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A Review on Plant Root Architecture and Methods for Measuring Root Growth Parameters

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Abstract

The longitudinal arrangement of root systems, as well as length, diameter, and hairs are referred to as root architecture. Plant root system plays key role in regulating plant growth and development. Root tuber crops (RTCs) that are vegetatively propagated offer food security for enormous populations. Root morphology is affected by soil, climatic, and plant factors. Parameters normally used to express root growth and distributions are root number, weight, surface area, volume, diameter, length, and the number of tips. A number of factors have been examined to influence root- shoot ratio; examples are deprivation of Nitrogen, and low supplies of Phosphorus, Potassium, and Magnesium. Most methods for measuring root growth parameter is labour intensive. Hence, new and improved methods for meas uring root growth in-situ have now been developed. Researches in crop physiology are always in progress. Hence, crop physiologists should keep themselves abreast of the most recent findings in their field of specialization.

Keywords: *root, architecture, morphology, root-shoot ratios, growth*

INTRODUCTION

Roots obtain mineral nutrients from the soil and transport them to the shoot. Solutes (including nutrient ions) are conveyed through the roots to the xylem vessels by means of apoplastic or symplastic pathways (Leigh, 2017). Roots are indispensable for plant efficiency and perform a diversity of roles including water and nutrient uptake, establishing associations with other microbes in the rhizosphere, attaching the plant to the soil, and stand-in as storage organs. The diverse associations of a root with its surroundings rest on its organization and configuration, from the cellular to entire plant level (Smith and De Smet, 2012). One of the utmost important plant morphological characters that justifies special attention is the root architecture; and symbolizes the longitudinal outline of root systems which is made up of root length, lateral roots (Khan *et al*., 2016), diameter, hairs (Fageria *et al*., 2006) and spread (Khan *et al*., 2016). Root systems play a main part in directing plant growth and development owing to their importance in engagement of water and nutrients (Barley, 1970; Lynch, 1995). Roots also offer mechanical support to plants from seedling through maturity to development. Furthermore, roots are important in nitrogen (N) chemistry whereby it can be conducted to shoots via xylem, either as inorganic nitrogen or as amino acids and amides (Fageria *et al*., 2006). Plant roots are involved in the manufacture of growth substances and hormones like cytokinin that may be vital in leaf function and possibly grain development (Evans and Wardlaw, 1976). Root morphology and physiology are closely connected to the growth of the aerial parts and shoot productivity (He,

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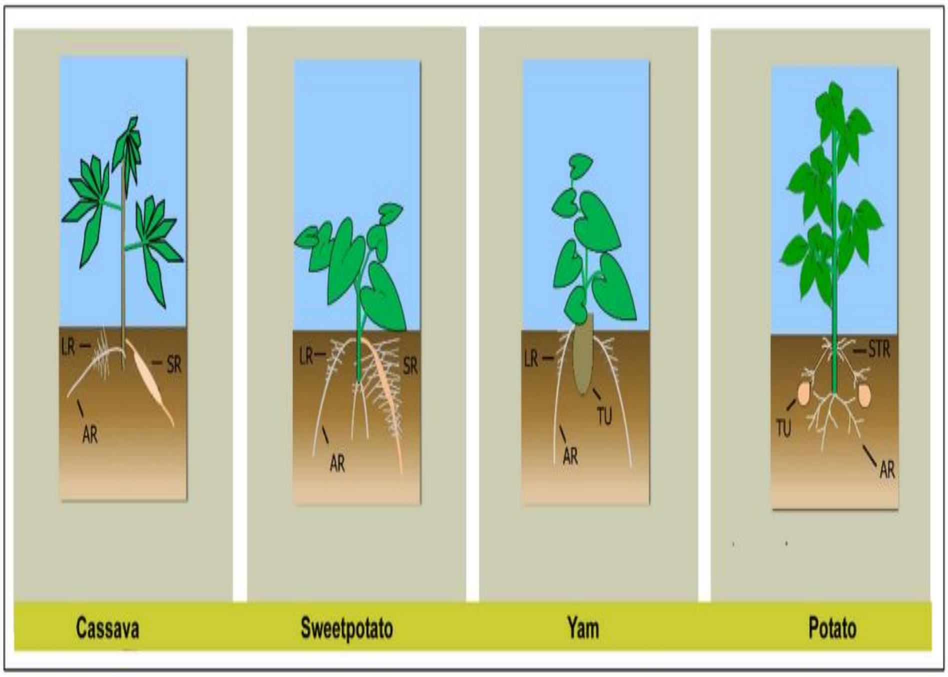
2016). An enhanced data on root architecture and root development dynamics is central for improving efficiency of annual crops (Khan *et al*., 2016).

ROOTS ARCHITECTURE FOR ROOT AND TUBER CROPS

Knowledge on root architecture and the mechanisms of its development provide an idea on how to manipulate and exploit the different root characters in order to advance plants' adaptation to fluctuating climates and rise yields to face the challenge of teeming world human population (Smith and De Smet, 2012). Root tuber crops (RTCs) such as potato (*Solanum tuberosum*), sweet potato (*Ipomoea batatas*), cassava (*Manihot esculenta*), and yam (*Dioscorea* spp.) that are vegetatively propagated offer food security for enormous populations, particularly in sub-Saharan Africa where various resource-poor small holder farmers offer the majority of food. The scanty literature existing also mostly pay attentions on the growth and development of the storage root or tuber, principally at harvest and post-harvest appraisals, rather than the complete root system. The following are the root systems of four major RTCs, based on available literature (Figure 1) (Khan *et al*., 2016).



AR-adventitious root; LR-lateral root; SR- storage root; SR- stolon root

Figure 1. Plant toot architecture displaying diverse root forms. Figure adapted from Villordon *et al*., 2014 as cited in Khan *et al*., 2016 with slight modification.

ROOT MORPHOLOGY

Hackett and Rose (1972) and Fitter (1982) reported that utmost studies on root have paid attention on root morphology and topology. Root characteristics such as length, mean diameter, surface area, and mass have been used to quantitatively and qualitatively designate morphology and topology (Costa *et al,* 2002). Components of root morphology such as root length, number of root tip, root diameter, root surface area, and root volume differ significantly depending on the plant species, soil composition, and water and mineral nutrients availability (He, 2016). Cereals and grasses are usually known as monocots and legumes as dicots. Monocots have single cotyledons or leaves arising at the first node of the lead shoot or stem, and dicots have two cotyledons evolving at their first node. Monocots have fibrous root system, whereas dicots have tap roots. The fibrous root systems of monocots consist of the following components:

SEMINAL ROOT, NODAL ROOTS, AND LATERAL ROOTS

Seminal roots: Seminal roots develop from primordial within seeds and help anchor the young seedling and provide it with nutrients and water before the nodal root system is developed. Seminal root growth slowly after seedling emergence (Litch and Clemens, 2023). The narrower is the seminal root angle, the longer is the seminal root length, thus permitting the roots to simply obtain remaining moisture in deep soils and increase the drought tolerance of wheat (Gao *et al*., 2016; Hodgkinson *et al*., 2017).

Nodal roots: Nodal roots develop adventitiously from inferior stem nodes. All adventitious roots of stem origins are termed nodal roots to differentiate them from other adventitious root that emerged from the mesocotyl or elsewhere on the plant (Fageria *et al*., 2006). Nodal roots are identified by the node number from which they come from. Nodal roots may be efficient or non-efficient (Thomas and Kaspar, 1997). A handful studies have been conducted on the nodal root system due to its disappearance at the primary seedling period under indoor environmental condition (Xu *et al*., 2021).

Lateral roots: Lateral roots are formed after the emergence of the embryo and originate from existing roots, and are usually branched (Arif *et al*., 2019) and develop from first- order roots are grouped as second- order roots, and supplementary roots that develop from these laterals are classified as third, fourth, etc, order roots (Yamauchi *et al*., 1987a, b). Tiller roots do not appear on cereals until tillers have two or three leaves (Klepper *et a*l., 1984). Maximum length of taproot systems is longer than cereal root systems (Fageria *et al*., 1997). In grass- legumes mixture, small K and P availability might lead to the dislodgment of legume by grasses. Once grasses were growing with or without legumes, there was no substantiation that N concentrations increased in grass foliage, although P concentrations were frequently lower (Wei *et al*., 2022). Collectively, grasses develop root systems than legumes (Fageria *et al*., 2006). Root system of numerous crop plants were observed to be fractural (unequal and disjointed patterns) (Yang *et al*., 2022) with fractal dimensions of 1.48 to 1.58 for complete root systems (Tatsumi *et al*., 1989).

SHOOT- ROOT RATIOS

The shoot and root denote to aboveground and underground fractions of higher plants, respectively (Aung, 1974) and shoot-root ratio changes during vegetation stage concomitantly with changes of other important characters of plants (Bláha, 2019). Root growth is meticulously connected to whole-plant growth. Root dry weight is associated to entire dry weight of a plant using the following calculation or equation (Yoshida, 1981):

$$W_R=HW_T^h \qquad (1.1)$$

Where W_R stands for root dry weight, W_T refers to total dry weight (Shoot dry weight+ root dry weight), and H and h remain constants. Equation 1.1 can be changed into a logarithmic formula by way of:

$$\log W_R = \log H + h \log W_T \qquad (1.2)$$

Consequently, log W_R remains a linear function of log W_T. The association has been confirmed for diverse rice cultivars cultivated under several ecological environments and can be stated by the subsequent equation (Yoshida, 1981):

$$W_R= 0.212\ W_T 0.936 \qquad (1.3)$$

Once plants are minute (substitute 1 for W_T), W_R / W_T is ~ 0.2; W_R/ W_T values move toward 0.1 as plants grow bigger (Substitute 10^5 for W_T). Specifically, ratios of root dry weight to total dry weight range from ~ 0.2 at the seedling phase to ~ 0.1 at the propagative phase (heading) for rice (Yoshida, 1981). The association observed directly above offers an approximation of root mass that remains in soil if shoot weight is identified. Assuming for example, shoot dry weights of 3t. ha^{-1} manufactured by plants at heading; the root dry weight left-over in soil would be ~ 330kg ha^{-1}. Evans and Wardlaw (1976) stated that dry matter in roots comparative to shoots is great in the seedling periods of growth and gradually drops all over development. Fageria (1992) reported that shoot-root ratios of common bean, rice, wheat, and cowpea increases as plants progress in age. Increases in shoot- root ratio designated that shoot had a higher preference for photosynthate amassing than roots (Nakanishi, 2021). If shoot-root ratios decline with time, roots have special consumption of photosynthates under the prevailing plant growth situations. In a similar study, Eghball and Maranville (1993) examined that ecological stresses increase relative weight of roots in comparison to shoots.

INFLUENCE OF PLANT NUTRIENTS TO SHOOT – ROOT RATIOS

Nutrient acquisition and uptake are controlled to enhance the take up of nutrients required for growth and reproduction. Under nutrient-deficient conditions, the root: shoot ratio increases to augment soil exploration and nutrient uptake (Leigh, 2017).The root- shoot ratios of perennial ryegrass were observed to be enhanced by reductions in availability of N, P, or water (Davidson, 1969). Deficiency of many mineral elements was reported to effect plant growth and root- shoot associations, with water and N deficit being the greatest constraint limiting plant growth (Fageria *et al*., 2006). Mackay and Barber (1985) reported that at volumetric soil moisture contents of 0.22, 0.27, and $0.32m^3 .m^{-3}$, root – shoot ratios of 28 - day - aged maize plants were found to be 0.27, 0.15, and 0.18, respectively. Roots of wheat seedlings nurtured uninterruptedly for 18-days in comprehensive nutrient solutions, were found to contain less sugar content than related roots of wheat seedlings of the same age (18 days) grown under one week denial of nitrogen (Talouizte *et al*., 1984). Withdrawal of N, and transportation of photo assimilates from shoots towards roots was found to rise-up due to increased sink power of roots in contrast to sinks (Champigny and Talouizte, 1981). Eghball and Maranville (1993) reported that root –shoot ratios of maize plants were greater when reared with little soil N as opposed to sufficient N. Salt stress was observed to be very harsh on shoot growth than on root (Fageria *et al*., 2006). Shoot-root ratios of rice was examined to be largely constant due to functional equilibrium, thus signifying that shoot growth was proportionate to root growth (Slaton and Beyrouty, 1992).

INFLUENCE OF PARTITIONING OF PHOTOSYNTHATES ON ROOT-SHOOT RATIOS

Several ecological factors including low temperature, water deficit, and dearth of mineral nutrient were examined to have impact on dry matter delivery in plants. For example, Costa *et al*. (2002) observed that mineral nutrients like P and N caused appreciable influence on photosynthate and dry matter partitioning between shoots and roots. In addition, phosphorus- and N- deficient plants generally have additional dry matter apportioned to roots than shoots, possibly due to advanced export rates of photosynthates to root. It is comprehended that leaf enlargement is extremely sensitive to low P absorptions in tissue, resulting into greater amount of sucrose and starch in P- scarce leaves consequent of decreased request (Fredeen *et al*., 1989). Roots were observed to compete more for photosynthates than shoots, and subsequently promoting greater transport of carbohydrates to roots with compatibly lower shoot- root dry weights (Rufty *et al*., 1993). Little supplies of P, K and Mg were examined to have different impacts on dry matter partitioning between shoots and roots of common bean (Cakmak *et al*., 1994). Potassium- and particularly Mg- undersupplied plants had greater shoot than root dry matter, and P- scarce plants had reduced root dry matter than shoot dry matter (Cakmak *et al*., 1994). In an experiment, Fageria *et al*. (2006) observed that shoot – root dry weights ratios recorded 1.8 in P- deficient, 4.9 in control, 6.9 in K- deficient, and 10.2 in Mg- deficient plants.

MEASURING PLANT ROOT GROWTH PARAMETERS

The study of plant growth allows a numerical deduction on how sound plants can flourish in or adjust to particular environmental situations (Muhlich *et al*., 2008). Frequently measured root- growth parameters include number roots, tips, weight, surface area, volume, diameter, and length (Bohm, 1979) and root density (Dong *et al*., 2018). Techniques for measuring root growth parameters can be grouped into traditional or manual methods and modern or machine vision methods.

Traditional or Manual Methods of Measuring Root Growth Parameters

Root weight is most usually used parameter for studies of root growth comparative to environment. The method is that rinsed roots are normally dehydrated and their weight established. In the course of determining the dry weight, washed and cleaned roots are dried up in an oven at 105^0c for approximately 10 to 20 hours, subject to the number of roots (Bohm, 1979), or dehydrating the roots in an oven at an extensive time between 60 to 75**°C** and this has the benefit of stopping roots from being crushed (Fageria *et al*., 2006). Even though root weight is a suitable parameter for describing total mass of roots in soil, it is however not very good parameter for describing absorbing ability of roots in soil (Pradhan *et al*., 2012). According to Bohm (1979), good roots epitomize only an insignificant portion of entire root weight but are the greatest vigorous percentage of the root arrangement. Root length is employed to forecast root reaction to fluctuations in environment (Calleja-Cabrera *et al*., 2020). It is broadly used as a pointer for acceptable root morphology (Bauhus and Messier, 1999). It is an important factor for estimating the soil volume explored by a root system and therefore the amount of water and nutrients obtainable to the plant (Faye *et al*., 2019). Root length (R) can be willingly quantified through a technique defined by (Newman, 1966):

$$R = AN/2H \qquad (1.4)$$

where R= the entire length of roots; N= the number of interrupts between root pieces and the straightforward lines on a grid; A= the area of a four-sided or rectangle of the grid; and H= the overall length of the straight lines. Matthew *et al*. (1986) reported the use of line intersect method described by Newman (1966), and refined by Tennant (1975) to determine root lengths of a perennial ryegrass (*Lolium perenne* L.). Tennant (1975) put forward that the Newman method might be shortened. Newman (1966) proposed that for a grid of unspecified magnitudes, the contact totals may be transformed to centimetre dimensions by means of the equation:

Root length (R) = 11/14 x number of crossings (N) x grid unit. (1.5)

To find a length change factor, the 11/ 14 is joint with the grid unit. The factors for 1, 2, and 5 cm grid squares are 0.786, 1.57, and 3.93, correspondingly (Bohm, 1979).

8.1.3 Root density has been stated as root length for each unit soil volume. It is computed as:

Root density = (total root length in cm)/ (soil volume)

where roots have been gathered in cm^3) = $cm.cm^{-3}$. (1.6)

Mean root ratio (r_o) might be computed from the subsequent equation:

(Teo *et al*., 1995):

$$r_o = (FW/L)^{1/2}. \qquad (1.7)$$

where FW = fresh weight and L = root length in cm.

Traditional measurements of seedling roots often rely on manual measurements and existing root scanners on the market. Manual measurement requires a lot of labor and time, and subjective reasons may cause the uncertainty of data; root scanners have limited scanning size and expensive. In case of fruit roots, coverage and occlusion issues will occur, resulting in inaccurate results, but our research will solve this problem (Liu *et al*., 2019).

Modern or Machine Vision Methods of Measuring Root Growth Parameters
Machine vision technology is a comprehensive technology involving many disciplines such as artificial intelligence, intelligent control, neural network, neurobiology and graphic image processing. With the development of machine vision technology and research in the field of plant science, it has been widely used in the identification of plant species, plant growth information detection, quality inspection and classification of agricultural products as well as visual navigation of farmland (Tian *et al*., 2020).

The root length and hypocotyl length of Arabidopsis seedlings grown under different light conditions and influence of TOR kinase inhibitor AZD were measured with Fiji ImageJ software (Yan *et al*., 2022). Key parameters of root growth which include simple morphological parameters such as: root length (mm) and area ($mm^{2)}$, root number or root diameter and composite parameters such as specific root length (SRL: mm g − 1) or those related to root activity could be evaluated using WinRHIZO™ – WR – (Regent Instruments Inc., Quebec, Canada, 2000). WinRHIZO™ – WR – is one of the best-known software programmes to measure morphological parameters in root images (Pang et al. 2011; Yan *et al*., 2022).Root parameters such as number of root tips (RT), total root length (TRL) in mm, average diameter (AD) in mm, and root length (mm) in diameter class could be calculated with the two software programs namely software (WinRHIZO Pro 2013d, Regent Instruments, Quebec, Canada)(WR) and software RhizoVision Explorer (© Noble Research Institute, LLC)(RVE)(Stoffel *et al*., 2022).

Computer Code is a method that quickly quantifies vertical root distribution parameters using limited root length density data obtained from soil cores under field conditions. It was implemented through the development of a Minitab macro and an R script, with sample calculations conducted on field collected root length density data from the winter wheat crop in Texas, USA. The method is potentially useful for quantifying plant root activities in natural soils where deep roots are difficult to access by physical means and data of root length density distribution are only available in the upper root zone (Dong *et al*., 2018). Liu *et al*. (2019) used combined root characteristic parameters algorithm, a set of measurement system that employed the Labview software platform adjusted by statistical methods to accurately measure root morphological parameters such as root surface area, average root diameter, root length and root volume of apple seedlings.

The root morphology parameters-surface area, length and volume of apple root were measured by the designed system which provided important data for the researchers to get the growth status of apple seedling roots, as well as the physical and chemical properties of soil in order to improve the cultivation and management of apple seedlings. A semi-automatic workflow for the measurement of 3D root system development from time-series X-ray computed tomography volumes using backward prediction were developed. The workflow took into cognizance changes in root elongation rate with growth, which indicated that the workflow is a useful tool for root growth measurement, and that it can be applied to the study of RSA plasticity responses to varying soil environments (Teramoto and Uga, 2022).

CONCLUSION

Plant root system architecture shows numerous flexibilities in response to environmental variations, and how can plant roots be critical to evolving crops with more efficient roots (Khan *et al.*, 2016). Most field techniques employed in studying root growth are labour demanding, methods were advanced to detect root growth *in - situ*. A historical outline of the development of innumerable transparent wall methods was offered (McMichael and Taylor, 1987). Currently, selected research organizations have established huge rhizotron facilities to study plant root schemes. Further analogous methods have been established in contemporary years which incorporate minirhizotrons and related micro video camera procedures to offer chances for measuring in *-situ* root systems of field- raised plants (Murphy *et al.*, 1994).Understanding RSA and the machineries of its progress will permit management and manipulation of diverse root traits to advance plants' version to altering climates and intensify yields for the increasing worldwide human population (Smith and De Smet, 2012).

There is need for unique methods and techniques for investigating root systems of plants since plant roots are buried in the soil or substrate in which they are grown. Conventionaltechniques like coring, trenching, excavating, rhizotrons, pinboards and root washing have been used to access roots in the field, yet some of these methods are destructive and recurring observations or measurements cannot be achieved. Methods of measuring root growth in the field have been extended with new techniques established to calculate root growth of container-produced plants. Methods such as the Horhizotron™, mini-Horhizotron, and rhizometer, are valuable tools in determining root features under production conditions of fertilizers, watering or plant growth regulators. Developing computer software for use in measuring roots has enhanced the depth and extent of root characteristics calculated. By means of these non-destructive methods in actual period seems to be the way for future methods in Rhizometrics (Judd *et al.*, 2015).

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