the crop has fully dried. Biomass samples usually comprise collections of culms of varying size; a representative subsample can be taken by selecting groups of culms from the base of the cut material and thus avoid bias caused by selecting only taller plants or those with larger spikes.

Example

The subsample should be related to the total by weight ratios (fresh or better dry) rather than number ratio, as in the following:

Total sample fresh weight = A
Subsample fresh weight = B
Subsample dry weight = C

Therefore: Total sample dry weight = (A * (C/B).

If 50 culms were subsampled, then the total number of culms in the total sample = Ax50/B.

The subsample should consist of a minimum of 50 culms and care is needed to include all leaves, etc. associated with the culm.

Fresh weights of both sample and subsample should be determined in the field as it is easier to draw a representative subsample at the time of sampling (note: wind protection for balances may be needed). Alternately, the subsample can be placed in a plastic bag to prevent water loss and weighed more accurately in the laboratory. If the whole sample is stored and later transported to the laboratory, moisture gradients can develop in the sample making it more difficult to accurately subsample on the basis of fresh weight; dry weights of the subsample and remainder become more appropriate in this case.

Note: Care is required when sampling for comparison of treatments that all samples are collected at the same time. If not, significant errors may be introduced due to plant growth. For example, a crop growing vigorously could be producing in the order of 200 kg of dry matter/ha/day. Consequently, treatments within a replication should be sampled preferably on the same day. Delays in sampling treatments within a replication could lead to errors. Sometimes, it may be more appropriate to sample at a given stage of development.

4.2 Sample transport and storage

The situation sometimes arises where storage of samples prior to processing or oven drying is required. Such samples should be kept as cool as possible (not frozen) to reduce respiratory losses (which could be 2-3%/day at room temperature). If samples require processing, then plastic bags should be used for storage to avoid drying out. If only drying is planned, then in some climates, sun drying is a quick way to initiate the drying process. At maturity under dry field conditions, all samples come down to the same low moisture content within a few days of cutting and lying in the field (e.g., at a RH of 60%, grain moisture content is around 13.5%; **Figure 3**. Figure 3 also shows how grain moisture content affects the storage life of the grain).

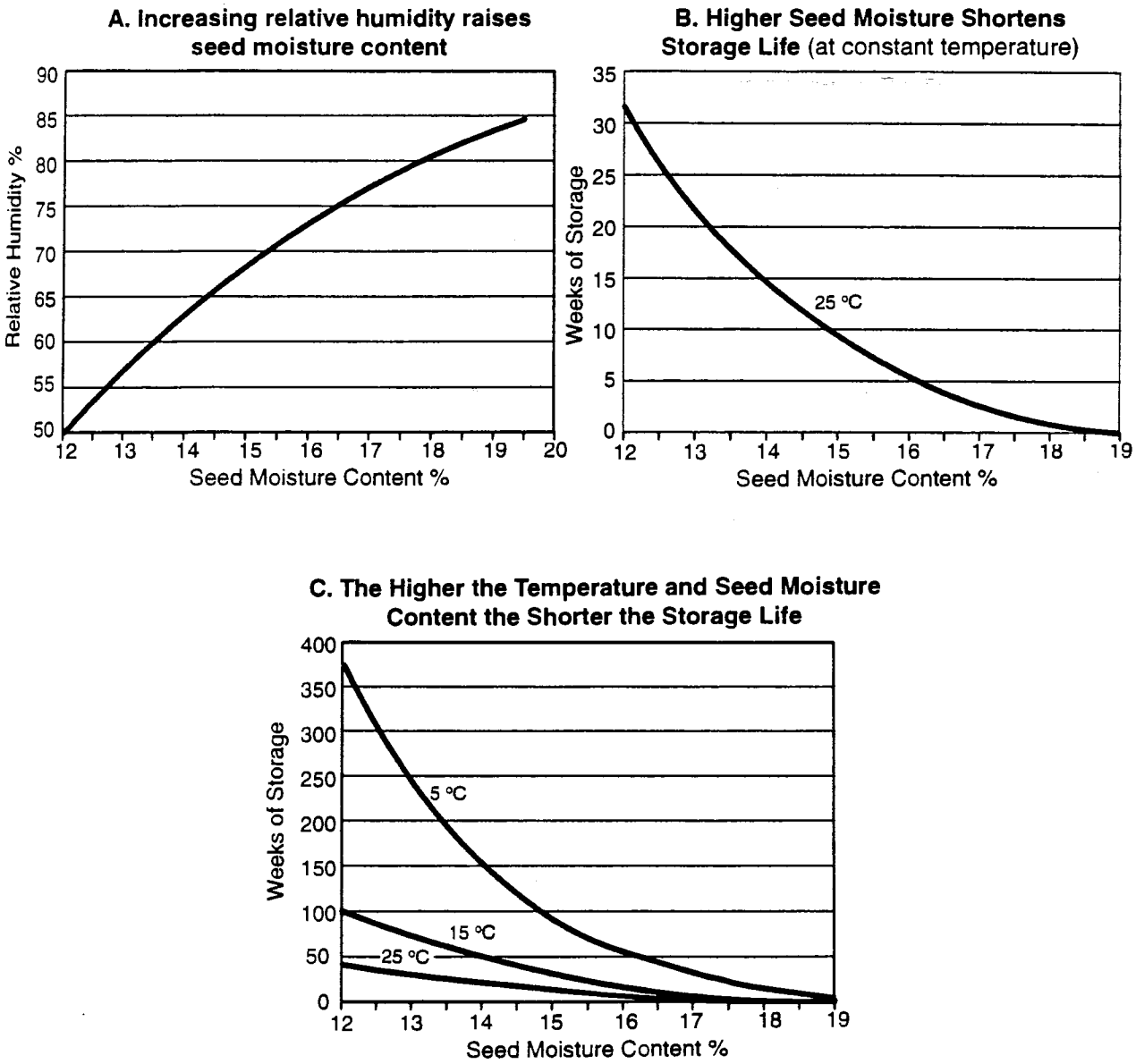

Figure 3. Relationships between a) relative humidity and seed moisture content, b) seed moisture and storage life, and c) seed moisture, temperature and storage life.
Source: Douglas (1980).

4.3 Crop growth rate and partitioning studies

Crop growth rate (CGR) is defined as the change in dry weight per unit land area per unit time. Determination of CGR requires biomass measurement at various times during the growth cycle (as outlined in sections 4.4-4.7). As an absolute minimum, it is recommended that biomass be sampled at the early seedling stage (say 4-5 leaf), at anthesis and at maturity.

Partitioning studies analyze the distribution of dry matter between the different plant parts. This requires biomass sampling throughout the life cycle of the crop and separation of the samples into the different plant parts. The different fractions are dried at 70°C before being weighed.

Information gained:

Crop growth rate varies primarily with captured radiation, with minor effects of some other factors (e.g., nitrogen, temperature). **Figure** 4 shows the general relationship between CGR and radiation. Crop growth rate changes throughout the cycle, starting slowly, reaching a maximum with maximum light interception and then decreasing as leaf area declines (**Figure 5**). **Figure 6** shows typical results of a partitioning study.

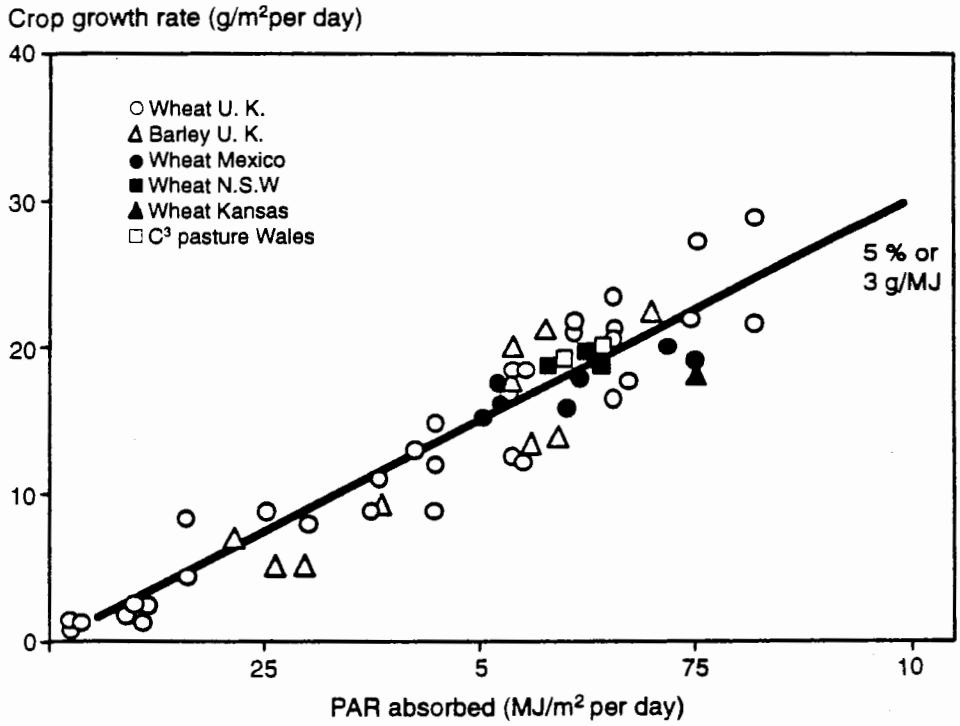

Figure 4. The relationship of crop growth rate to absorbed photosynthetically active radiation (PAR) for wheat and related species. Data from Gallagher and Biscoe (1978, UK); R.A. Fischer, unpublished (Mexico), Doyle and Fischer (1979, NSW); and Hodges and Kanesamu (1977, Kansas). The two points from Sheehy and Cooper (1973, Wales) refer to erect (uppermost point) and nonerect (lowermost point) genotypes (from Fischer 1983).

4.4 Biomass at early seedling stages (up to about five leaves)

If plant density (section 6.1) is known, biomass per plot can be calculated by sampling 10-20 plants at random. The plant material is dried at 70°C and then weighed. Calculate as:

$$g/m^2 = \frac{(\text{weight of X plants in grams}) \times \text{plants}/m^2}{X}$$

The sampling is accurate only if:

- The plants can be separated,
- A true estimate of plant population is made (see section 6.1)
- The plants are sampled strictly at random.

Plants sampled at this stage are usually most easily collected by uprooting (sometimes this is facilitated by loosening of the soil below the plant with a knife or spike). Plants must be rinsed to remove soil particles. The dry weight of seedlings can be substantially distorted by soil even when the plants are cut off at ground level because soil particles can adhere to stems and lower leaves; therefore, careful washing is essential. The roots need to be trimmed off in order to get a true weight (this may or may not include removal of the crown or stem bases); later samplings usually involve cutting at ground level, which leaves the stem base behind). A decision must be made prior to

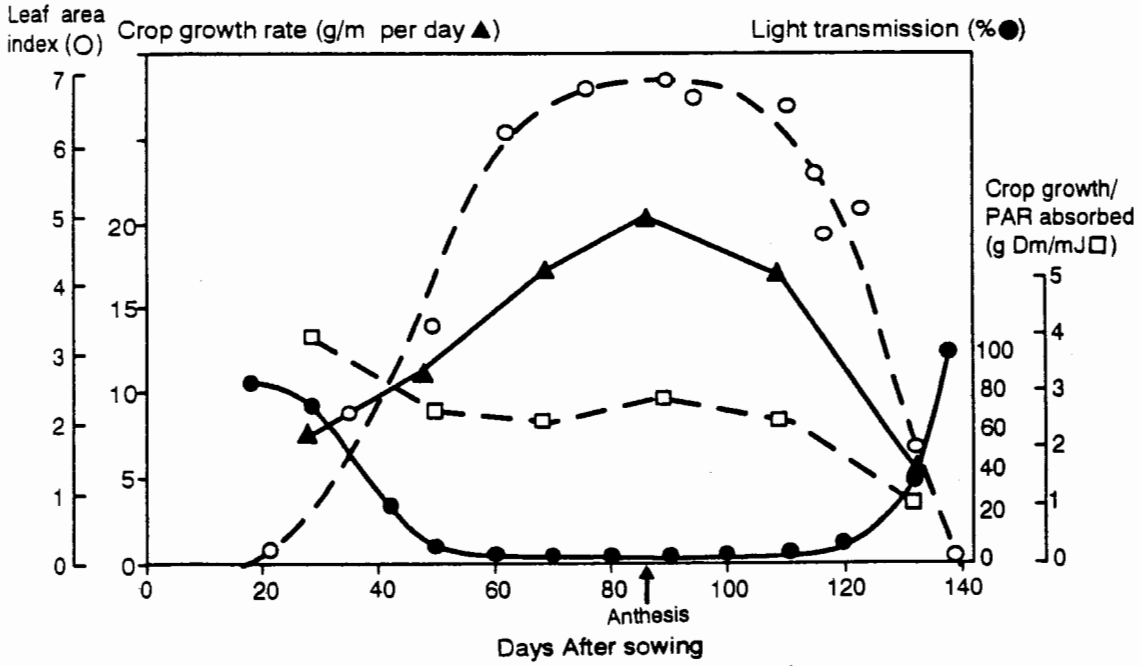

Figure 5. Variation in leaf area index, % incident photosynthetically active radiation (PAR) transmitted by the canopy, crop growth rate (CGR), and CGR per unit of absorbed radiation (from Fischer 1983).

sampling, whether to include that stem material that occurs above the crown, but below the ground surface, as this material can influence results by as much as 15% at this early stage (E. Acevedo, pers. comm.).

Information gained:
Early growth or biomass sampling (e.g., 4-5 leaf stage) is very useful as an indicator of early vigor, something influenced by genotype, and/or soil physical conditions such as mechanical impedance.

4.5 Biomass after 5-leaf stage (and before anthesis)

Biomass samples should be taken from a specified area in the plot (See section 1 for more details on sampling procedures). Generally, a single sampling area/plot in replicated trials suffices. The sample should be the width of the plot (after borders are discarded) and usually of at least 0.5 m in length. All above-ground plant matter should be cut and the sample dried at 70°C. Biomass is calculated as dry weight (0% moisture) per area harvested. Care must be taken to avoid samples that are not representative, particularly if too close to a border.

Sampling a farmer's field usually requires a larger number of samples than required for plot sampling (see section 1.4).

Three methods are possible:

- Biomass samples after the 5-leaf stage are usually much more accurate if taken by quadrat (i.e., cutting or pulling up plants over a given area; the area chosen as described earlier in section 1). Cutting can best be done with sharp shears (as used for wool shearing of sheep), operated by one hand, or a sharp sickle or electrically powered clippers. For consistency, cutting should be done as close to the soil surface as possible, leaving a stubble of no more

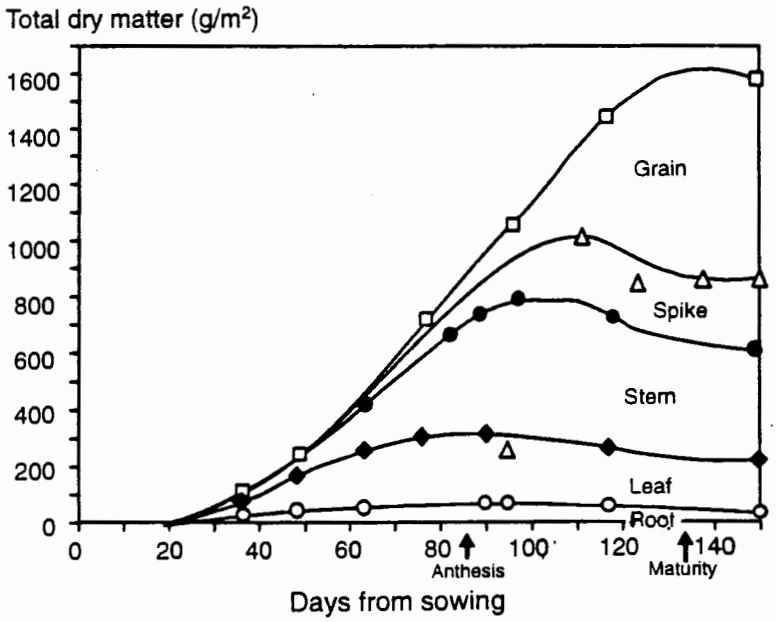

Figure 6. Dry matter accumulation in wheat crop parts (from Fischer 1983).

than 2 cm. If the total sample is to be used then it must be dried immediately to give total dry weight alone. In such cases, the sample should be collected in paper, cloth, or plastic mesh bags (bags preferably of constant weight to facilitate weighing without sample removal from the bag). Immediate drying is necessary to avoid maintenance respiration losses that could amount to 2-3% per day.

- Entire samples can be loaded from the bags into typical dehydrator trays for drying of the total sample. Complete drying has occurred when the innermost part of the sample is crisp as best indicated by brittleness of stem internodes: this may take several days.

- Biomass can be estimated from subsamples as outlined under section 5.

4.6 Biomass at anthesis and anthesis plus 7 days

Measurements at or near anthesis can reveal many important aspects of the crop. One of the most useful samplings is that made not exactly at 50% anthesis, but rather about 5-7 days later (A+7) when all the spikes have emerged (and can be counted), anthesed, the inflorescence structure has ceased growing (the number of competent florets has been established), and stem elongation is complete. Another advantage of sampling at A+7 is that scheduling of field activities is facilitated. Take samples as specified above (section 4.5). A subsample of spikes (10-15) can be retained after drying for later determination of true inflorescence weight (the most closely related factor to competent floret number) by separating the small grains inevitably present in some spikes in such a sample and subtracting their weight: gentle threshing and winnowing of the sample is required.

Information gained:
Biomass samples at A+7 can be used to give not only total dry weight [and by back-interpolation total dry weight at 50% anthesis (i.e., approximately total dry weight - weight of small grains), but also flag or total leaf area index and dry weight distribution (especially the weight of inflorescence)] at the beginning of grain-filling. The size and form of the crop at this stage represents the sum of all pre-anthesis factors affecting growth and yield.

Biomass at anthesis has been shown to be related to final yield, especially under irrigated conditions. Inflorescence dry weight may be an even better predictor of kernel number and yield than biomass (Fischer 1985a,1993).

4.7 Biomass at maturity

At least three sampling procedures can be adopted in order to determine biomass at maturity (along with harvest index and some numerical yield components). For the three approaches, it is recommend that sampling be done as soon after 90-95% physiological maturity (DC86) as possible in order to avoid processing losses.

The first two approaches (section 5.1) measure both biomass and yield on a sample cut from a given area and may or may not be followed at harvest ripeness by separate yield determination using a combine harvester.

The third approach (section 5.2) determines harvest index at physiological maturity from a grab sample and relies on an accurate grain yield determination by later combine harvesting to calculate biomass and spike number.

5 Yield Components and Harvest Index

Yield components can be measured individually (section 6) or can be calculated (sections 5.1 and 5.2).

5.1 Yield components from a given harvest area

Two related methods are outlined below. A third method for estimating yield components from a random grab sample is presented in section 5.2.

Method 1—As described previously in sections 1 and 4.5, above ground material is cut and collected from a given area (A). Total dry weight of the sample (DWS) can be determined directly, but more commonly a subsample (DWSS) of say approximately 50-100 random spike bearing culms is taken for oven-drying and DWS is calculated from the total sample fresh weight (FWS), or remainder fresh weight (FWR), and the subsample fresh weight (FWSS).

Note on unit and unit conversions: Depending on sample size, measurements will be in either g or kg and calculated on a per m^2 basis. To convert g/m^2 to kg/ha, multiply by 10; g/m^2 to t/ha divide by 100; kg/m^2 to t/ha multiply by 10.

Calculations

- Fresh and dry weight of samples:
  FWS = FWR + FWSS (g)
  DWS = DWSS x (FWS/FWSS) (g)

- Biomass on an area basis (TDW):
  TDW = DWS/A (g/m^2)

- Total spike number in the sample (SNS) can be calculated from a count of the exact number of spikes in the subsample (SNSS):
  SNS = SNSS * (FWS/FWSS)

- Spike number on an area basis (SNO):
  SNO = SNS/A ($spikes/m^2$)

- Often, the remainder of the sample is left to air-dry in the field and is then threshed to give grain in the remainder (GDWR). Grain dry weight of the total sample (GDWS) is as follows:
  GDWS = GDWR * (FWS/FWR) (g), or
  GDWS = GDWR * [FWS/(FWS-FWSS)] (g)

- Yield on an area basis (GY):
  Yield = GY = GDWS/A (g/m^2)

Notes: 1) This may refer to field-dry weight or oven-dry weight. It can be converted to a standard moisture content based on the moisture content of a subsample of the threshed grain: See section 8.4 or method 2 below. 2) If the above procedure is used, work load can be reduced as it is not really necessary to thresh the subsample).

- Harvest index (HI %) is given by:
  HI = 100 * (GDWS/DWS)

For greatest accuracy in calculating HI, GDWS, and DWS should refer to oven-dry weight (i.e., dried at 70°C). Moisture meters, designed for the purpose, can be used to determine grain moisture content. The HI could be calculated using grain and biomass weights at field-dry moisture; this saves much time but, there will be uncertainties relating to moisture contents, which may need to be checked out.

Method 2—To calculate yield components, the following procedure is recommended:

1) Cut all above-ground biomass in a pre-determined area (A) (e.g., 1-2 m^2). Avoid border effects by sampling away from edges of the subsample.

2) DWSS: Sample between 50-100 spikes (leaves and stem) randomly from the large sample and measure the fresh weight of the sample.

3) FWR: Measure the fresh weight of the remaining bulk sample.

4) DWSS: Dry the subsample of spikes (usually 70°C) and then weigh.

5) TG: Weigh the fresh weight of grain threshed from the large sample.

6) WG: Take a subsample of grain from the large sample and weigh before drying.

7) DG: Oven dry the subsample of grain and weigh.

8) W1, W2: Measure the weight of two subsamples of 100 entire oven-dried grains selected at random.

Calculation of yield components

- Biomass:
  1) Dry biomass = TDW = (DWSS/FWSS) * [(FWR + FWSS)/A] (g/m^2)
  2) Biomass at x% moisture = TDW * [100/(100 - x)] (g/m^2)

- Grain yield (GY):
  1) GY at 0% moisture = [(DG/WG) * TG] * [(FWR + FWSS)/FWR]/A (g/m^2)
  Note: This assumes that grain dried at 70°C is at 0% moisture.
  2) GY at x% moisture = GY * [100/(100 - x)] (g/m^2)

Note: Moisture content is calculated on a fresh weight basis, i.e., water content divided by fresh weight and then multiplied by 100. =[((WG - DG)/WG) x 100]

- Straw weight (SW):
  1) SW at 0% moisture = TDW - GY (g/m^2)
  2) SW at x% moisture = SW * [100/(100 - x)] (g/m^2)

- Harvest Index (HI):
  HI = GY (at 0% moisture)/TDW

- Thousand grain weight (TGW):
  TGW = (W1 + W2) * 5 (g)

- Spikes per m^2:
  = [(FWR + FWSS)/FWSS] * X/A (where X = number of spikes in subsample)

- Grains per m^2:
  = (GY * 1000)/TGW

- Grains per spike:
  = (grains/m^2)/(spikes/m^2)

Note: Various combinations of methods 1 and 2 can be used to suit time available, facilities, and purposes.

5.2 Yield components from a random grab sample

Yield components and harvest index can be determined directly by taking random culms from the crop at physiological maturity. Twenty to 40 culms can be taken from the rows (or area) to be harvested by reaching six to eight times into the crop close to ground level and then grabbing four to eight culms and cutting them off at ground level; take care to collect attached leaves. All harvest rows should be represented in the sample. Culms from a plot are bulked, put in a bag and dried at 70°C (take care not to break up spikes if yield components of the spike are going to be determined later).

Before threshing this sample to determine dry grain (GDWS), one can determine total dry weight (DWS), spike number (SNS, normally easiest to count when culms first collected), spikelet and floret number, and dry matter distribution.

Harvest Index (HI %) is determined as before, i.e., 100 * (GDWS/DWS).

The method requires an accurate determination of grain yield per unit area (GY) (often by mechanical harvest of the plot) for calculation of biomass (TDW) and spike number (SNO) calculation:

TDW = GY * (100/HI).
SNO = GY * (SNS/GDWS).

Note: The weight of 200 grains can be determined on grain either from the HI sample or from the larger plot harvest, and all other yield components can then be calculated as described under section 5.1.

This method has the advantage that the hand-sampling can be quite rapid (<5 minutes/plot) after which the samples can be readily stored for processing when time is available. It is suited for remote sites and when plots differ considerably in maturity. However, it does rely on accurate yield determination usually by a plot harvester. If this can be achieved, the method is both more accurate and generally less demanding of labor than methods outlined in section 5.1. Note that with this method, the measurement of HI is statistically independent of the measurement of grain yield, whereas that of TDW is not independent of GY.

5.3 Sampling biomass in lodged crops

It is difficult to cut a given area in a lodged crop especially if it has been sown by broadcasting. The process is facilitated by folding back spikes and stems to establish a starting reference line before inserting a quadrat (see section 1.4.2). Great care must then be taken to collect only those plants whose crowns fall within the randomly located quadrat.

6 Individual Yield Components by Field Measurement or Calculation

6.1 Plant population

A count of plant population should be made after the maximum number of plants is expected to have emerged and before tillering occurs (usually 10-14 days after the advent of suitable moisture for germination).

If plants are sown in rows, then 0.5 m length from each sampling row or from at least six such rows should be counted per plot.

If broadcast, then samples of at least 0.5-1.0 m^2 should be taken from each plot. The number of such samples required will vary with the degree of variation within the plots, but generally at least two per plot should be a recorded.

The mean plant density may disguise important variability in plant distribution (i.e., presence of gaps that will cause yield reduction). This should be noted and measured by estimating the percent of the plot that has missing plants.

Information gained:
Plant population can be used to assess the germination, vigor, and emergence of seed sown, and/or the extent of compensation under conditions advantageous to tillering. Also needed if early growth per unit area is going to be monitored by successive measurements of growth per plant. Plant population typically varies between 50-300 plants/m^2. The number of plants/m^2 has a broad optimum and will vary with variety, climate, and management; however, under good rainfed conditions, 100 plants/m^2 could be considered a minimum for maximum yield, unless the crop is growing on residual moisture when the optimum density may be less.

6.2 Spikes/m^2

The number of productive spikes can be measured nondestructively by counting in a given area or length of row, or calculated from sampling as demonstrated above under section 5. *Spikes per m^2 can be measured most easily before physiological maturity. Measuring just prior to physiological maturity can be advantageous because yield loss due to shattering caused by movement in the plots is reduced. In broadcast planting, direct measurement can be difficult especially if crops lodge (see sections 1.4.2 and 5.3).

*If measured directly, the procedure and number of subsamples are as for plant population (section 6.1).

Information gained:
Spikes/m^2 can be used to assess the final number of productive spikes/m^2 and can be combined with the plant population count to assess the extent of tillering. Tillering typically ranges from 1-10/plant. Spikes/m^2 is determined by events over the whole period from sowing to flowering and is variety-, management-, and environment-dependent.

6.3 Spikelets/spike

Sample a minimum of 10 spikes/plot at random (aim for a total of 30-40 spikes/treatment); select the culms from the base and count the number of spikelets. Take the average based on sample size. Most commonly, count the fully developed or grain bearing spikelets (or at least those large enough to be expected to have at least one grain). Potential spikelet number is obtained by counting all the nodes on the rachis; it can exceed the developed spikelet number because of abortion of spikelets at the base or tip of the spikes. Alternatively, under excellent environmental conditions, all potential spikelets can develop into grain-bearing spikelets.

Information gained:
The potential number of spikelets/spike is determined by the time of terminal spikelet formation (in wheat and triticale; barley does not form a terminal spikelet) around first node appearance. Subsequently, primordial spikelets at the base—and later at the tip of the spike—may abort because of stresses. Normally, 10-25 spikelets may form on each spike.

6.4 Grains/spikelet

Sample as for spikelets per spike, count the spikelets, thresh, count grains, and calculate; or less accurately simply calculate from calculated grains per spike and measured spikelet number. When large numbers of samples or plots need to be sampled, some time may be saved by randomly counting only one side of the spike and multiplying by two.

Information gained:

Grains/spikelet is the result of both the number of competent florets/spikelet and kernels/competent floret (or grain set; see section 6.5). Values for competent florets per spikelet typically vary from 1.5-5.0 and for kernels per competent floret from 0.6 to 0.99 (see discussion below in section 6.7).

6.5 Grain set

Grain set refers to the percentage of competent or entire florets (florets with fully formed plump green/yellow anthers at flowering), which actually produce grain (the opposite of the percent sterility) and should reflect conditions around anthesis (e.g., pollen fertility, early grain survival), in contrast to grains per spikelet, which can be influenced by earlier conditions as well. However, at maturity, it is difficult to know which florets were competent. It is suggested that the basal two florets of the 6-10 central spikelets are always competent, and therefore an index of grain set can be obtained by observing the percentage of such florets with grains. Sample as specified for section 6.3 or count 10 such spikelets in five random spikes per plot; the total of missed florets is the % sterility (= 100 - % grain set).

Alternatively, one can use matched spikes (i.e., spikes showing equal size and development). One spike is sampled at anthesis and the other at maturity, counting (destructively) competent florets at anthesis in the one spike (i.e., those florets showing normal development of anthers; noncompetent florets will show whitened, flattened anthers that have no fertile pollen and whose stamens never elongate) and grains in the other spike at maturity. At least 20 matched spikes per treatment are needed for reasonable accuracy; selection of matched spikes and counting at anthesis are time consuming.

Information gained:

Grain set is an indicator of the occurrence of stress events around anthesis (e.g., drought, temperature extremes, B deficiency, or genetic sterility, which can interact with the environment). It is a more precise and hence useful measure than grains per spikelet or grains per spike. See notes in section 2.2.6 for further discussion on grain development.

6.6 Thousand grain weight at maturity and during grain-filling

To measure thousand grain weight (TGW), count out two random samples of 100 entire grains (i.e., those possessing an embryo). Dry the grains at 70°C (48 hours should be sufficient) and weigh. This will usually give sufficient accuracy. If weights differ by more than 10%, a third sample of 100 should be taken or recheck the counts.

In order to study grain growth during grain-filling, greatest accuracy is achieved by selecting groups of sufficient number of spikes, matched for anthesis date and size, so that one can be sampled at random from each group on each sampling date. Four to eight such groups (four to eight spikes each date) per treatment should be sufficient to permit accurate calculation of grain growth rate by linear or curvilinear regression (Loss et al. 1989). The study can be based on all grains in the spike or on a given position in the spike (e.g., basal florets of central spikelets).

Information gained:
A reduction in TGW may be due to climatic or biological (e.g., pathogen) stress during grain-fill. Kernel weight (calculated as TGW/1000) usually varies between 20 and 50 mg. A decreased grain weight may not be an indication of stress during grain-filling, however, due to the plasticity of the yield components. For example, if the plant population is high leading to a high number of kernels/m^2 then TGW may be decreased without yield being seriously affected. The TGW tends to be characteristic of a variety and large differences exist between varieties even under good conditions. Within a variety, kernel weight usually shows a negative linear relationship to mean grain-filling temperature.

6.7 Grain or kernel number (per m^2)

Kernel number per m^2 (KNO) is usually calculated, dividing grain yield (GY, g/m^2) by kernel weight (KW, mg):

$$KNO = GY * (1000/KW).$$

Note: Using this calculation, KNO is statistically linked to GY and may give rise to spurious correlations between GY and GNO if GY is not determined accurately.

Kernel number can also be independently measured by determining directly spike number (SNO/m^2), and kernels per spike (KPS) from at least 20 spikes/plot sampled at random (aim to sample 60-100 spikes):

$$KNO = SNO * KPS$$

Information gained:
KNO acts as a summary of all events up to and a little beyond anthesis. For example, the combined effects of management and climate on plants/m^2, spikes/plant, spikelets/spike, and grains per spikelet are all combined in this single term. Competent floret number (the precursor of kernel number) is also well correlated with spike (inflorescence only) dry weight at anthesis; the relationship being of the order of 100 florets/1.0 g spike (10 mg/floret), although the range across varieties for grain number is from 70-140 kernels/g spike dry weight at anthesis.

Under many conditions, yield is a function of KNO, which is particularly dependent on crop growth rate during the period of rapid spike growth (emergence of the second last leaf—or about 1 month before anthesis for spring wheats—until just after anthesis).

7 Grain Quality

Reference

Pomeranz, Y., ed. 1988. Wheat Chemistry and Technology. Vol. 1. and 2 Pub. Amer. Assoc. Cereal Chemists Inc. 562 pp.

One important aspect of grain quality that can be easily assessed is the weight per unit volume or the hectoliter (hl) weight. Hectoliter weight is measured in the laboratory (Halverson and Zeleny 1988). For small quantities of grain (e.g., as little as 100 g can be used), simple micromethods can be devised and calibrated against standard ones. The standard test involves using about 1 kg of grain, and the weight in a given volume (1 L; specifications: 11.577 cm diameter, 9.5 cm deep) is measured. Obviously, as the sample size becomes smaller, the accuracy decreases. Another, although less precise, evaluation of quality can be made from TGW (see section 6.6).

Figure 3 (page 17) shows how grain moisture content rises with relative humidity and how storage life is reduced as grain moisture increases.

Information gained:
Hectoliter weight, to be considered acceptable, should be greater than 78 kg/hl for hard spring wheats and greater than 76 kg/hl for soft spring wheats (A. Amaya, pers. comm.). The hl weight bears a close relationship to the flour yield (i.e., percent of flour produced with milling a given weight of grain).

See also Ghaderi and Everson (1971) and Ghaderi et al. (1971) for work relating hl weight to grain characteristics such as TGW.

8 Yield Estimation and Measurement

8.1 Visual estimates

Yield can be estimated by visual assessment (this generally requires experience and a knowledge of the variety and area) or by use of yield components from the stage of mid-grain-fill onward.

8.2 Yield estimates from yield components

To calculate yield from yield components, first estimate the number of spikes/m^2 from *in situ* counts as outlined in section 6.2. Next, randomly sample and then count the number of grains/spike as described in sections 6.3-6.4. Then, assume a TGW based on the variety and conditions expected during the rest of the grain-filling period (Typical TGWs range from 30 to 40 under reasonable grain-filling conditions and temperatures). Calculate yield with the following equations:

- Yield (g/m^2) = spikes/m^2 * grains/spike * (TGW/1000)

- Yield (kg/ha) = yield (g/m^2) * 10

Variability of the field or treatment, which is being studied, and the desired accuracy will determine the number of spike counts made. Inevitably, for greatest accuracy many small samplings are best and are feasible when dealing with nondestructive sampling. For example, for a 1-ha drill-planted field, take spike number counts in 20 random but well dispersed 2-row x 50-cm quadrats and combine these counts with kernel number counts in 50 random spikes; a reasonable estimate of kernels/m^2 should result (but be careful to select sampling sites and spikes at random). Counting kernels per spike (one side x 2) while walking between quadrat sites saves time and the whole job should take less than 30 minutes. Be sure row spacing is known accurately and/or measure the spacing to confirm; replicate the measurements for accurate assessment.

Example

Average row spacing: 15 cm
Average spike count (2 row by 50 cm): 40 spikes
Spike sample area: 2 rows * 15-cm row spacing * 50 cm = 0.15 m^2
=> 40 spikes/0.15 m^2 = 266.7 spikes/m^2
Average kernel count/spike = 21.5
=> Kernels/m^2 = 266.7 * 21.5 = 5734
Assume TGW = 40 (based on experience)
=> Yield = 5734 * 40/1000 = 229.4 g/m^2 = 2294 kg/ha.

8.3 Yield estimates from samples

Yield can be estimated as outlined for yield components (section 5). Alternatively, borders can be discarded and yield estimated from the remaining plot that is harvested. In some instances, it is not possible (especially in on-farm trials) to harvest the entire plot or to thresh the entire plot sample. In these cases, follow the options outlined in section 5, or sample 5x 1m^2/field or 2 samples of 1m^2/plot if there are 3 replicates.

8.4 Yield moisture contents

The grain trade and farmers usually express yield at a given moisture content (e.g., 10% in Australia, 12 or 14% in Europe on a fresh weight basis). Therefore, conversion factors are required to adjust moisture. Moisture content is calculated as the weight of moisture relative to the fresh weight: i.e.,

[moisture/(moisture + dry weight)]

The following formulas outline the various moisture calculations:

- Yield and grain moisture calculations:
  Field weight of harvested grain = FW (kg)
  Harvested area = A (m^2)
  Fresh weight of subsample = WS
  Oven-dry weight of Subsample = DS

- Grain moisture conversions:

  Grain moisture content (M%)

  $M\% = [(WS - DS) * 100]/WS$

  Yield (unknown moisture, GYm)

  $GYm\ (t/ha) = (FW * 10)/A$

  Yield (0% moisture, GY(0%))

  $GY(0\%) = [GYm * (100 - M)]/100$

  Yield (X% moisture, GY(X%))

  $GY(X\%) = [Y(0\%) * 100]/(100 - X)$

Throughout the previous discussion, all weights of plant parts including grain refer to constant weight after drying at 70°C. However, the American Association of Cereal Chemistry (AACC) defines 0% moisture as that achieved by drying ground grain at 130°C. Therefore, further conversion factors are actually required to the above to have a true 0% moisture reading. J. Peña (unpublished data) determined that, in order to convert the weight of grain dried for 20 hours at 70°C to a moisture content as defined by AACC, the weight needs to be divided by 1.025 (because grain dried at 70°C has approximately 2.5% moisture). The factor dropped to 1.012 as drying time at 70°C increased to 48 hours. This means that, to express grain at 10% moisture, the oven-dry weight (70°C for 24 hours) needs to be multiplied by 1.084 (i.e., 1.00/1.025); R.A. Fischer (pers. comm.) had previously advocated a multiplication factor of 1.068.

9 Crop Residue Amount

Collect and bulk the straw found within five random samples (or two per replicate) of at least 1.0 m^2. Dry the straw in an oven (70°C) and weigh. Convert g/m^2 to t/ha by dividing by 100.

Due to the generally large spatial variation in ground cover, a visual estimate (with experience) is often sufficiently accurate (see Section 4.13 of Bell and Fischer 1993).

Information gained:
Residue amount can be used for an assessment of soil cover and thus potential evaporation reduction but, more importantly, erosion control. Assuming uniform distribution, approximately 4 t of wheat straw lying horizontally are required to give 100% ground cover (Roth et al. 1988). Straw will also be a potential source of disease for infection of subsequent crops and may immobilize N during the decomposition process.

10 Root Weight

Root weight can be approximated as about 10% of total above ground biomass at physiological maturity (or as approximately 25% of grain yield). Note that these two approximations are often not equal.

When root length density (cm/cm^3) and its distribution with depth is to be determined (e.g., in studies of nutrient or water uptake from different layers of the profile), instruments and techniques exist for determining the length of washed roots before they are dried. The common technique is the line intercept method (Tennant 1976). Where it is only desired to determine root distribution approximately (but nonetheless directly), the core break method (Ellis and Barnes 1980; section 10.1) is reasonably rapid.

Note: Roots decay throughout the cycle and are at a maximum around anthesis. Dead roots from previous crops need to be excluded before roots are measured for length or dried. Living roots can be distinguished by their turgidity or by staining (McGowan et al. 1983).

Information gained:
From root and shoot weights, the root/shoot ratio can be used as an indication of nutrient imbalances. Rooting depth indicates the maximum depth of stored water available to the crop (excluding that from capillary rise) and is needed for estimating water balances and irrigation scheduling. Rooting pattern may be a guide as to the presence of hardpans, chemical barriers (e.g., acid subsoil), earthworm holes, moisture through the profile, etc.

10.1 Core break method
Undisturbed cores (at least two per plot) of around 50-70 mm diameter, taken vertically from the profile, are broken apart at positions corresponding to the depths of interest. The roots visible on each face of the broken core revealed by the break are counted to give an estimation of the number of vertical axes/cm^2.

Root biomass can also be measured using the samples collected as above. Roots must be washed gently and dried, shortly after sampling (e.g., 1/4 hour after sampling), as weight loss may begin within 3-4 hours after sampling (because much of the root weight is solute).

The core break method is extremely useful as a rapid means of detecting root barriers, simply by determining the presence or absence of roots within parts of the soil profile.

10.2 Profile method
Another method, where the precise distribution of roots needs to be known more accurately (e.g., in the case of studies of the effect of hardpans and their disruption), is to dig a vertical trench in the crop (perpendicular to the rows or direction of subsoiling) and map the root distribution revealed by carefully washing the soil from the trench wall (Bohm et al. 1977, Bohm 1979). This method is extremely time consuming.

11 Crop Canopy Measurements

11.1 Leaf area index
Leaf area index (LAI), or green leaf area index, is the dimensionless ratio of the leaf area over the area of land sampled. LAI can be measured by cutting plants from a given area or by measuring *in situ*.

Information gained:
LAI and leaf angle interact to determine percent ground cover and the percentage of light interception. Before ear emergence in the case of wheat, a LAI of around 6 will generally intercept 95% of incident radiation. LAI is generally at a maximum at or just before anthesis. Figure 5 (p. 19) shows one example of the change in LAI over time for an irrigated wheat crop. Generally, 95% interception should occur during elongation with an LAI of 5-6 for a well grown crop under good conditions. LAI is very sensitive to N nutrition and water stress. Also, it will begin to decline even in good conditions around 30 days after anthesis.

11.1.1 Sampling plants—Plant sampling involves removing or cutting plants from a known area (see section 4 for biomass sampling procedure). All green leaf lamina material (not leaf sheaths or senescent leaf material) intercepting radiation is then removed from the stem.

11.1.2 Measurement—The easiest method for measuring area of leaf lamina for a crop like wheat is the use of automated instruments designed for that purpose. Other methods, such as estimation through length and breadth measurement (see section 11.1.3) are quite tedious. Area of green sheaths and stems can also be determined, the leaf area equivalent usually being assumed to be half the surface area (i.e., length x diameter x Pi/2) or more simply the area projected on a vertical surface (length x diameter). Spike green area may also be considered, again by taking the area projected on a vertical surface.

Alternatively, the leaf area can be estimated by photocopying the leaves, cutting out the leaf photocopies, and weighing the leaf copies and the remaining paper. The ratio of leaf copy to total weight of the page is multiplied by the total area of the page to give the area of leaves.

11.1.3 *In-situ* plants—*In situ* sampling is less precise and rather tedious for a crop like wheat. The length (l) and maximum breadth (b) of each leaf must be measured. Area is then calculated as:

$$l * b * k$$

where: k is a constant that depends upon leaf shape (Montgomery 1911). In general, k = 0.75 can be used as a reasonable estimate for wheat.

11.2 Leaf area duration

Leaf area duration relates to the maintenance of leaf area through the whole growing cycle or part thereof. It is measured as the integral in days of green leaf area index with respect to time. Often, leaf area duration refers to duration during grain-filling. This can be reasonably determined by an accurate crop sampling at or just after anthesis when the flag leaf area is separately determined (see section 4 for biomass sampling procedure). From this point on, weekly visual scoring of the area of other leaves relative to the flag and the percent of the flag leaf area that is green gives sufficient information for calculating leaf area duration. A healthy crop with good nitrogen nutrition should have three to four fully green leaf lamina on each spike-bearing culm at anthesis and a green flag leaf at physiological maturity if there is no water stress.

Information gained:
Reduction in leaf area index due to factors such as nutrition, water stress, disease, or insect attack reduce radiation interception and thus will adversely affect the supply of carbohydrates to the grain. **Figure 7** shows a comparison of green area index with time for two crops in different seasons.

11.3 Ground cover

Ground cover refers to the percent of the ground covered by green leaves or green tissue in general (or after harvest by dead crop residue). It can be estimated by laborious point quadrat or photographic techniques, or it can be estimated visually with reasonable accuracy.

Although ground cover can be assessed vertically, an estimation of solar radiation interception at a 45° view is more appropriate because it better represents an average sun angle. The sun should be at ones back when taking the readings. Confining ones views to a given small randomly chosen part of the crop permits use of a simple scale (e.g., 0-10 scale) to accurately assess cover in that portion (this can be delineated by viewing the crop through a circle made by the fingers and thumb of an outstretched arm). Ten such random views of a plot or field, when summed, give an estimation of percent ground cover. In crops with clear patterns due to drilling in rows, the angle view should be at 45° with respect to the row direction as well. When the crop is planted in wide beds, the random views should sample both the bed and the interbed regions.

Information gained:
The value in estimating ground cover lies in the realization by crop physiologists that the effect of leaf area on crop photosynthesis and dry matter accumulation depends more on solar radiation intercepted by green tissue, than on the green leaf area itself. Obviously, ground cover will depend to some extent upon the angle at which the crop is viewed, as direct radiation interception depends on the sun angle; however, for wheat in narrow rows, the percent interception of total radiation changes little with time of day (sun angle).

When full ground cover occurs, this indicates a period of constant growth as light interception is also likely to be complete and growth is primarily driven by daily radiation (assuming that other factors are not limiting).

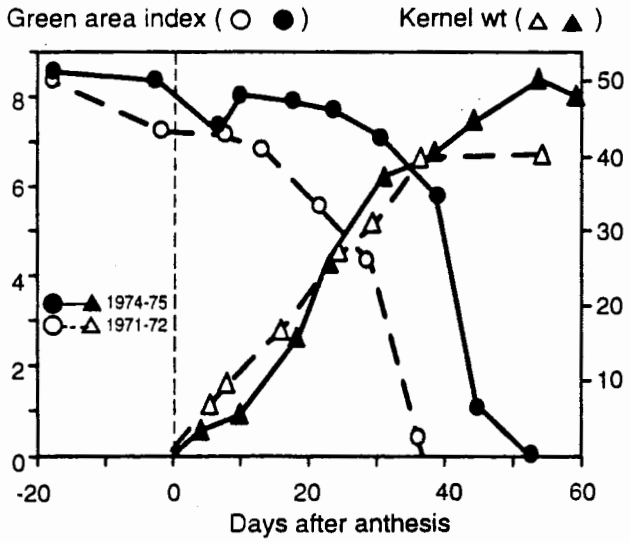

Figure 7. Changes in green area index (leaf + surface area of green stem and sheath) and kernel weight during grain-filling in hot (1971-1972) and cool (1974-1975) years. Yecora 70 under irrigation and high fertility in northwestern Mexico (from Fischer, 1983).

11.4 Light interception

Selected references:

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Light interception refers to the amount of solar radiation intercepted by foliage and other green tissues. Usually, the percentage of incoming radiation reaching the soil surface under the crop is measured (i.e., transmitted radiation, T) and intercepted percentage (INT) is given by:

$$INT = 100 - T$$

This is only approximate because, in fact, some radiation that is intercepted by leaves is actually transmitted through the leaf (and some is reflected back to the sky). The strict radiation balance (short wave) is given by:

$(I_o) = I + R + A$, where:

I_o = Downwards solar radiation above the crop,
I = Net radiation flux at the soil surface, usually assumed to be the downwards flux at the soil (T),
R = Radiation reflected, and
A = Radiation adsorbed.

Measurement is outlined in sections 11.4.2 and 11.4.3.

11.4.1 Radiation terminology—The amount of short wave radiation received at the top of the atmosphere (Ra) depends upon both latitude and time of year (**Table 1**). Part of Ra is adsorbed and scattered when passing through the atmosphere. The remainder, including some that is scattered, is identified as solar radiation (Rs). Rs is dependent on Ra, and the transmission through the atmosphere, which is largely dependent on cloud cover (Doorenbos and Pruitt 1984). **Table 2** shows the common units used in measuring radiation.

Table 1. Extra terrestrial radiation (Ra) expressed in equivalent evaporation in mm/day (Doorenbos and Pruitt 1984).

Northern Hemisphere													Southern Hemisphere										
Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sept	Oct	Nov	Dec	Lat	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sept	Oct	Nov	Dec
3.8	6.1	9.4	12.7	15.8	17.1	16.4	14.1	10.9	7.4	4.5	3.2	50°	17.5	14.7	10.9	7.0	4.2	3.1	3.5	5.5	8.9	12.9	16.5	18.2
4.3	6.6	9.8	13.0	15.9	17.2	16.5	14.3	11.2	7.8	5.0	3.7	48	17.6	14.9	11.2	7.5	4.7	3.5	4.0	6.0	9.3	13.2	16.6	18.2
4.9	7.1	10.2	13.3	16.0	17.2	16.6	14.5	11.5	8.3	5.5	4.3	46	17.7	15.1	11.5	7.9	5.2	4.0	4.4	6.5	9.7	13.4	16.7	18.3
5.3	7.6	10.6	13.7	16.1	17.2	16.6	14.7	11.9	8.7	6.0	4.7	44	17.8	15.3	11.9	8.4	5.7	4.4	4.9	6.9	10.2	13.7	16.7	18.3
5.9	8.1	11.0	14.0	16.2	17.3	16.7	15.0	12.2	9.14	6.5	5.2	42	17.8	15.5	12.2	8.8	6.1	4.9	5.4	7.4	10.6	14.0	16.8	18.3
6.4	8.6	11.4	14.3	16.4	17.3	16.7	15.2	12.5	9.6	7.0	5.7	40	17.9	15.7	12.5	9.2	6.6	5.3	5.9	7.9	11.0	14.2	16.9	18.3
6.9	9.0	11.8	14.5	16.4	17.2	16.7	15.3	12.8	10.0	7.5	6.1	38	17.9	15.8	12.8	9.6	7.1	5.8	6.3	8.3	11.4	14.4	17.0	18.3
7.4	9.4	12.1	14.7	16.4	17.2	16.7	15.4	13.1	10.6	8.0	6.6	36	17.9	16.0	13.2	10.1	7.5	6.3	6.8	8.8	11.7	14.6	17.0	18.2
7.9	9.8	12.4	15.8	16.5	17.1	16.8	18.8	13.4	10.8	8.5	7.2	34	17.8	13.1	13.5	10.5	8.0	3.8	7.2	9.2	12.0	14.9	17.1	18.2
8.3	10.2	12.8	15.0	16.5	17.0	16.8	15.6	13.6	11.2	9.0	7.8	32	17.8	16.2	13.8	10.9	8.5	7.3	7.7	9.6	12.4	15.1	17.2	18.1
8.8	10.7	13.1	15.2	16.5	17.0	16.8	15.7	13.9	11.6	9.5	8.3	30	17.8	16.4	14.0	11.3	8.9	73.8	8.1	10.1	12.7	15.3	17.3	18.1
9.3	11.1	13.4	15.3	16.5	16.8	16.7	15.7	14.1	12.0	9.9	8.8	28	17.7	16.4	14.3	11.6	9.3	8.2	8.6	10.4	13.0	15.4	17.2	17.9
9.8	11.5	13.7	15.3	16.4	16.7	16.6	15.7	14.3	12.3	10.3	9.3	26	17.6	16.4	14.4	12.0	9.7	8.7	9.1	10.9	13.2	15.5	17.2	17.8
10.2	11.9	13.9	15.4	16.4	16.6	16.5	15.8	14.5	12.6	10.7	9.7	24	17.5	16.5	14.6	12.3	10.2	9.1	9.5	11.2	13.4	15.6	17.1	17.7
10.7	12.3	14.2	15.5	16.3	16.4	16.4	15.8	14.6	13.0	11.1	10.2	22	17.4	16.5	14.8	12.6	10.6	9.6	10.0	11.6	13.7	15.7	17.0	17.5
11.2	12.7	14.4	15.6	16.3	16.4	16.3	15.9	14.8	13.3	11.6	10.7	20	17.3	16.5	15.0	13.0	11.0	10.0	10.4	12.0	13.9	15.8	17.0	17.4
11.6	13.0	14.6	15.6	16.1	16.1	16.1	15.8	14.9	13.6	12.0	11.1	18	17.1	16.5	15.1	13.2	11.4	10.4	10.8	12.3	14.1	15.8	16.8	17.1
12.0	13.3	14.7	15.6	16.0	15.9	15.9	15.7	15.0	13.9	12.4	11.6	16	16.9	16.4	15.2	13.5	11.7	10.8	11.2	12.6	14.3	15.8	16.7	16.8
12.4	13.6	14.9	15.7	15.8	15.7	15.7	15.7	15.1	14.1	12.8	12.0	14	16.7	16.4	15.3	13.7	12.1	11.2	11.6	12.9	14.5	15.8	16.5	16.6
12.8	13.9	15.1	15.7	15.7	15.5	15.5	15.6	15.2	14.4	13.3	12.5	12	16.6	16.3	15.4	14.0	12.5	11.6	12.0	13.2	14.7	15.8	16.4	16.5
13.2	14.2	15.3	15.7	15.5	15.3	15.3	15.5	15.3	14.7	13.6	12.9	10	16.4	16.3	15.5	14.2	12.8	12.0	12.4	13.5	14.8	15.9	16.2	16.2
13.6	14.5	15.3	15.6	15.3	15.0	15.1	15.4	15.3	14.8	13.9	13.3	8	16.1	16.1	15.5	14.4	13.1	12.4	12.7	13.7	14.9	15.8	16.0	16.0
13.9	14.8	15.4	15.4	15.1	14.7	14.9	15.2	15.3	15.0	14.2	13.7	6	15.8	16.1	15.6	14.7	13.4	12.8	13.1	14.0	15.0	15.7	15.8	15.7
14.3	15.0	15.5	15.5	14.9	14.4	14.6	15.1	15.3	15.1	14.5	14.1	4	15.5	15.8	15.6	14.9	13.8	13.2	13.4	14.3	15.1	15.6	15.5	15.4
14.7	15.3	15.6	15.3	14.6	14.2	14.3	14.9	15.3	15.3	14.8	14.4	2	15.3	15.7	15.7	15.1	14.1	13.5	13.7	14.5	15.2	15.5	15.3	15.1
15.0	15.5	15.7	15.3	14.4	13.9	14.1	14.8	15.3	15.4	15.1	14.8	0	15.0	15.5	15.7	15.3	14.4	13.9	14.1	14.8	15.3	15.4	15.1	14.8

Note: 1 mm = 59 cal/cm^2 = 2.4717 MJ/m^2.

In summary:

- Extra terrestrial radiation (Ra) is the total radiation arriving on a horizontal surface at the earth's atmosphere (a certain percent depending on moisture, etc., will be adsorbed by the atmosphere).

- Total radiation is the total incoming short- and long-wave radiation that arrives at the ground surface, i.e., solar radiation (Rs).

- Net radiation is the total incoming radiation less reflected incoming radiation and less the long wave radiation that is given off by the earth due to the 'black body effect'.

- Only about 50% of total solar short wave radiation is visible or photosynthetically active.

- Photosynthetically active radiation (PAR) = visible radiation = radiation between 0.4 to 0.7 μm.

11.4.2. Radiation interception by crop—Downwards radiation above (I_0) and under (I) the crop can be measured directly with radiant energy flux sensors like tube solarimeters Io-I is radiation intercepted by the crop. A single point or bar sensor is adequate. As radiation will change during the day, measurement is required throughout the day using an automatic recording device.

Table 2. Units used to measure radiation and conversions between units.

Type of measurement	Instantaneous measurement	Integrated measurement
Quantum	1 μE/s/m^2 = 1.0 mol/s/m^2 = 6.022 * 10^{17} photons/s/m^2	1 E/m^2 = 1 mol/m^2 = 6.022 10^{23} photons/m^2
Radiometric	1 W/m^2 = 1.433 * 10^{-3} cal/cm^2/min = 1.0 J/s/m^2	1 Wh/m^2 = 0.0860 cal/cm^2 = 3600 J/m^2
Photometric	1 lux = 0.0929 footcandle	
Unit conversions		
MJ/m^2 1 Cal/cm^2	= (1/0.0419) Cal/cm^2 = 1 langley = 0.0419 MJ/m^2) = (1/60) mm water evaporated	
1 Cal 1 Watt = 1 J/s	= 4.19 J	

Sources: LI-COR (1984) and Heyne (1987).

Visible radiation interception can be measured with various instruments, but particularly rapid devices have been the linear photosynthetically active radiation (PAR) sensor (also called a light bar). This sensor can be rapidly inserted under the crop to give an instantaneous readout of I; measurements above the crop (or in unshaded alleys) with the bar held horizontal are required to give I_0 so that I/I_0 can be readily calculated. It is best to have two bars and read one above the crop or in the alley at the same time as the crop reading is taken. This avoids the problem of slight variation in radiation due to cloud or atmospheric particles.

Note: With intercepted total radiation, the percentage interception will be somewhat less than for PAR, because leaves intercept non-PAR less efficiently than PAR.

For greatest accuracy, the light bar should be located at 90° to rows and the sensor window length should be a multiple of the row spacing, so as to sample all row and interrow situations equally (oblique readings may be taken at an angle to the row so that the sensor window samples an integral number of rows). Leveling of the sensor is important, especially for the above-crop reading, which have only to be made with sufficient frequency to allow for changing sun angle and cloudiness. I/I_o is not particularly sensitive to time of day or cloudiness (Monteith 1969), especially as crops approach full cover and assuming the sun never gets close to the zenith or is shining directly down the rows (e.g., north-south rows at solar noon).

The best time to make readings is within the period of 3 hours either side of solar noon (the exception would be for planting in wide beds orientated north-south where estimation of I around solar noon will give an untrue picture of the light intercepted by the crop during the rest of the day. Usually, 10-15 readings are enough to characterize a treatment and it is assumed that the mean value of I/I_o applies to all of the daytime period.

Several new instruments exist for measuring not only the quantity of light beneath the crop, but also the angular distribution of the beams reaching the ground. With built-in micro-processors, this permits the calculation of apparent LAI and leaf angle (in addition to light interception). More experience is needed with such instruments in wheat crops before the full extent of their usefulness is clear.

Information gained:
Due to the importance of radiation in determining yield (many factors affect yield primarily through their effect on radiation interception and utilization; see Charles-Edwards 1982), a detailed discussion on radiation is presented here.

It is adsorbed visible or photosynthetically active radiation (PAR_A; 0.4-0.7 μm) that is available to drive crop photosynthesis and growth; this is given by:

$$PAR_A = 0.9(I_o - I),$$

where: I_o and I in this equation refer to PAR, and which allows for the fact that about 10% of intercepted visible radiation is reflected by green leaves (hence a factor of 0.9). We ignore the small amount of radiation reflected by the soil into the crop.

Measurement of I/I_o is rapid, nondestructive and integrative—it assesses the real importance of leaf area and angle in trapping solar energy for photosynthesis. For many studies, it is more useful than LAI determination, but once after flowering, a substantial portion of the above-ground crop becomes nonphotosynthetic, it becomes less useful as it is difficult to measure.

Of particular interest is the date or stage at which a crop reaches "full light interception", usually meaning $I/I_o = 0.05$. Beyond this point, further increases in LAI will not significantly increase the amount of PAR intercepted.

Total radiation and its interception and transmission are also important because this drives transpiration and evaporation (2.41 MJ/mm water). Leaves transmit more of the non-PAR radiation than PAR so that the I/I_0 ratio for total radiation is greater than that for PAR. Total radiation downwards at the soil surface drives soil evaporation. It can be measured directly with tube solarimeters or calculated from I/I_0 for PAR with allowance for the more rapid attenuation of PAR in the canopy.

The absorbed visible solar radiation (ΣPAR_A) driving photosynthesis and crop dry matter accumulation over given periods of growth is given as follows:

$$\Sigma PAR_A = 0.5 * \Sigma Rs * 0.9 * (1-I/I_0) = 0.45 * \Sigma Rs * (1-I/I_0)$$

Note: I and I_0 refer to PAR fluxes, and the factor 0.5 converts total incident solar radiation to PAR.

where: Σ Rs is the accumulative total radiation during the period (in $MJ/m^2/d$) and $(1-I/I_0)$ is the mean fraction of "intercepted" visible solar radiation or PAR. Crop dry matter accumulation in the period (ΔTDW) is commonly related to the quantity:

$$\Delta TDW = RUE * \Sigma PAR_A,$$

where: ΔTDW is in g/m^2 and RUE, the efficiency of utilization of absorbed visible solar radiation (g/MJ) and ΣPAR_A, the accumulative absorbed PAR during the period. The value of RUE is commonly around 3 g/MJ under good growing conditions and before anthesis. After full ground cover is reached (i.e., $(1-I/I_0)_{PAR} > 95\%$), dry matter accumulation can be related directly to cumulative solar radiation (Σ Rs):

$$\Delta TDW = 0.45 * RUE * \Sigma Rs$$

Similarly, mean crop growth rate or dry weight accumulation per day (CGR) can be related to mean daily Rs or PAR_A over given periods by dividing by duration in days. Cloud cover decreases the amount of Rs. Rs is related to latitude and altitude (see section 11.4.3).

Many agronomic practices influence yield largely via changes in ΣPAR_A, through changes in LAI and hence $(1-I/I_0)$.

The photothermal quotient (PTQ) (Fischer 1985b) which is a useful measure of radiation in respect to temperature over given intervals in the crop cycle is defined as:

Total daily solar radiation/(Average daily temperature - 4.5°C).

The numerator is related to the rate of crop dry matter accumulation after full ground cover and the denominator to the rate of development; thus the PTQ ratio is related to growth per unit of development time. For example, kernel number in well watered disease free crops over a range of locations and planting dates has been shown to be closely related to PTQ in the 30-day period preceding anthesis (Fischer 1985b).

11.4.3 Estimating solar radiation—At times solar radiation (Rs) needs to be estimated (e.g., interpretation of historical records lacking radiation data, specialized equipment not being available, etc.). Three methods for calculating solar radiation are listed below. The methods use extra-terrestrial radiation, sunshine hours, and/or daily temperatures. The accuracy of these estimates varies. For example, Bell (1990) found that radiation estimated for the Yaqui Valley in Mexico from sunshine hours gives a better correlation with measured radiation than radiation estimated from daily temperatures.

Sunshine hours and extraterrestrial radiation
Solar radiation (Rs) can be estimated using:

$$Rs\ (mm/day) = (0.25 + 0.50\ n/N)\ Ra$$ (Doorenbos and Pruitt 1984)

where: the constants 0.25 and 0.50 are suitable for many regions (but not all—see Doorenbos and Pruitt 1984), n/N is the ratio between actual measured bright sunshine hours and maximum possible sunshine hours (**Table 3**).

Note: 1 mm/day = 59 cal/cm^2/day = 2.4717 MJ/m^2/day).

Evaporation and temperature
If sunshine hours are not available, an estimate of solar radiation can also be made using monthly values of potential evaporation and temperatures:

Table 3. Mean daily duration of maximum possible sunshine hours (N) for different months and latitiudes.

Northern Latitudes	Jan	Feb	Mar	Apr	May	June	July	Aug	Sept	Oct	Nov	Dec
Southern Latitudes	July	Aug	Sept	Oct	Nov	Dec	Jan	Feb	Mar	Apr	May	June
50	8.5	10.1	11.8	13.8	15.4	16.3	15.9	14.5	12.7	10.8	9.1	8.1
48	8.8	10.2	11.8	13.6	15.2	16.0	15.6	14.3	12.6	10.9	9.3	8.3
46	9.1	10.4	11.9	13.5	14.9	15.7	15.4	14.2	12.6	10.9	9.5	8.7
44	9.3	10.5	11.9	13.4	14.7	15.4	15.2	14.0	12.6	11.0	9.7	8.9
42	9.4	10.6	11.9	13.4	14.6	15.2	14.9	13.9	12.6	11.1	9.8	9.1
40	9.6	10.7	11.9	13.3	14.4	15.0	14.7	13.7	12.5	11.2	10.0	9.3
35	10.1	11.0	11.9	13.1	14.0	14.5	14.3	13.5	12.4	11.3	10.3	9.8
30	10.4	11.1	12.0	12.9	13.6	14.0	13.9	13.2	12.4	11.5	10.6	10.2
25	10.7	11.3	12.0	12.7	13.3	13.7	13.5	13.0	12.3	11.6	10.9	10.6
20	11.0	11.5	12.0	12.6	13.1	13.3	13.2	12.8	12.3	11.7	11.2	10.9
15	11.3	11.6	12.0	12.5	12.8	13.0	12.9	12.6	12.2	11.8	11.4	11.2
10	11.6	11.8	12.0	12.3	12.6	12.7	12.6	12.4	12.1	11.8	11.6	11.5
5	11.8	11.9	12.0	12.2	12.3	12.4	12.3	12.3	12.1	12.0	11.9	11.8
0	12.0	12.0	12.0	12.0	12.0	12.0	12.0	12.0	12.0	12.0	12.0	12.0

Source: Doorenbos and Pruitt (1984).

Rs (Langleys/day) = ETP/[0.0135 * (T + 17.8)] (Hargreaves 1975)

where: ETP = Potential evapotranspiration (mm total for month) (Pan A monthly totals can be used), and T = Monthly average temperature (°C) = (Tmax + Tmin)/2.

(*Note:* 1 Langley = 1 cal/cm^2.)

Extraterrestrial radiation and temperature
A third method for estimating solar radiation is using extraterrestrial radiation and temperature:

Rs (mm/day) = 0.16 Ra TD$^{0.5}$ (Hargreaves and Samani 1982)

where: Rs = solar radiation, Ra = extraterrestrial radiation (mm/day) (Table 1), and TD (°C) = Daily temperature difference = (Tmax-Tmin). Other values for the coefficient are sometime used. For example, Samani and Hargreaves (1989) found 0.165 better, and J. Williams (pers. comm.) uses 0.20.

(*Note:* 1 mm/day = 59 cal/cm^2/day = 2.4717 MJ/m^2/day)

12 Crop Stress Observations

12.1 Stress scoring

Stress symptoms can be caused by herbicide damage, nutrient imbalances, diseases, lack or excess of water, and/or temperature extremes. The specific symptoms associated with each stress should be noted (e.g. wilting, leaf rolling, chlorosis, necrosis, twisting, etc.) and the severity of the stress recorded. Stress symptoms (e.g., necrosis of the leaf) can be scored on a visual scale of 0-5, where 0 = no symptoms and 5 = severe symptoms, or alternatively on a 0-9 scale. It is preferable to use a scale that is positively related to the symptom being scored.

12.2 Plant nutrient status

12.2.1 Visual observations—Nutrient deficiencies may be identified by using the visual symptoms described in **Table 4** or in Snowball and Robson (1991). Care must be taken, however, as symptoms may be due to the presence of either toxic or inhibiting factors, unusual weather, or disease rather than a deficiency per se.

The severity of a nutrient deficiency can be visually rated based on a scale of, for example, 0-5, where 0 indicates no nutrient deficiency and 5 indicates severe deficiency. For field diagnosis, the percent area of field affected should be noted.

12.2.2 Nutrient content—The interpretation of nutrient content depends especially on stage of development and on both variety and growing conditions, however, general values for interpretation are shown in **Table 5**.

Plants can be sampled as whole plants or, especially when the plant is more developed (e.g., anthesis), it can be divided into leaves, stem, and spike. Plants can be selected at random, a specific area can be harvested as described for biomass sampling, or plants with and without symptoms can be separately sampled (the latter often being the most useful). Many systems of diagnosis concentrate on the youngest fully expanded leaf blade (YEB). However, for nitrogen diagnosis in early stages, the nitrate concentration of the basal 5 cm of the stem or pseudostem (= leaf sheaths) is commonly used (Papastylianou and Puckridge 1981), as its bulk buffers the effect of weather on NO_3^- concentration.

If micronutrients are to be analyzed, special care must be taken to avoid contamination. Hands and harvesting implements (e.g., scissors) should be washed and the collector should not be smoking. When cut, the leaves should be rinsed rapidly in preferably deionized water (although distilled is suitable) and then placed in paper bags. Samples are then dried as soon as possible at 70°C before grinding.

Information gained:
Tissue values can be used as an indication of potential deficiencies or toxicities occurring in the soil. Comparisons are often useful of plus and minus plots. Care is required, however, as antagonisms between elements may occur in the soil indicating the deficiency of one element in the plant, when

Table 4. Visual symptoms typical of deficiency.

Nutrient	Location	
	Localized on old leaves	**Whole plant**
N	Chlorosis, Turns white before necrosis	Light green
P	Chlorosis	Dark green
K	Necrotic speckling, Necrosis of tip and margin	Dark green and spindly
	Localized on middle leaves	**Whole plant**
Zn	Necrotic areas, Leaf collapse	Grey green
Mo	Chlorotic stripping, Necrotic tips and margins	Pale green
	Localized on new leaves	**Whole plant**
B	Cellular distortion, Split leaves (saw tooth)	Dark green
S	Chlorosis (not interveinal)	Light green
Mn	Light grey, flecking, Necrosis middle of leaf	Light green
Fe	Interveinal chlorosis whitening of leaf	Old leaves green new leaves pale
Mg	Chlorotic and spindly necrotic tipping	Old leaves green new leaves pale
	Localized on terminal shoot	**Whole plant**
Ca	Terminal shoot unopened	Green
Cu	Tips necrotic and twisted	Light green

Adapted from: Snowball and Robson (1991).

in reality the problem is the excess of another. Interpretation of tissue analysis should therefore be combined with interpretation of soil analysis results. Snowball and Robson (1991) summarize nutrient deficiency symptoms.

12.3 Disease scoring

Selected references

Prescott, J.M., P.A. Burnett, E.E. Saari, J. Ransom, J. Bowman, W. de Milliano, R.P. Singh, and G. Bekele. 1986. Wheat Diseases and Pests: A Guide for Field Identification. CIMMYT. Mexico, D.F., Mexico. 135 pp.

Zillinski, F.J. 1983. Common diseases of Small Grain Cereals: A guide to Identification. CIMMYT. 141 pp.

Rust Scoring Guide (Available at CIMMYT).

12.3.1 Soil- and residue-borne foliar diseases—The most important time to note the development of soil- and residue-borne foliar diseases (e.g., *Helminthosporium, Septoria*) is from heading stage on. Consequently, an initial score should be given at the time of heading and further readings taken at

Table 5. Critical and typical nutrient concentrations in wheat. Values a guide only.

Nutrient	Concentration					
	Plant stage		Whole plant (%)		Typical concentration	
Macro	Zadoks scale	Deficient	Critical	Adequate	Grain	Straw
N	30-31	<3.4	3.7-4.2	4.2-5.1	1.9	0.4
P	31	<0.2	0.3	0.3-0.5	0.4	0.08
K	57	<1.3	1.5	>1.6	0.5	1.4
S	37	<0.15	0.15	0.2-0.3	0.19	
Ca	30	<0.15	1.2	0.2-0.3	0.05	0.2
Mg	20-30	<0.10	0.15	0.15-0.3	0.17	0.2
	Plant stage		Youngest leaf (mg/g)		Typical concentration	
Micro	Zadoks scale	Deficient	Critical	Adequate	Grain	Straw
Cu	20-45	<1.3	1.3-1.5	>2.0	5 - 20	8.2
Zn	20-30	<12	14	15-70	25 - 50	20
Mn	31	<10	10-13	20-100	10 - 100	55
B		<3		3-25	5 - 50	1.1
Mo	10-45	<0.05	0.075	>0.1	0.1 - 0.3	0.1
Fe	10-45	<		25-100	50 - 250	60

Sources: Halvorson et al. (1987) and Snowball and Robson (1991).

weekly or fortnightly intervals thereafter. Two scores are given, one for the height to which the disease has risen through the canopy and the second for the average degree of infection throughou the canopy (**Figure 8**). Care should be taken not to confuse natural leaf senescence (or other physiological disorder e.g., water stress) with disease.

For some diseases that may occur in patches (e.g., *Gaeumannomyces graminis*), an indication of the area of the plot infected will also be required.

Information gained:
Diseases can cause severe yield loss. The extent of loss depends on the disease and the severity of infection relative to the time and development of the crop. Foliar diseases originating from soil inoculum rise through the crop canopy, beginning first on the older leaves and rising to the spike.

The primary effect of soilborne foliar diseases is on a reduction in leaf area for photosynthesis. This can be expressed as the area under the disease progress curve, calculated, for example, during the grain-filling period.

Selected references: Prescott et al. 1986, Zillinsky 1983, Rust scoring guide (available from CIMMYT), Stubbs et al. 1986.

12.3.2 Soilborne root pathogens—Often, the effect of soilborne root pathogens (except Rhizoctonia and the effect of nematodes) is apparent during grain-fill, when the presence of 'white heads' is generally a clear indication of their presence. Generally, a score is given to indicate simply the

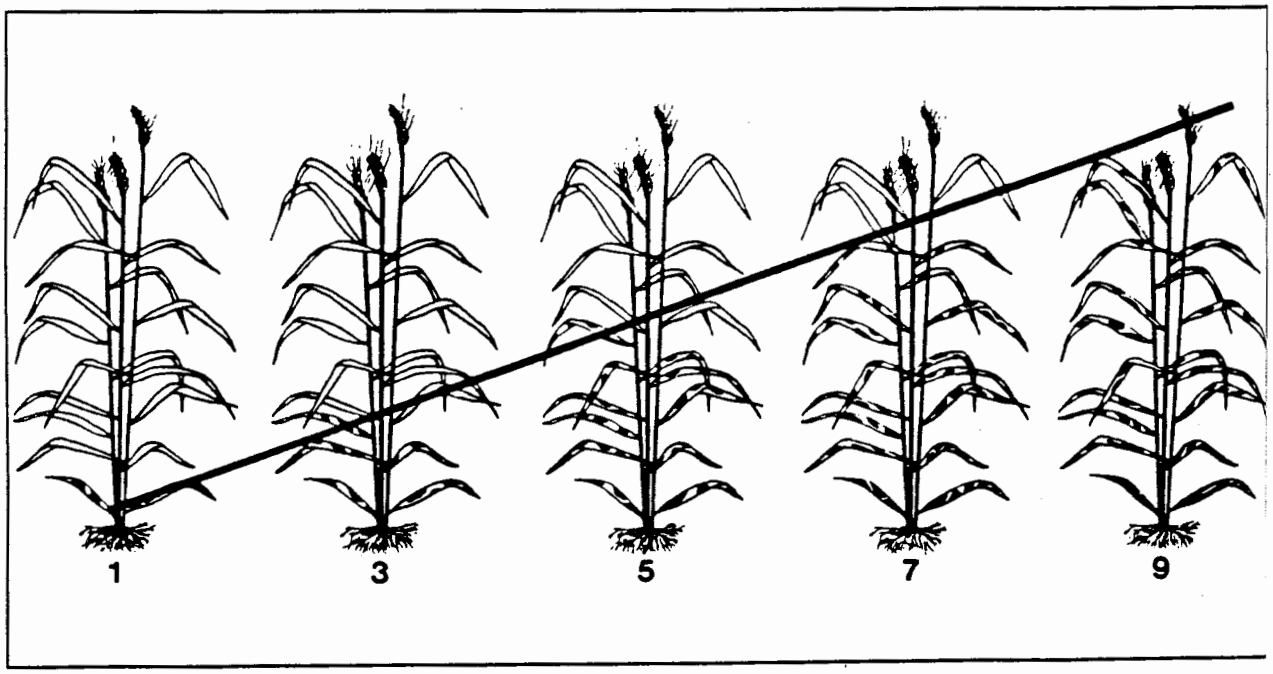

Figure 8. Scale for appraising the intensity of foliar diseases in wheat and barley (Saari and Prescott 1975).

percent of plants or culms affected. Further quantification of certain root diseases can be given by digging up plants, washing soil off the roots, and scoring based on disease symptoms on roots or the subcrown internode.

Nematodes should be evaluated when the roots are actively growing. Consequently, during the period of anthesis or before is best. For nematodes, a soil sample should be collected and nematodes estimated as follows (Cobb 1918). The soil should be dispersed in a bucket filled with water. On saturation, the soil is passed through a series of sieves (20-, 80-, and 325-mesh, which equates to 850, 180, and 44 mm). Soil is caught on the sieves, while the nematodes pass through. Counts and identification of the nematodes are conducted using a microscope.

Information gained:
Yield losses due to soilborne pathogens are often overlooked. Symptoms and disease scores can be correlated with yield reductions across treatments, but it is important to have check plots with low disease incidence. This is difficult to achieve with soilborne diseases—fungicides are not very effective and nematocides are very toxic to humans. Crop rotation is often the best way to produce clean check plots.

12.3.3 Rusts—Rusts are scored according to the percent of total leaf infected (**Figure 9**) and the type of pustule or reaction (i.e., resistant, susceptible, etc.). Again, as for soilborne diseases, it is important to note the stage of development of the plant as well as the rust infection; the earlier the disease begins to develop the greater the expected loss. The area under the disease progress curve can be calculated if repeated observations are made.

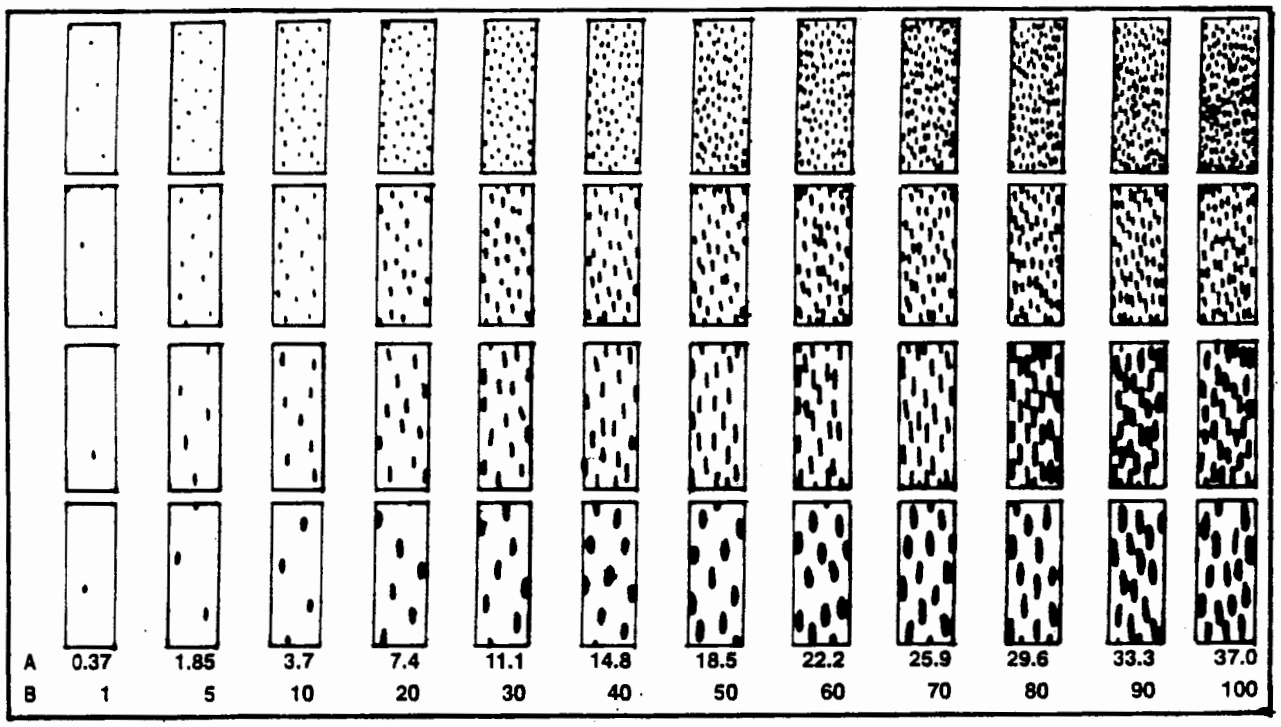

Figure 9. The modified Cobb scale: A, actual percentage occupied by rust uredinia; B, rust severities of the modified Cobb scale after Peterson *et al.* (1948).

Information gained:
Rusts can completely devastate a crop if infection begins early. Leaf rust (brown rust) generally attacks the leaf lamina and sheath. Stem rust (black rust) can attack the leaf, leaf sheath, and spike. Stripe rust (yellow rust) attacks the leaf and spike. Roelfs et al. (1992) outlines the key concepts and methods of rust management.

Rusts are airborne multi-cycle diseases and, consequently, infection can begin in any part of the canopy and develop quickly; initial infections may be patchy in a spatial sense leading to "hot spots". Rusts reduce both photosynthetic area and phloem flow in the plant.

12.4 Plant water status

Plant water status can be characterized by plant observations or measurements, including: leaf relative water content, stomatal opening/conductance, canopy temperature, leaf color change, leaf rolling, tip firing, or by decreased height, vigor, canopy cover, or leaf or stem water potential.

12.4.1 Visual evaluation—Leaf rolling is a typical symptom of drought and should be assessed in both the morning and afternoon, as afternoon rolling may often be reversible by the next morning. The dual scores allow assessment of the ability of the plant to recover from stress. Another symptom of drought stress is tip firing. Some care is required, however, as tip firing can also be characteristic of certain genotypes.

A light bar (see section 11.4.2) can be also be used to measure light interception by the canopy, as leaf length is often reduced under stress (as leaf angle, and thus canopy cover can be altered). This is useful when a control can be compared with the stressed treatment to assess rates of canopy expansion and determine the point of onset of water stress.

Other symptoms of drought stress include decreased plant height and the development of a grey-blue color (due to increased wax production).

Information gained:
Many of the water stress measurements (see below) are indirect. Along with the direct measures, like plant water status, they are most useful in comparing treatments at a given point in time. Absolute measures of drought stress for comparisons over a long period of the crop's life, or with other locations or years, require some integrative measures. One such measure is the cumulative days for which leaf water status (at some standard time of the day, usually dawn, but also mid-afternoon) is below some pre-defined threshold. The maximum degree of leaf water stress reached is another, though less satisfactory, measure. The time-integrated area under the stress threshold is another such measure. Considerable success has been achieved with the integrated stress index derived from leaf canopy temperature readings (e.g., Idso 1982, Clawson et al. 1989). See sections 12.4.5 and 12.4.6. Since canopy temperature measurement is rapid, this is in fact the most feasible method at present to evaluate stress, but requires measurement of air vapor pressure deficit (see section 12.4.7) as well as knowledge of the development-dependent canopy temperature baselines of wheat (Idso 1982).

Finally, the pattern of available water depletion in the crop root zone and cumulative crop water use relative to a well watered control have traditionally given a physiologically-sound integration of the stress regime that is satisfactory for most purposes (see Bell and Fischer 1993).

12.4.2 Stomatal conductance—Stomatal conductance is measured directly with a diffusion porometer or indirectly with an air flow porometer (Fischer et al. 1977, Meidner and Mansfield 1968). Two to four measurements/plot (across four replications) should be made on the youngest fully emerged leaf lamina receiving full sunlight.

Information gained:
Stomatal conductance is very sensitive to plant water status when other factors are equal.

12.4.3 Leaf relative water content—Leaf relative water content (LRWC) is measured by cutting the leaf lamina and immediately weighing (FW). Duplicate samples/plot should be collected (if there are four replications). The leaf is then rehydrated by having its base in water for at least 3 hours and re-weighed (RFW) then dried (at 70°C) and weighed again (DW). The samples should be collected either at sunrise or mid-afternoon, being the time at which highest and lowest LRWC are usually recorded.
Calculate as:

$$LRWC = [(FW - DW) * 100]/(RFW - DW)$$

Information gained:
Although LRWC had been essentially abandoned for other methods (e.g., leaf water potential), Sinclair and Ludlow (1985) found crop performance to be more related to LRWC than other water status measurements.

12.4.4 Leaf water potential—Leaf water potential can be measured using a pressure chamber. The pressure required to expel 'sap' from a recently cut surface (usually a stem or leaf xylem bundle) is related to the amount of water in the leaf. At least eight youngest fully expanded leaves (YFEL) should be sampled per treatment (e.g., two leaves/plot with four replications). The measurements can be made either pre-dawn to assess lowest stress values and/or mid-early afternoon to assess maximum stress values (the former reflect soil water status in the root zone). Samples should be measured immediately as collected—measurement is usually conducted in the field beside the plots. However, if some sample transport is required, samples can be stored for a short time (< 5 minutes) in a humid bag (moisten the bag by blowing into it)—keep the bag out of the sun.

Due to the changes that can occur throughout the day in leaf potential, it is extremely important that any comparisons of treatments be conducted at the same time of day.

Information gained:
Differences in leaf water potential between treatments or with time can be related to changes of plant water status (See Fischer et al. 1977).

12.4.5 Soil, air and canopy temperature—Both soil and air temperature can be measured with a small thermometer or thermocouple designed for that purpose (Rawlins and Campbell 1982). However, the process for measuring soil temperature can be relatively tedious using thermocouples, especially if more than a few plots are involved. Although soil probe thermometers are easy to use and relatively quick to equilibrate for point readings (i.e., 2-5 minutes), they lack the precision of a thermocouple. Most commonly soil temperature is measured in the germination zone (i.e., 0-5 cm). However, if temperature changes through the profile are required, then measurements through the profile up to at least 50 cm depth are recommended.

An effective means of measuring canopy temperature is outlined in section 12.4.6.

Information gained:
Air temperature is critical for determining the rate of crop development (heat units). The maximum rate of photosynthesis occurs between 20-30°C for most temperate plants, while prolonged exposure to temperatures above 35°C often causes damage (Gusta and Chen 1987). Optimal soil temperature is important in terms of germination conditions. An interesting development in the area of temperature-crop development is the concept of evaporative cooling. The basis of this effect is that a well grown transpiring crop reduces the temperature considerably below that of the air and so slows crop development below that otherwise expected and may increase yield potential. The effect seems to be more important in irrigated areas of high temperatures and low relative humidities (e.g., Sudan).

12.4.6 Infrared thermometer—Infrared thermometry measures temperature of the object (leaves) in view of the thermometer. Leaf or canopy temperature is usually expressed relative to air temperature (known as leaf to air temperature depression) and well watered crops usually are cooler than air, a difference that increases as the air becomes drier. The readings should be taken with the gun at an angle of 15 degrees to the canopy and aimed towards the center of the plot. A minimum of two readings should be taken per plot during the hottest part of the day for maximum differentiation of treatments. The 'gun' should never be pointed at the sun (or false readings will subsequently be given). Interference from the soil background should be avoided. Another problem is that air temperatures are cooler over irrigated soils, so that direct comparisons should not be made between treatments that, at the time of measurement, have different surface moisture. Measurement of vapor pressure deficit (see section 12.4.7) at the same time as the air-leaf temperature difference is recorded helps understanding the absolute levels of stress by permitting calculation of the water stress index. It may be necessary to measure net radiation levels for an accurate absolute interpretation of canopy temperature depression (see Smith et al. 1986).

Information gained:
A greater difference between air and canopy temperature is associated with greater transpiration by the crop and less soil water stress in comparisons of irrigation treatments or other treatments, which may affect plant water stress levels. Under such conditions, canopy temperature depression is closely related to stomatal conductance. Recent data of Balota et al. (1993) also indicate that canopy temperature depression could be used as a screening trait for heat tolerance in wheat. Canopy temperature measurements can be used to schedule irrigations provided other weather variables are considered too, in particular vapor pressure deficit.

12.4.7 **Vapor pressure deficit**—The vapor pressure deficit (VPD—mbar or kPascals) is the difference between saturated vapor and actual vapor pressure of the air. The vapor pressure can be estimated from dry and wet bulb temperatures using the equations given in **Table 6**, which apply to unventilated bulbs. To measure the wet bulb temperature, the specially designed bulb needs to be shielded from radiation. Dry bulb temperature is the temperature of the air free from radiation effects.

Information gained:
The VPD is key in determining crop water use efficiency (higher VPD = lower efficiency), in interpreting canopy temperature (higher VPD = cooler crop canopies; see Jalali-Farahani et al. 1993, Smith et al. 1986), and in determining crop water needs (see Doorenbos and Kassam 1979, Doorenbos and Pruitt 1984).

12.5 Temperature stresses

12.5.1 **Heat stress**—The symptoms of heat stress can be difficult to distinguish from those of drought stress, particularly as the two often occur together. However, in addition to the symptoms listed above, organs are reduced in size, the spike may be deformed, the head may show sterility (see section 6.5), the awns may not emerge properly from the boot, and senescence is generally very early.

Table 6. Algorithms for calculating vapor pressure and saturation vapor pressure of air in mb.

VPair = VPsat*(Twet) - A P (Tdry-Twet), where:
P is pressure in mb (1013 at sea level and - 1mb per 10 m altitude change)
A = 0.00066 (1+0.00115 Twet)
Twet = wet bulb temperature (°C)
Tdry = dry bulb temperature (°C)

Saturation vapor pressure (mb) of air
Short equation:

$$VPsat = 6.1078 \exp[(17.269 \text{ Av temp})/(\text{Av temp} + 137.3)]$$

Long equation

$$VPsat = a + bT + cT^2 + dT^3 + eT^4 + fT^5 + gT^6,$$

where

$a = 6.1078$, $b = 4.4365\ 10^{-1}$
$c = 1.4289\ 10^{-2}$ $d = 2.6505\ 10^{-3}$
$e = 3.0312\ 10^{-6}$ $f = 2.0341\ 10^{-8}$
$g = 6.1368\ 10^{-11}$

Sources: WMO (1965, 1981, 1985a,b).

Information gained:
Not enough is known about the direct effects of heat per se since the indirect effects of hastened development are so dominant and the direct effects appear to operate at the cellular level (e.g., membranes become leaky).

12.5.2 **Cold (frost) stress**—Chorosis and/or necrosis of affected tissue are the primary symptoms of cold stress. A light frost may only affect new tissue, resulting in a banding or striping on the leaves or spike (Prescott et al. 1986). Spike sterility or early senescence can also be apparent, if a frost occurs during anthesis and grain-filling.

Information gained:
The effects of frost are strongly dependent upon the time of frost damage. Plants may recover from early damage, especially when the growing point is protected below the soil surface. However, frost damage during elongation and particularly during anthesis can lead to severe or complete crop failure.

12.6 Weed competition and control

Besides the direct effects of weeds in terms of competition for light, nutrients and water, and/or allelopathy, weed growth during fallow periods will influence water extraction from the soil profile.

12.6.1 Weed populations and biomass—Weed populations can be assessed by randomly placing at least two quadrats (minimum area of 1.0 m^2/quadrat) in each plot and recording the number of each weed species occurring within the quadrats. The number of each species of weed can then be converted to a population per hectare.

To measure weed biomass, the total amount of material within a specified area (as described above) should be cut at ground level and divided into crop and weed portions. The samples should then be dried at 70°C before weighing. Weed biomass is determined at a key stage of development (e.g., anthesis or maturity) and provides an absolute measure of the amount of weed growth, and hence the competitive outcome. Crop dry matter is likely to be reduced in an absolute sense by an amount similar to that of the weight of weed growth (or more if alleleopathic effects occur).

12.6.2 Weed scores—Weed intensity can be evaluated by a variety of scales. **Tables 7ABC** show various scales that can be used to assess weed populations. The easiest method is to calculate the percentage of the plot covered by the different weed species, although more accurate is to measure the percent of biomass in weeds (either at different stages during the crop or just at harvest—see section 12.6.1).

12.6.3 Phytotoxicity of chemicals—Symptoms of phytotoxicity, resulting for example from the use of certain pesticides, often include leaf flecking, head distortion, chlorosis, necrosis, and stunting. Phytotoxicity can be estimated by various scales as shown in Tables 7ABC. Plants often grow out of a toxicity, so readings should be recorded on more than one occasion.

Information gained:

Phytotoxicity due to control of weeds or pests with chemicals can occur if the chemical is applied incorrectly, at the wrong time, or to a wheat variety susceptible to the chemical. Significant yield losses can result.

Table 7A. Qualitative (0-100) scale used for the evaluation of weed populations (W) and/or crop phytotoxicity (P).

Rating	Description of main categories	Detailed description	
0	No effect	W	No weed control
		P	No crop reduction or injury
10		W	Very poor stand
		P	Slight crop discoloration or stunting
20	Slight effect	W	Poor weed control
		P	Some crop discoloration, stunting or stand loss
30		W	Poor to deficient weed control
		P	Crop injury more pronounced, but not lasting
40		W	Deficient weed control
		P	Moderate injury, crop usually recovers
50	Moderate effect	W	Deficient to moderate weed control
		P	Crop injury more lasting, recovery doubtful
60		W	Moderate weed control
		P	Lasting crop injury, no recovery
70		W	Weed control somewhat less than satisfactory
		P	Heavy crop injury and stand loss
80	Severe effect	W	Satisfactory to good weed control
		P	Crop nearly destroyed, a few surviving plants
90		W	Very good to excellent weed control
		P	Only occasional live crop plants left
100	Complete effect	W	Complete weed control
		P	Complete crop destruction

Table 7B. Comparison of differenet scales used for the evaluation of weed populations and/or phytotoxicity showing qualitative values for weed control and crop phytotoxicity scoring.

Percent	Scales			EWRS[a]	% Activity
	0-10	0-5	1-5		
0-10	0-1	0	1	1	100
10-20	1-2			2	99.9-98
		1			
20-30	2-3		2	3	97.9-95
		2			
30-40	3-4			4	94.9-90
40-5	04-5				Limit of acceptability
		3	3		
50-60	5-6			5	89.9-82
60-70	6-7			6	81.9-70
		4	4		
70-80	7-8			7	69.9-55
80-90	8-9			8	54.9-30
		5	5		
90-100	9-10			9	29.9-0

[a] European System of Weed Control and Crop Injury Scoring (EWRS) is a logarithmic scale (See Table 7C).

Table 7C. Suggested European System of Weed Control and Crop Injury (EWRS) (a logarithmic scale). (Note that the system is for either weed control or crop damage.)

Rating	Effect on Weeds	Effect on Crop
1	Complete kill	No effect
2	Very good	Very light
3	Good	Light symptoms
4	Sufficient in practice	Symptoms not reflected in yield
5	Medium	Medium
6	Fair	Fairly heavy damage
7	Poor	Heavy damage
8	Very poor	Very heavy damage
9	No effect	Complete kill

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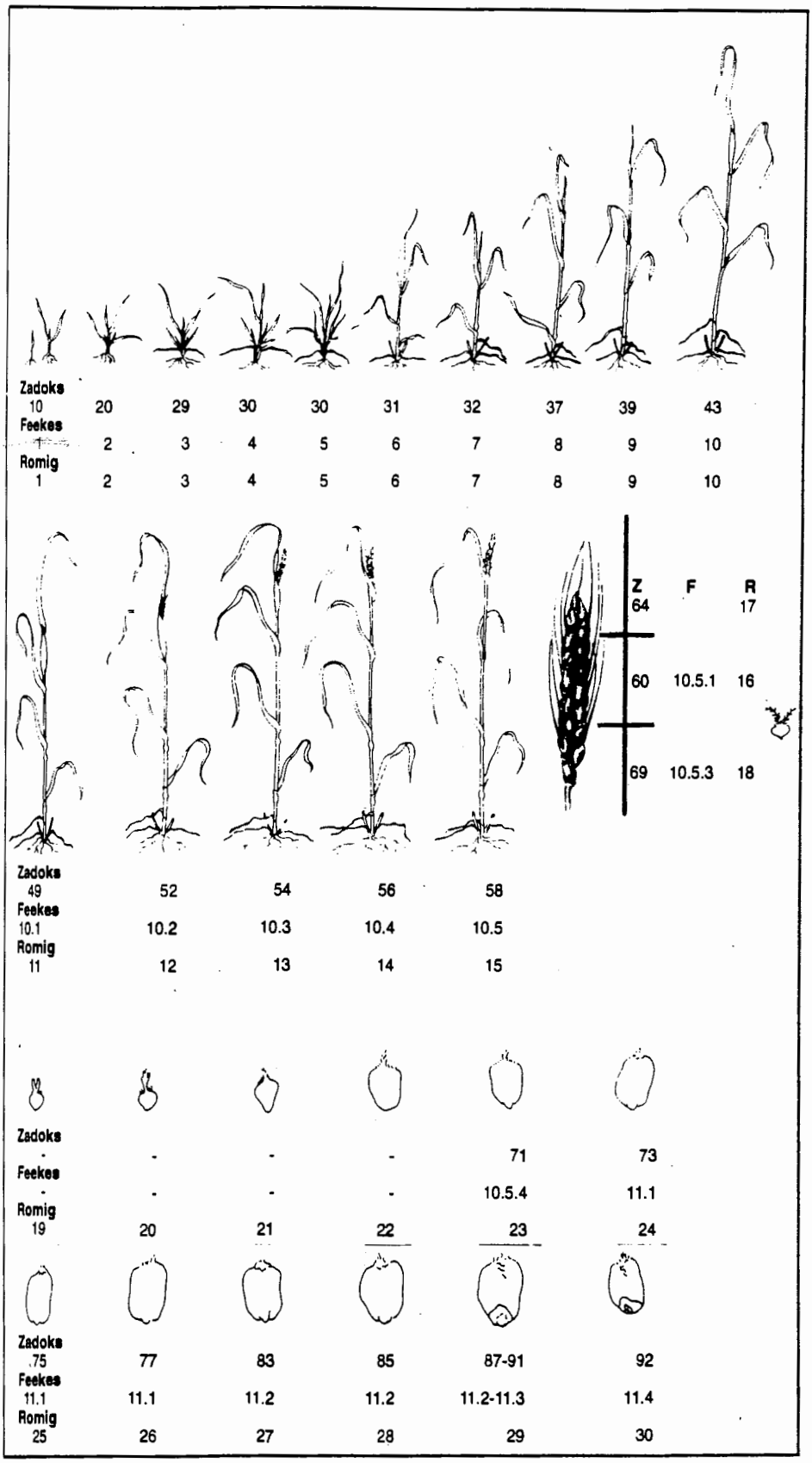


Appendix 1. Descriptions of the growth stages for wheat using the Zadoks, Feekes, and Romig Scales.

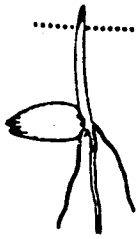
Description	Growth scale		
	Zadoks	Feekes	Romig
Dry seed	00	–	–
Start of imbibition	01	–	–
Leaf just at coleoptile	09	1	–
Firs leaf Through coleoptile	10	1	1
First leaf unfolded	11	–	1
Two leaves unfolded	12	–	–
One or more eaves unfolded	19	–	–
Main shoot only	20	2	–
Main shoot and 1 tiller	21	3	2
Main shoot and 2 tillers	22	3	2
Main shoot and 9 or more tillers	29	3	3
Pseudo-sterm	30	4-5	4-5
1st node detectable	31	6	6
2nd node detectable	32	7	7
6th node detectable	36	–	–
Flag leaf just visible	37	8	8
Flag leaf ligule/collar just visible	39	9	9
Flag leaf sheath extending	41	10	–
Boot just visibly swollen	43	10	10
Boot swollen	45	10	10
Flag leaf sheath opening	47	10.1	–
First awns visible	49	10.1	11
1st spikelet of inflorescence just visible	50	10.1	–
1/4 of inflorecence emerged	52	10.2	12
1/2 of inflorecence emerged	54	10.3	13
3/4 of inflorecence emerged	56	10.4	14
Emergence of inflorecence completed	58	10.5	15
Beginning of anthesis	60	10.5.1	16
Anthesis half-way	64	–	17
Anthesis complete	69	10.5.3	18
Kernels near middle of head 1/8 formed	–	–	19
Kernels near middle of head 1/4 formed	–	–	20
Kernels near middle of head 1/2 formed	–	–	21
Kernels near middle of head 3/4 formed	–	–	22
Caryopsis watery ripe	71	10.5.4	23
Early milk	73	11.1	24
Medium milk	75	11.1	25
Late milk	77	11.1	26
Early dough	83	11.2	27
Soft dough	85	11.2	28
Hard dough	87	11.2	29
Caryopsis hard, 16% water	91	11.3	29
Caryopsis hard	92	11.4	30

Appendix 2. Plant parts and Zadoks development scale.

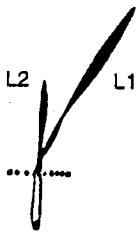
Adapted by M. Stapper from Ann. appl. Biol. 93: 221-234

DC 05
radicle emerged
from seed

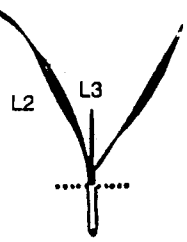
DC 07
coleoptile emerged
from seed



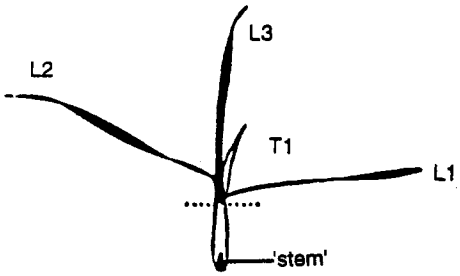
DC 10
coleoptile at
surface



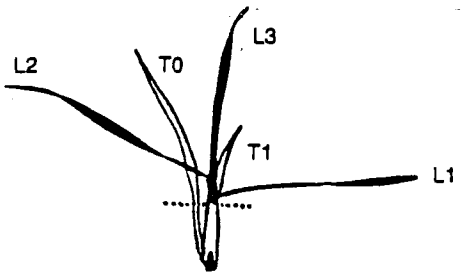
DC 11
one leaf on main (L1)
second leaf appears (L2)



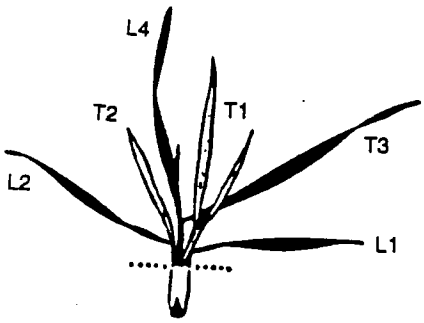
DC 12
two leaves on main
third leaf appears (L3



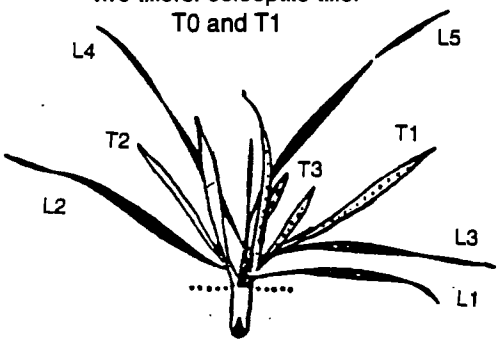
DC 13, 21
three leaves on main shoot
first tiller appears (T1)



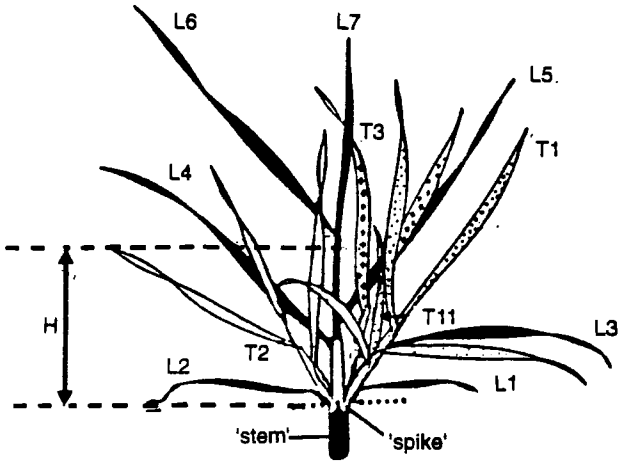
DC 13, 22
three leaves on main shoot
two tillers: coleoptile tiller
T0 and T1



DC 14, 22
four leaves on main shoot
two tillers: T1 with two leaves
T2 with one leaves

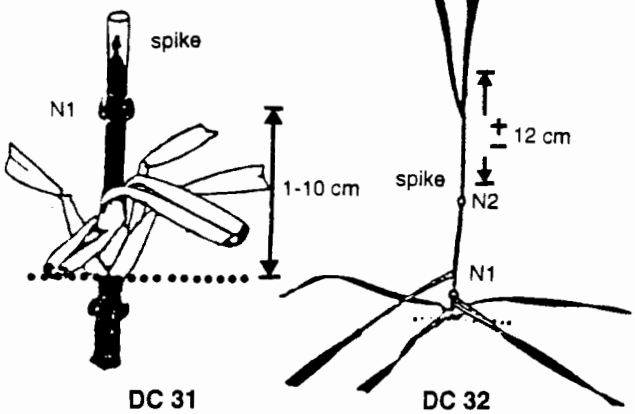


DC 15, 23
three leaves on main shoot
three tillers: T1 with three leaves
T2 with two leaves
T3 with one leaf



DC 17, 24
seven leaves on main stem
four tillers: T1 with four leaves and 1 tiller (T11)
T2 with three leaves
T3 with two leaf

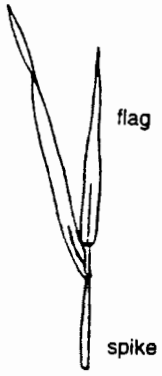
or DC 30
if height (H) is 12 cm or more and the true stem has elongated raising the young spike above the soil surface



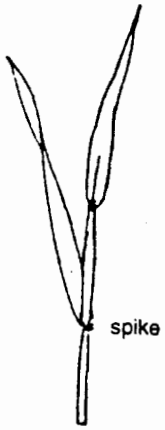
DC 31
first node detectable (N1)

DC 32
second node (N2)

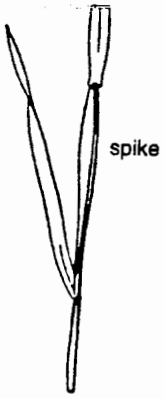
DC 49
awns visible
last node formed
shoot has five green leaves



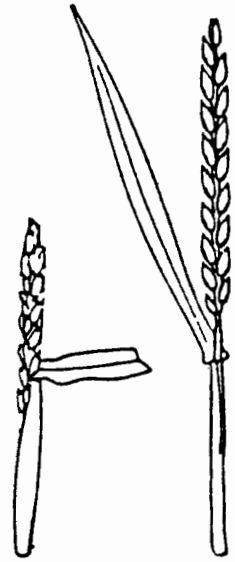
DC 39
flag leaf fully emerged



DC 43
spike opposite collar
of second last leaf



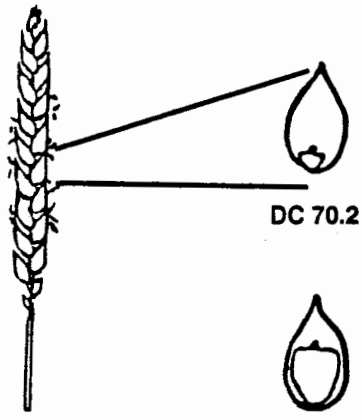
DC 45



DC 55
half of spike
visible

DC 60
full spike
visible

floret from centre of spike



DC 65
yellow anthers visible
on 50% of the spikes

DC 70.2

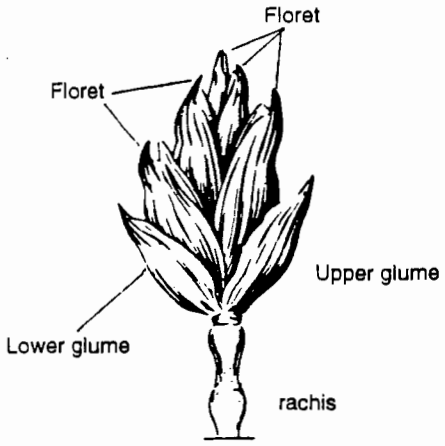
DC 70.8

DC 70.5

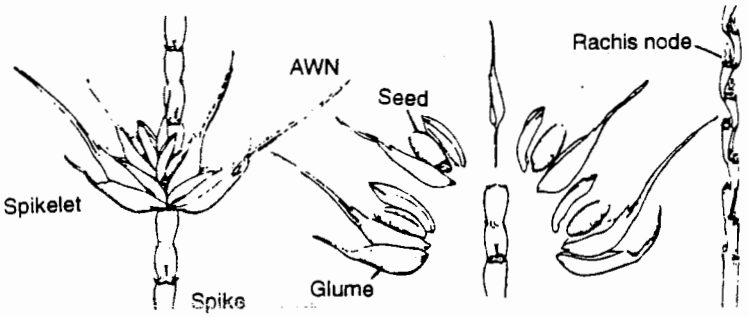
DC 71

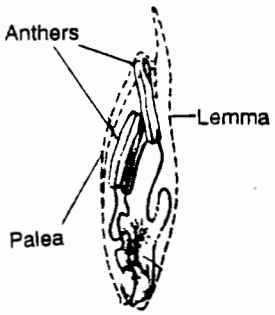
Appendix 2. Continued.

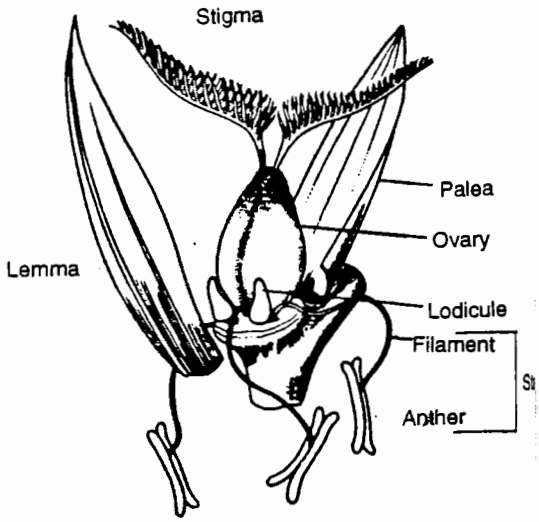
Adapted from: Stoskopf (1985) and Lerston (1987)



Spikelet







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Appendix 3. Table of useful field conversions and units.

To convert column 1 into column 2, multiply by	Column 1 SI unit	Column 2 non-SI unit	To convert column 2 into Column 1, multiply by
	Length		
0.621	kilometer, km (10^3 m)	mile, mi	1.609
1.094	meter, m	yard, yd	0.914
1.0	micrometer, um (10^{-6} m)	micron, u	1.0
3.94×10^{-2}	millimter, mm (10^{-3} m)	inch, in	25.4
3.94×10^{-1}	cm (10^{-2} m)	inch, in	2.54
	Area		
2.47	hectare, ha	acre	0.405
2.47	square kilometer, km^2	acre	4.05×10^{-3}
2.47×10^{-4}	square meter, m^2	acre	4.05×10^3
10.76	square meter, m^2	square foot, ft^2	9.29×10^{-2}
	Volume		
6.10×10^4	cubic meter, m^3	cubic inch, in^3	1.64×10^{-5}
2.84×10^{-2}	liter, L (10^{-3} m^3)	bushel, bu	35.24
0.265	liter, L (10^{-3} m^3)	gallon (US)	3.78
0.220	liter, L (10^{-3} m^3)	gallon (imperial)	4.546
9.73×10^{-3}	cubic meter, m^3	acre-inch	102.8
35.3	cubic meter, m^3	cubit foot, ft^3	2.83×10^{-2}
	Mass		
2.205	kilogram, kg	pound, lb	0.454
10^{-2}	kilogram, kg	quintal, q	10^2
1.1×10^{-3}	kilogram, kg	ton (2000 lb), ton	907
1.102	megagram, Mg (ton)	ton (2000 lb), ton	0.907
9.843×10^{-4}	kilogram, kg	ton (2240 lb)	1.016×10^3
	Yield and rate		
0.893	kilogram/hectare, kg ha^{-1}	pound/acre, lb $acre^{-1}$	1.12
1.49×10^{-2}	kilogram/hectare, kg ha^{-1}	bushel/acre, 60 lb	67.19
1.59×10^{-2}	kilogram/hectare, kg ha^{-1}	bushel/acre, 56 lb	62.71
0.107	liter/hectare, L ha^{-1}	gallon/acre	9.35
893	megagram/hectare, Mg ha^{-1}	pound/acre, lb $acre^{-1}$	1.12×10^{-3}
0.446	megagram/hectare, Mg ha^{-1}	ton (2000 lb) per acre ton $acre^{-1}$	2.24
0.399	megagram/hectare, Mg ha^{-1}	ton (2240 lb) per acre	251
2.24	meter/second, m s^{-1}	mile/hour	0.447
	Pressure		
9.90	megapascal, MPa (10^6 Pa)	atmosphere	0.101
10	megapascal, MPa (10^6 Pa)	bar	0.1
1.45×10^{-4}	pascal, Pa	pound/square inch, lb in^{-2}	6.90×10^3

Appendix 3. Continued

To convert column 1 into column 2, multiply by	Column 1 SI unit	Column 2 non-SI unit	To convert column 2 into Column 1, multiply by
		Temperature	
1.00 (K-273)	Kelvin, °K	Celcius, °C	1.00 (°C + 273)
(9/5 °C) + 32	Celcius, °C	Fahrenheit, °F	5/9 (°F - 32)
		Electrical conductivity	
10	siemen/meter, S m^{-1}	millimho per centimeter, mmho cm^{-1}	0.1
		Water measurements	
9.73×10^{-3}	cubic meter, m^3	acre-inches, acre-in	102.8
9.81×10^{-3}	cubic meter/hour, $m^3 h^{-1}$	cubic feet/second, $ft^3 s^{-1}$	101.9
4.40	cubic meter/hour, $m^3 h^{-1}$	U.S. gallon/minute, gal min^{-1}	0.227
		Concentrations	
1	centimole/kilogram, $cmol_c kg^{-1}$, ion exchange capacity	milliequivalents/100 grams, meq $100g^{-1}$	1
0.1	gram/kilogram, g kg^{-1}	percent, %	10
1	megagram/cubic meter, Mg m^{-3}	gram/cubic centimeter g cm^{-3}	1
1	milligram/kilogram, mg kg^{-1}	parts per million, ppm	1
		Energy, Work, Quantity of Heat	
0.239	joule, J	calorie, cal	4.19
10^7	joule, J	erg	10^{-7}
2.387×10^{-5}	joule/square meter, J m^{-2}	calorie/square centimeter (langley)	4.19×10^4
10^{-5}	newton, N	dyne	10^{-5}
1.43×10^{-3}	watt/square meter W m^{-2}	calorie/square centimeter minute (irradiance), cal cm^{-2} min^{-1}	698
23.87	(Watt = 1 Js^{-1}) MJ m^{-2}	cal, cm^{-2}	0.0419

Appendix 4. Useful agronomic conversions and sprayer calibrations.

$g\ m^{-2} \times 10 = kg\ ha^{-1}$

$kg\ ha^{-1}/10 = g\ m^{-2}$

$g\ m^{-2}/(100) = t\ ha^{-1}$

$t\ ha^{-1} \times 100 = g\ m^{-2}$

$kg\ m^{-2} \times 10 = t\ ha^{-1}$

$t\ ha^{-1}/(10) = kg\ m^{-2}$

1 acre = 0.4047 ha 1 ha = 2.24 acre
1 lb = 0.4536 kg 1 kg = 2.20 lb
1 bushel wheat = 27.2 kg

Sprayer calibration (L/ha)

$$= \frac{\text{output/nozzle (L/min) x number of nozzles x 10000}}{\text{speed (m/sec) x width of swath (m) x 60}}$$

Calculating herbicide to be added to a solution

Sprayer tank capacity (L) = TC

Calibration (L/ha) = C

Desired rate of product (L/ha) = DR

Required rate of product (L/tank) = TC x (DR/C)

Desired rate of active ingredient (L/ha) = DAI

Active Ingredient (%) = AI

Required rate of product (L/tank) = TC x [DAI/(C x AI)]

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