

ABSTRACT

CAVINS, TODD JASON. Adaptation of the PourThru Nutrient Extraction Procedures to Greenhouse Crop Production. (Under the direction of B. E. Whipker and T.E. Bilderback.)

The purpose of this research was to adapt the PourThru nutrient extraction technique, which is the displacement of the bulk solution from the production container without a destructive harvest, to commercial greenhouse crop production. PourThru is a quick and easy nutrient sampling technique that is not laboratory oriented and can help prevent costly nutritional problems for greenhouse crop producers. Additionally, time domain reflectometry (TDR) was evaluated as an experimental tool to measure substrate moisture content, which may affect PourThru extraction. Time domain reflectometry is quick, non-destructive and has potential for use in automation of moisture content determination in greenhouse production. Previous PourThru research had focused on techniques for use on large nursery containers ($\approx 3800 \text{ cm}^3$) versus the smaller floriculture containers ($\approx 1500 \text{ cm}^3$) and no exact calibration of PourThru nutrient values to saturated media extract (SME) values, the current standard for nutrient testing, had been completed. Therefore, studies were implemented to examine the relationship of PourThru to SME, evaluate irrigation systems and timing effect on PourThru results as well as develop recommended influent and leachate volumes to ensure an unadulterated sample. Calibration curves were developed between PourThru and SME values and r^2

values ranged from 0.91 to 0.99 for linear relationships. Irrigation systems did affect electrical conductivity (EC) values and alternative interpretive standard values were developed dependent upon irrigation system. Timing of the PourThru was important to ensure adequate leachate was collected for sample analysis and the amount of influent affected EC values and the amount of leachate collected. The use of TDR was effective in small containers (980.6 and 2177.5 cm³); however, care should be taken to match probe size to container size to ensure representative sampling. The largest limitation to TDR use was the bulk density of the substrate being analyzed. Approximately a 50 to 75% underestimation occurred when substrate bulk densities were below 1.2 g·cm⁻³, but material specific calibration improved TDR accuracy to within 4%.

**ADAPTATION OF THE POURTHRU NUTRIENT EXTRACTION PROCEDURES
TO GREENHOUSE CROP PRODUCTION**

by

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DEDICATION

I dedicate this to my girls.

BIOGRAPHY

I am Todd Jason Cavins, born to T.J. and Judy Cavins on 22 Sept. 1974 in El Reno, Oklahoma. I have one older sister, Kristi J'mae Cavins. My paternal grandparents are Thomas J. and Juanita A. Cavins and maternal grandparents are Atmer O. and Thelma M. Taylor all of El Reno, Oklahoma. I graduated from El Reno High School in May 1993. On 12 June 1993 I married Ruth Christine Geppert, my high school sweetheart. We attended Southwestern Oklahoma State University, Weatherford, Oklahoma, where I received my B.S. in Biology. I continued my education at Oklahoma State University, Stillwater, Oklahoma, receiving my M.S. in Horticulture in December 1999. That same year our first daughter, Chloe Elaine was born on 14 April. Ruthie, Chloe, and I then moved to Cary, North Carolina and I pursued my Ph. D. at North Carolina State University, Raleigh, North Carolina. While in North Carolina, our second daughter, Wren Suzette, was born on 2 February 2002. We lived in Cary, North Carolina until the completion of my Ph. D. and then moved to Stillwater, Oklahoma where I obtained a position of Assistant Professor of Floriculture in the Department of Horticulture and Landscape Architecture at Oklahoma State University.

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Chapter 1.

INTRODUCTION AND LITERATURE REVIEW

Introduction

Proper plant nutrition is critical for producing healthy, profitable floriculture crops. Crop time, plant quality, and production cost are influenced by nutrition. In today's floriculture production systems rapid changes can occur with nutrient status. The use of soilless media allows the producer greater control over pH and nutrient management. However, the greater control requires intensive monitoring to maintain proper nutrition parameters. A substrate testing protocol that provides producers with a simple, quick, and quality sample is necessary to make on-site decision about nutrition and fertility management.

Routine substrate monitoring of floriculture nutrition and implementing corrective procedures, when warranted, can eliminate a majority of nutrition related problems. The simple monitoring and management of the substrate solution pH and electrical conductivity (EC) with occasional complete substrate and tissue nutritional analysis can thwart most plant nutrition problems. Informal surveys have indicated that less than 10% of floriculture producers perform routine pH and EC monitoring (D. Bailey, personal communication). The major citation for lack of monitoring was time constraints. Most stated that nutritional analysis, in-house pH and EC or

outsourced complete substrate and tissue analysis, was only completed when a visual enigma was present.

Tissue testing is the best way to truly assess what nutrients are being absorbed by the plant and is necessary from time to time. However, many producers hesitate to use this procedure as a nutritional monitoring tool due to the time (both collection and waiting for laboratory results), semi-destructive harvest, and cost. Over the years, many correlations, between substrate nutrient levels, plant growth, and/or tissue nutrient levels have been established (Ku and Hershey, 1991; Ku and Hershey, 1992; Lang and Pannkuk, 1998; Wright et al., 1990). In addition, a substrate nutrient test is indicative of plant available nutrients (Marschner, 1995). Therefore, it is practical for growers to use simple substrate testing methods to monitor plant nutrition.

Comparison of Substrate-Testing Methods

Many substrate-testing methods exist. Some methods are laboratory oriented while others offer a more in-house friendly protocol. Saturated media extract (SME) is a laboratory-oriented substrate testing method that is commonly used among commercial and university substrate laboratories (Dole and Wilkins, 1998; Lang, 1996). Nutritional values and recommendations on plant fact sheets and extension publications are generally based on SME values (Lang, 1996).

Saturated media extract is a time-consuming plant destructive method. The SME yields operator dependent results due to protocol factors such as water content and location of substrate removal from the container. Saturated media extract is

also a laboratory-oriented technique that is unpractical for floriculture producers to use for in-house nutritional analysis. The SME (adapted from Warncke, 1998) is performed as follows:

❶ Collect root substrate. Remove the plant from the container and remove approximately 250 cm³ of substrate from the rootball. If necessary, remove slow release fertilizer capsules (ruptured capsules will release fertilizer and provide non-representative EC or nutrient values). Place the substrate into a container.

❷ Add water. Mix in a volume of distilled water that is just enough to cause the substrate and water mixture to “glisten”.

❸ Allow the mixture to equilibrate. The amount of time varies depending on laboratory; usually 30 minutes to an hour is normal protocol.

❹ Extract the substrate solution. Three options are available, the first is a vacuum system, the second is gravity filtration, and the third is squeezing the substrate-water mixture through cheesecloth. *Note: Some labs test the pH of the substrate-water mixture and some test the water after it has been removed by vacuum. Testing after vacuum extraction can result in pH values approximately 0.5 units lower than measures taken in the substrate-water mixture previous to vacuuming or pH values obtained by gravity filtration or cheese cloth extraction. The vacuum increases CO₂ content of substrate solution, which causes H₂CO₃ (carbonic acid) to form; thus, decreasing the solution pH.*

Volumetric slurry dilutions are simple substrate tests used by floriculture producers to measure pH and EC. However, the high degree of dilution makes the extraction unsuitable for complete nutrient analysis. The volumetric slurry (Adapted from Dole and Wilkins, 1998) dilutions are performed as follows:

- ❶ **Collect root substrate.** Remove the plant from the container and remove approximately 250 cm³ of substrate from the rootball. Place substrate into a clean container. If controlled release fertilizer is present, remove the capsules and ensure no capsules are damaged (damaged capsules will release fertilizer and provide non-representative EC or nutrient values).
- ❷ **Add water.** Add a volume of distilled water equal to 2 times the volume of the root substrate (500 mL water).
- ❸ **Mix.** Thoroughly mix substrate and water. Allow the mixture to set for a minimum of **20** minutes up to an hour for equilibration.
- ❹ **Test the samples for pH and EC.** Test the slurry 20 minutes to 1 hour after mixing. Take care not to damage the pH or EC probes on substrate components when inserting into slurry.

PourThru is a bulk solution displacement method that is increasing in popularity (Cavins et al, 2000a,b; Cavins et al., 2001; Whipker et al. 2000). This simple substrate solution extract can be easily performed in the greenhouse and/or sent to commercial or university laboratories for complete nutritional analysis. PourThru substrate solution extraction can be performed in the growing container without special equipment or the need to transport substrate samples to a

laboratory. The ability of horticultural producers, consultants, and researchers to save valuable time for nutrient analysis can vastly influence the profitability of a crop. On-site analysis allows fertility regime adjustments in a timely manner. The PourThru (Adapted from Wright, 1986) is performed as follows:

❶ Irrigate the crop one hour before testing. Make sure the substrate is satiated.

If the water supplied by the automatic irrigation system varies, then water the pots/flats by hand. If using constant liquid feed, irrigate with fertilizer solution as usual. If using periodic feeding: (A) irrigate with clear water and test a day or two before fertilizing and (B) test on the same day in the fertilizing cycle each time.

❷ Place saucer under container. After the container has drained for 30 to 60 minutes, place saucers under the containers to be sampled. If you are testing seedlings in bedding plant flats, place one cell pack in each saucer.

❸ Pour enough distilled water on the surface of the substrate to get 50 mL of leachate in the saucer (10 cm and smaller containers only require approximately 30 ml). The amount of water needed will vary with container size, crop, irrigation system, and environmental conditions.

❹ Collect and test the leachate for pH and EC. Allow the containers to drain for 5 minutes to obtain 50 mL of leachate. Leachate volumes over 70 mL (based on 15.2 cm pot extractions) will begin to dilute the sample and give non-representative EC values.

Bulk Solution Displacement

As mentioned above, PourThru is a bulk solution displacement method. Bulk solution is analogous to soil solution, which is a quasi-equilibrium solution that contains a particular concentration of electrolytes under saturated conditions (Faber and Nelson, 1984; Pearson, 1971). The bulk solution contains plant available water and nutrients. Therefore, unadulterated bulk solution is the ideal testing media for plant substrate nutritional analysis (Burd and Martin, 1923). Researchers have been investigating procedures to extract an unadulterated soil or bulk solutions for over 130 years (Schloesing, 1866).

Many methods and displacement solutions have been studied to determine optimum bulk solution displacement. However, most methods are not appropriate for greenhouse use. An optimum bulk solution displacement method should have practical applications that maintain the integrity of the plant and yield consistent and reliable nutrient analysis. An in-house greenhouse practical application of bulk solution displacement immediately puts the information of nutritional analysis in producers' hands.

Early studies on soil solution displacement involved oil as the displacement solution (Morgan, 1916, 1917; van Suchtelen, 1912). The oil was used to prevent mixing of the displacement solution with the leachate. However, complications were noted with oil surrounding the soil solution and inconsistent solution extractions were obtained. The method was unclean and used complicated tension applying machinery to remove the soil solution (Parker, 1921). In addition, the use of oils

would yield deleterious effects on plant growth if used in a production setting. Therefore, oil displacement is not a practical option.

Alcohol displacement solutions have also been evaluated. Ischerekov (1907) used ethyl alcohol to displace the soil solution and reported that an unadulterated soil solution was obtained. However, Faber and Nelson (1984) reported bulk solution displacement using methyl alcohol yielded lower concentrations of Ca, K, and P compared to using water as a displacement solution. Differences were attributed to the lower dielectric constant (which affects electrolyte solubility) of alcohol, which reduced ion solubility compared to water. Thus, reduced concentrations of the nutrients were displaced.

Parker (1921) noted that alcohol mixes with the soil solution; therefore, a true displacement may not occur. However, Parker also reported that the displacement solution forms a zone of soil solution saturation just below the displacement solution; therefore, no mixing should occur. Other observations by Parker include that methanol has no advantage over water and is inferior to ethanol as a displacement solution. The use of alcohols as displacement solutions would have deleterious effects on plant growth if used in a greenhouse setting. Therefore, alcohol displacement is not a practical option.

Centrifugation is another soil/bulk solution displacement method (Faber and Nelson, 1984; Merek and Carle, 1974, Mubarak and Olsen, 1976). Although solution extraction is achieved, the centrifugation system is not a practical, quick method for nutrient analysis. Substrate must be removed from the container, which may be destructive to the plant. A benefit of centrifugation is that the non-miscible

solutions can be added to soil samples with low moisture content and still achieve displacement of the soil solution (Merek and Carle, 1972). In contrast, Faber and Nelson (1984) noted that samples having an initially low moisture content did not yield sufficient amounts of solution for complete analysis.

An equally or more – effective bulk solution displacement method is column displacement (Faber and Nelson, 1984). This practice has been adapted to a greenhouse method known as PourThru (Cavins et al., 2000a; Cavins et al., 2000b; Wright, 1986; Wright et al., 1990; Yeager et al., 1983). Column displacement and PourThru possess limitations with soil solutions due to small particle size and porosity as well as strong adsorptive forces (Spomer, 1979). Soil solution displacement may take several days to complete depending upon soil moisture content, degree of packing in column, soil type, and column design (Parker, 1921).

However, PourThru facilitates rapid on-site results and is an ideal nutrient analysis tool for most floriculture substrates. The components of floriculture substrates have been chosen due to the properties associated with overcoming small container plant production anomalies. These same substrate properties, high porosity, well draining, low bulk density, make the PourThru solution extraction method ideal for bulk solution displacement in production containers.

A Limitation of PourThru Monitoring

A major limitation in the use of PourThru monitoring is the lack of recommended floriculture nutrition values based on PourThru extractions. Yeager et al. (1983) compared PourThru and SME values, but results were based on 100% pinebark substrate. Most commercial floriculture substrates are peatmoss based which may significantly alter the values. Albeit, Wright (1986) observed no differences between peat:perlite (50:50 v/v) and 100% pinebark when compared without plants in the substrates. Plants growing in the substrate would affect nutrient concentration in the bulk solution (Compton and Nelson, 1997).

Yeager et al., (1983) and Wright (1986) did not state the amount of time between irrigation and the PourThru extraction; although, containers were allowed to drain to container capacity. Compton and Nelson (1997) noted that time after irrigation affects nutrient content of the substrate solution. One or two hours is recommended after an irrigation to allow for plant nutrient uptake equilibration.

Wright et al. (1990) used a peat-based substrate to compare PourThru and SME values for poinsettia. However, both of the methods' values were correlated to tissue levels and not to each other. Wright et al. (1990) noted that PourThru EC values were nearly two times higher than SME EC values. Pilot studies conducted by the author have indicated that PourThru EC values are approximately 1.4 times higher than SME EC values. This discrepancy may be a result of variations in the PourThru and SME protocols used in each study.

Wright (1990) waited two hours between irrigating and completion of the PourThru extraction. Two hours is sufficient time for the water content to change drastically from container capacity due to plant uptake or evaporation; thus, affecting nutrient concentration in the bulk solution (Reitemeier, 1945). Wright (1990) also noted that 100 mL of displacement solution was used to obtain the leachate from a 15 cm container, but did not report the volume of leachate collected. Another variable is the number of samples analyzed. The data presented by Wright (1990) is only from two sampling dates with limited plants (10) per treatment. A larger sampling quantity is needed to help strengthen recommendations.

Handreck (1994) used various substrates to compare PourThru pH values to SME pH values obtained in various regions of the pot. Handreck (1994) noted that PourThru pH values were approximately 0.53 units higher than SME pH values obtained using the entire rootball. PourThru pH values most closely resemble SME pH values obtained from the lower portion of the rootball. Hipp et al. (1979) noted that leachate and SME pH values correlated well ($r^2 = 0.95$).

Factors affecting PourThru Values

Several major factors may affect PourThru pH, EC, and nutrient values. The type of irrigation system can affect water content and solute movement; thus, affecting EC and nutrient values (Argo and Biernbaum, 1995; Blom and Piott, 1992; Guttormsen, 1969; Morvant et al., 1997). The amount of displacement fluid may mix with the leachate, altering pH values and diluting EC and nutrient concentrations. And, the water content of the substrate and length of time following irrigation that the

PourThru extraction is performed may alter the nutrient retention and affect the amount of leachate produced for a given volume of displacement solution (Reitemeir, 1945; Yeager et al. 1983).

Overhead-hand irrigation is the traditional method of watering floriculture crops. Overhead-hand irrigation causes a head of water, which forms on the top of the substrate from the relative high rate of water applied, to move quasi-uniformly down through a container, but can increase substrate compaction due to the mass of the water collapsing the substrate components (Fonteno et al, 1981). The water head movement and compactive properties promote uniform substrate wetting within a pot by reducing the influence of channeling and capillary water movement. In addition, the water head movement also moves solutes via mass flow to the bottom portion of the container. Morvant et al. (1997) noted that EC was higher in the middle and bottom region of the pot when plants were irrigated by overhead-hand versus sub-irrigation (ebb and flow) or drip-tube irrigation. With a higher concentration of soluble salts in the lower portion of the pot, EC values may be higher for PourThru extractions performed on overhead-hand watered containers versus drip-tube or subirrigated containers due to the nature of displacement.

Drip-tube irrigation is a reduced-labor irrigation method versus overhead-hand irrigation. Similar to hand irrigation, salts can move to the bottom of the container. However, a significant amount of capillary movement of water and solutes occurs due to a lack of dominant bulk-head water movement. This promotes movement of water and solutes towards the middle and upper 1/3 of the container as noted by Morvant et al. (1997) that observed higher EC values in the top region of the pot for

drip-tube versus overhead-hand irrigation. Therefore, PourThru EC values obtained from containers that were drip irrigated may lead to lower EC values than overhead-hand irrigated containers.

Subirrigation systems allow automation of large areas and are becoming more popular among large greenhouse-floriculture operations. Benefits of subirrigation are reduced foliar-disease spread, reduce labor, and prevention of nutrient run-off (Morvant et al., 1997). Water movement within a container that is subirrigated is capillary. Therefore, solutes may accumulate in the top 1/3 of the substrate because there is no bulk water head movement to transport the solutes to the lower portion of the container.

Wright (1986) noted that containers must be near “water holding capacity” when the PourThru extraction is performed. Water holding capacity is the amount of water held by the substrate after the substrate has been saturated and allowed to drain (Fonteno, 1996). Water holding capacity would most likely be achieved immediately after overhead-hand irrigation, but likely would never be achieved using drip-tube or subirrigation.

Excessive displacement solution application causes mixing with the leachate during PourThru extractions (P. Nelson, personal communication). Excessive mixing could alter pH, EC, and nutrient values. The proper amount of displacement solution to use may be affected by pot size and plant development as increasing root volume may affect displacement and mixing.

Water content of the substrate at the time of PourThru did not affect PourThru values when large nursery containers (3 L) with a sand bark mix were investigated

(Yeager et al. 1983). However, typical floriculture production uses smaller containers than those investigated by Yeager et al. (1983). Therefore, water content may be important due to the small volume of substrate and the substrate components (i.e. peatmoss and vermiculite) used in floriculture production systems. Reitemeier (1945) noted that water content affects nutrient availability.

Wright (1986) noted that containers must be near “water holding capacity” when the PourThru extraction is performed. However, if PourThru extractions are performed immediately after irrigation non-representative values may be attained. Compton and Nelson (1997) suggested that substrate testing be performed one to two hours after fertilization to allow for nutrient equilibration in order to obtain values that represent plant available nutrients. Time following fertilization and/or irrigation must be examined for PourThru extractions.

The PourThru substrate solution extraction offers a simple and timely assessment of plant nutrition. However, to become a complete nutrient analysis tool, several issues must be addressed. Major issues include: What is the effect of irrigation systems on PourThru values?; How much displacing fluid is necessary for a given pot size so that an unadulterated substrate solution is obtained?; How does time after fertigation and water content affect PourThru values?; and What is the definitive relationship between PourThru and SME values?

Time Domain Reflectometry

A novel system to measure soil water content was introduced by Topp et al. (1980). The time domain reflectometer (TDR) measures the apparent dielectric constant of a substrate via the propagation and reflection of an electromagnetic wave. Although TDR has proven effective to measure water content of soils, its effectiveness on floriculture potting mixes has not been investigated. Initially, the hope was to use TDR to measure soil water content in the PourThru experiments to describe the affects of substrate moisture content upon PourThru nutrient extraction values. However, several issues concerning TDR were necessary to resolve.

Time domain reflectometry works by using metal rods (probes) as wave-guides for the transmission of the TDR signal (electromagnetic wave). The TDR generates a high-frequency-pulse that propagates along the wave-guides generating an electromagnetic field around the TDR-probe. At the end of the wave-guides, the pulse is reflected back to its source. The resulting wave velocity or transit times and dielectric constant are dependent on the water content and soluble salt content of the material (Topp et al., 1980).

Time domain reflectometry offers many advantages over conventional moisture analysis in horticultural substrates. Conventional analysis for gravimetric moisture content required a destructive harvest of the substrate and extensive drying times so that dry weights of the substrate could be compared to wet weights. The instantaneous, non-destructive results associated with TDR moisture analysis are advantageous. Other advantages include portability and data logger capabilities.

Several researchers have noted that soils with high organic contents skew TDR accuracy (Pepin et al., 1992; Anisko et al., 1994). Most modern floriculture substrate mixes are primarily peatmoss (organic matter) with various portions of rockwool, coir, bark, sand, vermiculite, perlite, or other components. In addition to the high organic matter content, the addition of materials such as rockwool, coir, and perlite, which have insulating properties, may affect TDR values.

The use of TDR to measure the dielectric constants of various chemicals was introduced in 1969 by Fellner-Feldegg. The dielectric constant is a measure of a material's ability to resist the formation of an electric field within the material. In simple terms, it is a value assigned to rate the electrical conductance potential. Water has a very high dielectric constant ($\epsilon_r=80$) while mineral soils or other porous materials have low dielectric constants ($\epsilon_r \cong 4$). A vacuum has a dielectric constant equal to one.

Time domain reflectometry has been used for determining cable integrity and circuit faults as well as mineral soil moisture and electrical conductivity (EC) analyses. However, little information exists on the potential application to horticultural substrate analysis, especially applications to floriculture production, which typically involves small containers and small substrate volumes.

Soil scientists have used the TDR technology for many years to measure moisture content of field soils (Topp et al., 1980; Topp et al., 1984; Herkelrath et al., 1991). Recent advances in TDR technology have yielded small, portable, and

inexpensive units. These units show promise as becoming standard analytical tools for horticultural substrate analyses.

Previous studies on TDR in horticultural substrates have primarily dealt with larger containers than traditionally used in floriculture production (Murray et al., 2001; Anisko et al., 1994; da Silva et al., 1998). In addition, these studies also used TDR probes 15 cm or longer. Studies must be conducted to determine if TDR is applicable using shorter probes and smaller containers used in floriculture production.

The previous studies with TDR/horticulture substrates have been correlated to gravimetric water content, but have yet provided an exact match of volumetric water content to gravimetric content. Anisko et al. (1994) noted that the dielectric constant and water content relationship (θ) is not purely linear. Therefore, an exact match should not be expected. No studies have provided the actual volume of media at the completion of moisture additions. This is very important to floriculture application as shrinking/swelling of peatmoss can change the media volumes.

In addition, conventional TDRs have only provided a wave velocity from an oscilloscope that then had to be mathematically converted to volumetric water content. These conversion do not provide an exact relationship to volumetric water content, but have proven to be within reasonable limits. Pepin et al. (1992) and Anisko et al. (1994) have noted that organic substrates, such as peat, should be measured using an alternative calibration equation compared to those used for mineral soils (Topp et al., 1980).

Objectives

The objectives of this research were:

- A. To determine the effects of over-head-hand-, drip tube-, and sub-irrigation on PourThru pH and EC values.
- B. To determine the relationship between SME and PourThru extract on NO_3 , NH_4 , P, K, Ca, and Mg values in the leachate and develop interpretive PourThru standard values based upon the relationship.
- C. To determine the effects of water content and time of PourThru extraction on pH and EC values and optimize PourThru protocol to ensure a representative sample.
- D. To evaluate the practical use of TDR in small containers by varying probe and container sizes as well as evaluate the accuracy over moisture contents and material specific calibration.

Investigation and analysis of the above problems and objectives will ensure that the PourThru is a viable tool for complete nutrient analysis. Not only will PourThru enable producers to facilitate routine nutritional monitoring, but complete nutritional analysis can be performed with confidence. The evaluation of TDR will help determine if TDR analysis of water content is practical in small containers with floriculture type substrates.

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Chapter 2.

Establishment of Calibration Curves for Comparison of PourThru and Saturated Media Extract Nutrient Values

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Abstract. Most commercial and university substrate testing laboratories' recommended floriculture nutritional values are based on the saturated media extract (SME) method. With the recent gain in popularity of PourThru nutritional monitoring, alternative recommended values are needed for nutrient analyses based upon PourThru extracts. Therefore, PourThru nutritional values were compared to the SME values to develop calibration curves and recommended nutritional values. *Euphorbia pulcherrima* 'Freedom Red' Willd. ex Klotzch. were grown for two consecutive growing seasons in 16.5 cm plastic pots with Fafard 4-P root substrate and fertigated with 200, 300, or 400 mgL⁻¹ N from a 13N-0.88P-10.8K fertilizer. PourThru and SME extractions were performed at 6 and 9 weeks (0 representing potting of rooted cuttings) and analyzed for pH (prior and post SME vacuum extraction), electrical conductivity, divalent cations [calcium (Ca²⁺) and magnesium (Mg²⁺)], monovalent cations [potassium (K⁺), ammonium (NH₄⁺)], and monovalent anions [nitrate (NO₃⁻), phosphorous (H₂PO₄⁻)]. Linear relationships among the PourThru and SME extracts existed and calibration curves were developed (r^2

values ranged from 0.91 to 0.99). The established calibration curves will allow substrate-testing laboratories to make nutritional recommendations based on PourThru extractions.

Introduction

PourThru was introduced as a quick and simple method to analyze pot-plant pH and electrical conductivity (EC) on-site (Wright, 1986; Wright et al., 1990; Yeager et al., 1983). PourThru extraction occurs by the displacement of the bulk solution by distilled water poured over the top of a substrate as a means to obtain a nutrient sample. This method has been widely accepted by researchers and producers of woody ornamentals for approximately 20 years (Ruter, 1992; Wright, 1986; Yeager and Wright, 1981; Yeager and Wright, 1982; and Yeager et al., 1983). However, the greenhouse industry has been reluctant to accept this method until recent promotion at grower conferences and scientific meetings (D. Bailey, personal communication).

A possible reason for lack of acceptance may be that producers are already using a simple 1:2 (v:v, substrate:water) dilution slurry for on-site pH and EC testing. However, the 1:2 slurry generally does not provide a sample that is suitable for complete nutritional analysis due to the diluted nature of the substrate sample and inconvenience of shipping a slurry to a commercial or university laboratory. A PourThru extract has the advantage that it can be shipped easily to a commercial or university laboratory in a sealed container and since the substrate solution is unadulterated, it can be a preferred testing medium for plant available nutrients (Burd and Martin, 1923). The ability to use a single testing method for on-site pH

and EC analysis and to provide a sample to commercial or university laboratories would be advantageous to producers. Advantages would include the nondestructive nature of the test, the simple sample collection, and a single set of nutritional standard values to monitor crop performance.

A limitation for PourThru acceptance has also been the lack of standard nutrient values to interpret proper nutrient concentrations in the substrate solution. Previous researchers have noted that PourThru or leachate samples and saturated media extract (SME) values, the current substrate analysis standard procedure used by many commercial or university laboratories (Dole and Wilkins, 1999; Lang, 1996), provide comparable results (Cabrera, 1998; Jarrell et al., 1979; Handreck, 1994; Hipp et al., 1979; Wright, 1986; Wright et al., 1990; Yeager et al., 1983). However, the researchers did not provide calibration curves that would allow the establishment of recommended PourThru values based on established SME values (Cabrera, 1998; Jarrell et al., 1979; Hipp et al., 1979; Wright, 1986; Wright et al., 1990; Yeager et al., 1983) or the methods used to obtain the sample were not representative of the procedures used by many greenhouse producers (Cavins et al., 2000; Jarrell et al., 1979; Handreck, 1994; Hipp et al., 1979).

Yeager et al. (1983) provided PourThru and SME extraction correlation coefficients; however, the values were based upon comparisons of nitrogen, phosphorous, potassium, or dolomitic limestone applications. The researchers did not provide calibration equations to compare the two testing methods directly. The data presented by Wright et al. (1990) represented correlation coefficients of PourThru or SME with tissue nutrient levels. While the researchers validated

PourThru as an interpretable substrate test, they did not provide calibration equations to compare the two testing methods directly. A direct comparison of PourThru to SME is needed so that calibration curves may be established and commercial or university laboratories can make recommendations based on PourThru extracts from established SME standard values.

Yeager et al. (1983) data were based on 100% pinebark substrate without plants and the values were obtained at “container capacity”. Substrates used in floriculture production are peatmoss or peatmoss and pinebark-based (K. Santner, personal communication). Although Wright (1986) reported no difference in nutrient extraction between a peat:perlite (1:1, v:v) and pine bark (100%) substrate, nutrient retention may be differentially altered for each substrate by plant uptake. Lang and Elliot (1991) noted that ammonium oxidation varied among soilless substrate types and that plants in the substrates affected oxidation.

Compton and Nelson (1997) noted that plants could drastically alter nutrient values in small substrate volumes and recommended testing one to two hours after irrigation to obtain values representative of plant available nutrients. One to two hours is needed for equilibration of the substrate solution and containers with actively transpiring plants may be dryer than container capacity in that allotted time. Standardized PourThru values need to be based on conditions similar to those used by commercial floriculture producers that represent plant available nutrients.

Wright et al.'s (1990) comparison of PourThru and SME nutrient values was based upon PourThru extractions 2 hours after irrigation. The substrate samples for SME analyses were collected at the same time. Both the PourThru extract and

substrate samples were then shipped to a commercial lab. The SME nutrient values may be ambiguous due to nitrification that was likely occurring during shipment versus little nitrification occurring in the PourThru extract (Wright et al., 1990). While shipping the samples may be representative of producer practices, the comparison of PourThru and SME nutrient values should also be examined for samples that are handled in similar environmental conditions, with testing or sample preservation occurring in a timely manner.

Wright et al. (1990) and Yeager et al. (1983) only examined SME pH values obtained from the suctioned extract. Warncke (1998) suggests that the pH samples should be obtained directly in the slurry. Values for pH obtained in the slurry may differ from pH values obtained in the vacuumed extract. The vacuum extraction can change the CO₂ content of the solution, which affects the pH (Lang, 1996; Van Leirop, 1990).

The objective of this study was to develop calibration curves of PourThru and SME nutrient values. The calibration curves attained will allow producers to use a single substrate solution extraction to achieve both on-site and complete nutritional analysis by a commercial or university laboratory. The calibration curves will also allow laboratories to develop standard PourThru nutrient values based upon established SME standard values.

Materials and Methods

Euphorbia pulcherrima 'Freedom Red' Willd. ex Klotzch. rooted cuttings were transplanted on 15 August into 16.5 cm plastic pots with Fafard 4P commercial root substrate (Conrad Fafard Inc., Anderson, S.C.) and fertigated with 200, 300, or 400 mg·L⁻¹ N from a 13N-0.88P-10.8K Cal – Mag fertilizer (The Scotts Co., Marysville, Ohio). Polyethylene covered greenhouses were set at 21/16°C, day/night and the plants were grown under natural days. Substrate samples were collected at 6 and 9 weeks after transplanting rooted cuttings. PourThru extractions were made 1 hour after irrigation using 75 mL of distilled water to displace approximately 50 mL of leachate (Cavins et al., 2000; modified, Wright, 1986). PourThru samples were tested for pH and EC immediately following extraction. Substrate samples were selected at the same time for SME (modified, Warncke, 1998). Approximately 350 cm³ of substrate from the middle of the container was saturated with distilled water until “glistening”, then allowed to equilibrate for 30 minutes. pH was measured in the slurry. After pH values were obtained, the samples were vacuum filtered through a Buchner funnel using Whatman 40 filter paper and pH and EC were measured in the vacuumed extract. All extractions were preserved by refrigeration (5°C) and the addition of 0.05 mL phenolmercuric acetate saturated solution until the completion of the study. Samples were then analyzed for NO₃⁻, NH₄⁺, and P photometrically (Cataldo et al, 1975; Chaney and Marbach, 1962; Murphy and Riley, 1962) and for K⁺, Ca²⁺, and Mg²⁺ using atomic absorption spectrophotometry (Christian and Feldman, 1970).

The study was a split plot design with a 2 x 3 x 2 x 2-factorial treatment combination. Two years served as the main plots, three nutrient levels, two extraction times, and two extraction methods were the subplots. There were five replications per treatment. Analysis of variance was obtained using procedure general linear models ($\alpha = 0.01$) and the nutritional values were regressed and calibration equations developed using PROC REG (SAS Institute, Cary, N.C.).

Results and Discussion

The substrate pH did not differ between years or extraction procedures (data not presented). The lack of differences between years indicates that PourThru and SME are reliable, consistent methods for obtaining substrate pH. Surprisingly, no differences occurred among extraction procedures (PourThru, SME slurry, and SME vacuum extract) for pH. Yeager et al. (1983) noted that pH values for PourThru and SME were similar.

However, Lang (1996) noted that differences (0.1 - 0.5 pH units) are commonly obtained between SME pH values when they are taken from a slurry or vacuum extracted sample. These differences are attributed to partial pressure concentrations of CO₂ in the extract versus the atmosphere (Van Leirop, 1990). Evidently, our samples were allowed adequate time to equilibrate with atmospheric carbon dioxide (CO₂) after vacuum extraction to allow the pH values to return to the initial pH values prior to vacuum extraction.

As the fertilizer rate increased (200, 300, or 400 mg·L⁻¹), the pH decreased (6.12, 5.95, and 5.69, respectively). This pH decrease likely occurred due to the

increased ammonium (NH_4) content of the fertigation solution; indicating that the uptake of NH_4 from the fertilizer, which acidifies the root environment (Marschner et al., 1991), was high enough to counteract the basicity effects of the high nitrate fertilizer formulation (155 kg of calcium carbonate [CaCO_3] equivalent) (The Scotts Company, personal communication).

Sampling date affected pH such that the substrate pH increased from week 6 to week 9 (5.80 to 6.17, respectively), presumably due to build up of basic residues, such as hydroxide ions (OH^-) from NO_3 uptake (Raven, 1986) and carbonates (HCO_3^- and CO_3^{2-}). As noted above, the fertilizer formulation generally produces basic residues, which increases the substrate pH.

Electrical conductivity (EC) was not affected by year (data not presented). The lack of differences between years indicates that PourThru and SME are reliable, consistent methods for obtaining substrate EC.

The substrate EC was affected by extraction method. PourThru EC values (3.54 mS/cm) were higher than the SME EC values (2.56 mS/cm). PourThru EC values are generally higher due to less “dilution” compared to SME EC values (Yeager et al., 1983; Wright et al., 1990). The EC values between PourThru and SME extractions were compared and a calibration curve was developed to predict values (Fig. 1A). Table 1 lists an inverse of the calibration equation so that SME values can be predicted based upon PourThru values. This inversion is necessary because laboratories will likely obtain PourThru nutrient values and want to predict the SME value. The predicted SME values can then be compared to established standard values.

Electrical conductivity (EC) was affected by fertilizer rate. Increasing fertility rates (200, 300, or 400 mg·L⁻¹) increased EC values (2.09, 2.99, and 4.07 mS/cm, respectively). Similar trends have been noted by Yelanich and Biernbaum (1993) for SME and Wright et al. (1990) for PourThru.

The EC was not affected by sampling date (data not presented). During weeks 6 and 9 of poinsettia production, plants are in a state of rapid vegetative growth (Whipker and Hammer, 1997). The lack of differences between sampling dates may be due to the plants actively consuming a majority of the supplied nutrients.

Nitrate extraction values were not affected by year (data not presented). Similar to EC, NO₃ values were affected by extraction method. As with EC, PourThru NO₃ values (478.8 mg·L⁻¹) were higher than SME NO₃ values (313.2 mg·L⁻¹). The NO₃ values of PourThru and SME extractions were compared and a calibration curve and inverse calibration equation were developed to predict values (Fig. 1B and Table 1, respectively).

Substrate NO₃ values increased with increasing fertilizer rates. As the fertilizer rate increased (200, 300, or 400 mg·L⁻¹ N), so did the NO₃ content of the substrate (253.0, 387.1, and 547.9 mg·L⁻¹, respectively). The NO₃ and EC components behaved similarly as EC was measuring the total concentration of dissolved salts and NO₃ was being supplied in the largest quantity. As with EC, NO₃ substrate content was not affected by sampling date, which was attributed to the poinsettias being in a state of rapid vegetative growth and consuming the same

amount of NO_3 (relatively) during the three-week interval (Whipker and Hammer, 1997).

Ammonium (NH_4) values were not affected by the variables in this study (data not presented). The fertilizer formulation used contained only 1.1% ammoniacal or urea derived nitrogen (N). The plant likely readily absorbed the small amount of NH_4 provided. Therefore, only minute amounts of NH_4 were detected.

A year by week interaction affected phosphorus (P) substrate values (Table 2). Phosphorus values in year one were much lower than in year two. Phosphorus also decreased from week 6 to 9 in both years. Warncke and Krauskopf (1983) noted that 6 to 9 $\text{mg}\cdot\text{L}^{-1}$ was sufficient P for proper plant growth and development. In our study, plants were supplied 13.5 to 27.0 $\text{mg}\cdot\text{L}^{-1}$ P at each irrigation. The decrease from week 6 to 9 is likely due to increased P uptake from the substrate to support the larger plant mass and growth requirements (Whipker and Hammer, 1997).

The variation in P values between years was attributed to the starter charge variation in the commercial mix. Analysis conducted on the mix prior to planting indicated the P values in year one were 16.8 $\text{mg}\cdot\text{L}^{-1}$ (SME) versus 54.3 $\text{mg}\cdot\text{L}^{-1}$ (SME) in year two. This initial starter charge continued to affect P substrate values throughout the crop cycle. Argo and Biernbaum (1995) noted that containers irrigated only with tap water maintained near optimal levels of nutrients 42 d after initial irrigation.

A year by fertilizer rate interaction affected P substrate values (Table 3). As noted above, the P content in the starter charge varied between years such that year

one P substrate values were lower than year two. Increasing fertilizer rates increase P substrate values; albeit, year one had much lower P substrate values than year two.

Similar to EC and NO_3 values, P values were affected by extraction method. PourThru P values ($118.4 \text{ mg}\cdot\text{L}^{-1}$) were higher than SME P values ($62.6 \text{ mg}\cdot\text{L}^{-1}$). The P values of PourThru and SME were compared and calibration curves and inverse calibration equations were developed to predict values (Fig. 1C, D and Table 1, respectively).

Potassium extraction values were similar between years (data not presented). However, extraction method affected K values. Like EC, NO_3 , and P, PourThru K values ($336.8 \text{ mg}\cdot\text{L}^{-1}$) were higher than SME K values ($232.2 \text{ mg}\cdot\text{L}^{-1}$). A calibration curve and an inverse calibration equation were developed to predict values (Fig. 1E and Table 1).

Potassium concentrations increased with increasing fertilizer rates. Fertilizer rates of 200, 300, or $400 \text{ mg}\cdot\text{L}^{-1}$ N produced substrate K values of 178.5, 280.4, and $394.6 \text{ mg}\cdot\text{L}^{-1}$, respectively. Similar trends have been noted by Wright et al. (1990) for PourThru extractions.

As with EC and NO_3 values, K substrate values were not affected by sampling date (data not presented). This is attributed to the poinsettias being in a state of rapid vegetative growth and consuming the same amount of K (relatively) during the three-week interval (Whipker and Hammer, 1997).

Calcium substrate values varied between year one ($348.1 \text{ mg}\cdot\text{L}^{-1}$) and year two ($140.6 \text{ mg}\cdot\text{L}^{-1}$). Similar to P, the initial starter charge provided in the commercial

mix caused the variation. Analysis conducted on the substrate prior to planting indicated the Ca values in year one were $277.0 \text{ mg}\cdot\text{L}^{-1}$ (SME) versus $42.6 \text{ mg}\cdot\text{L}^{-1}$ (SME) in year two. This initial starter charge continued to affect Ca concentrations throughout the crop cycle. Argo and Biernbaum (1995) noted that containers irrigated only with tap water maintained near optimal levels of nutrients 42 d after initial irrigation.

Extraction method affected Ca values. Like EC, NO_3^- , P, and K values, PourThru Ca values ($295.4 \text{ mg}\cdot\text{L}^{-1}$) were higher than SME Ca values ($193.3 \text{ mg}\cdot\text{L}^{-1}$) due to lack of dilution of the PourThru extract compared to SME extract (Yeager et al., 1983; Wright et al., 1990). The Ca values of PourThru and SME were compared and calibration curves and an inverse calibration equation were developed to predict values for years one and two (Fig. 1F, G and Table 1).

Fertilizer rate affected Ca concentrations. The 13N-0.88P-10.8K Cal – Mag fertilizer provides 6% Ca ($12, 18, \text{ or } 24 \text{ mg}\cdot\text{L}^{-1}$ for the N rates chosen in this study). Therefore, as fertilizer rates increased ($200, 300, \text{ or } 400 \text{ mg}\cdot\text{L}^{-1} \text{ N}$), so did the Ca substrate concentration ($177.6, 237.4, \text{ and } 318.1 \text{ mg}\cdot\text{L}^{-1}$). As with EC, NO_3^- , K, and P values, Ca substrate values were not affected by sampling date (data not presented).

Magnesium (Mg) extraction values were not affected by year (data not presented). However, an extraction by rate interaction affected Mg substrate values (Table 4). Similar to EC, NO_3^- , P, K, and Ca values, Mg PourThru values were higher than SME Mg values and the values increased with increasing fertilizer rates

for both extraction methods. PourThru Mg values are higher due to lack of dilution compared to SME Mg values (Yeager et al., 1983; Wright et al., 1990).

The Mg values of PourThru and SME were compared and a calibration curve was developed to predict values (Fig. 1H). Table 1 lists an inverse of the calibration equation so that SME values could be predicted based upon PourThru values. As with EC, NO₃, K, P, and Ca values, Mg substrate values were not affected by sampling date (data not presented).

Conclusion

Calibration curves were established for EC, NO₃, P, K, Ca, and Mg (Fig. 1). Due to lack of differences in our study, PourThru pH versus SME pH_{slurry} and PourThru pH versus SME pH_{vacuum extract} as well as NH₄⁺ calibration curves were not developed.

Differences between years were noted with P and Ca, which were attributed to the initial “starter” charge difference in the commercial substrate. The commercial substrates contain an initial nutrient charge to counteract substrate acidity and aid in plant nutrition. The exact contents of these starter charges are proprietary and are frequently adjusted to compensate for differences observed in the substrate chemical properties.

For P, year one had lower initial concentrations than year two. This lower concentration likely allowed P to bind with more affinity to substrate particles due to a lower ionic strength substrate solution. Since P was bound to particles with more affinity, less P was “freely” available to the unadulterated PourThru substrate

solution. Therefore, PourThru P extraction values were not as high, relative to SME values, in year one compared to year two. The year one calibration curve should be used for P comparisons, as most substrate levels will be less than $100 \text{ mg}\cdot\text{L}^{-1}$ P. The P values presented in Table 5 were based upon the calibration curve obtained in year one.

For Ca, year one had higher initial concentrations than year two. This higher concentration may have saturated the substrate binding sites. Therefore, when the substrate samples were diluted (SME), a greater amount of Ca was able to move into the bulk solution (year one) than would have if the binding sites were not saturated (year two). The result was that PourThru and SME Ca values were closer in year one than in year two. The year one calibration curve should be used for Ca comparisons, as most Ca substrate values will generally be higher than $250 \text{ mg}\cdot\text{L}^{-1}$. The Ca values presented in Table 5 were based upon the calibration curve obtained in year one.

In agreement with Yeager et al. (1983) and Wright et al (1990), a relationship exists among PourThru and SME values. In our study, PourThru nutrient values (excluding pH and NH_4) were approximately 1.4 to 1.6 times higher than SME values, except for Ca (year one) whose relationship was approximately 1.2 times higher (Fig. 1A – H). This was similar to Yeager et al. (1983) who reported PourThru values approximately 1.3, 1.4, and 1.2 times higher than SME values for N, P, and K, respectively; albeit, calibration curves were not developed to describe the relationship in their study. Yeager also noted that pH values were similar, which agrees with our findings.

However, we did not find a similar relationship of PourThru and SME values as described by Wright et al. (1990) who noted that PourThru values were nearly double that of SME values. These differences are likely due to variances in testing procedures (water content of the SME) that may have altered nutrient values.

Based on the calibration curves developed in our study and recommended SME values (Warncke and Krauskopf, 1983), recommended-PourThru nutrient values for greenhouse substrates have been developed (Table 5). Additionally, calibration curves (Table 1) that allow substrate-testing laboratories to convert PourThru values to SME values and compare those values to established standard values have been presented.

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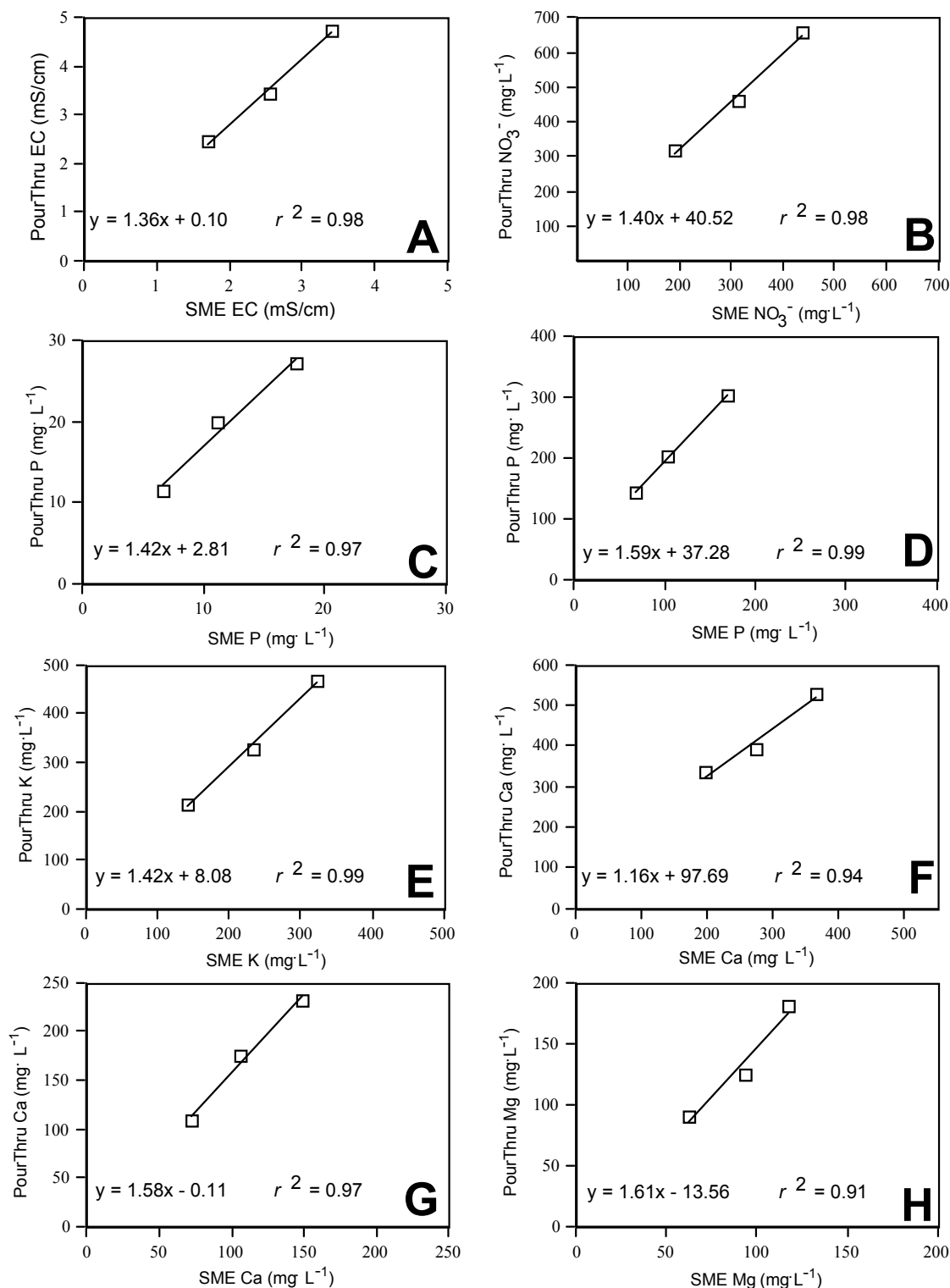


Fig. 1. Calibration curves and equations for comparison of PourThru and SME nutrient values over three fertilizer rates for poinsettia 'Freedom Red' for (A) EC, (B), NO₃, (C) P – year one, (D) P – year two, (E) K, (F) Ca – year one, (G) Ca – year two, and (H) Mg.

Table 1. Inverse calibration equations for PourThru and SME comparisons. PourThru extraction values are represented as the independent variable (x) and SME values are represented as the dependent variable (y).

Nutritional		
Parameter	Regression Equations	Adjusted r^2
EC	$y = 0.74x - 0.05$	0.98
NO ₃	$y = 0.71x - 25.13$	0.98
^z P	$y = 0.69x - 1.74$	0.97
^x P	$y = 0.63x - 23.39$	0.99
K	$y = 0.70x - 4.66$	0.99
^z Ca	$y = 0.84x - 73.43$	0.94
^x Ca	$y = 0.63x + 1.55$	0.97
Mg	$y = 0.59x + 12.17$	0.91

^z Equation is based upon values obtained in year one.

^x Equation is based upon values obtained in year two.

Table 2. Year by week interaction effect on P nutrient extraction values. Means are an average of 5 replications per treatment.

Year	Week	P
		(mg L ⁻¹)
1	6	15.8
	9	15.5
2	6	189.0
	9	141.7
<i>Interaction</i>		
Year * Week		0.0004 ^z

^z $P > F$

Table 3. Year by fertilization rate interaction effect on P nutrient extraction values. Means are an average of 5 replications per treatment.

	Rate	P
Year	(mg·L ⁻¹)	(mg·L ⁻¹)
1	200	9.1
	300	15.5
	400	22.4
2	200	106.6
	300	153.5
	400	236.5
<i>Interaction</i>		
Year * Rate		0.0001 ^z

^z $P > F$

Table 4. Extraction method by fertilization rate interaction affect Mg nutrient extraction values. Means are an average of 5 replications per treatment.

Extraction	Rate (mg L ⁻¹)	Mg (mg L ⁻¹)
PourThru	200	91.2
	300	125.6
	400	182.3
SME	200	61.9
	300	93.3
	400	117.5
<i>Interactions</i>		
Extraction * Rate		0.0045 ^z

^z $P > F$

Table 5. Recommended PourThru substrate values transformed from

^zSME substrate values.

Nutritional parameter	PourThru	SME
^y pH	5.6 – 6.0	5.6 – 6.0
EC (mS/cm)	2.8 – 4.8	2.0 – 3.5
NO ₃ (mg·L ⁻¹)	180 – 320	100 – 199
^x P (mg·L ⁻¹)	11 – 16	6 – 9
K (mg·L ⁻¹)	220 – 360	150 – 249
^x Ca (mg·L ⁻¹)	330+	200+
Mg (mg·L ⁻¹)	100+	70+

^z Values adapted from Warncke and Krauskopf, 1983.^y pH values are equivalent for both extracts.^x Values based upon year one data.

Chapter 3.

Irrigation Systems Affect PourThru Nutrient Extraction Values

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Irrigation Systems Affect PourThru Nutrient Extraction Values

Additional index words. Bulk solution displacement, EC, electrical conductivity, leachate, *Pelargonium xhortorum* 'Rocky Mountain Salmon', pH, soil testing, soluble salts,

Abstract. The type of irrigation system used affects water content and solute movement; thus, affecting electrical conductivity (EC), nutrient, and pH values within a container. However, the PourThru nutrient extraction method has not been investigated across irrigation systems. Geraniums were grown and PourThru pH, EC, and leachate volume values were compared among over-head hand irrigation (HI), drip-tube irrigation (DI), and subirrigation systems (SI). PourThru pHs were lower for the HI treatment versus the DI or SI treatments. However, the differences observed in pH values would not likely have practical implications in commercial floriculture production. PourThru EC values were much higher for the HI treatment versus DI or SI treatments due to the salts being flushed towards the bottom of the container by the bulkhead water movement. When data were pooled by irrigation system, the HI values were two times higher than the DI and SI values. This large variation in EC values over irrigation systems warrants alternative interpretive standard PourThru EC values dependent upon the irrigation system used.

Introduction

Numerous irrigation systems are used in greenhouse production ranging from manual overhead hand irrigation, to automated boom irrigation, to drip tubes, to flood floor subirrigation systems. The type of irrigation system used can affect water content and solute movement; thus, affecting electrical conductivity (EC), nutrient, and pH values within a container (Argo and Biernbaum, 1995; Blom and Piott, 1992; Dole et al., 1994; Guttormsen, 1969; Morvant et al. 1997). However, the PourThru nutrient extraction method has not been investigated as a method to obtain those values.

Overhead-hand irrigation (HI) is the traditional method of watering floriculture crops. Overhead-hand irrigation causes a bulkhead of water to move quasi-uniformly down through a container, but can increase substrate compaction and is labor intensive (Fonteno et al., 1981). The water head movement promotes uniform substrate wetting within a pot. Additionally, the water head movement moves solutes via mass flow and displacement to the bottom portion of the container. Morvant et al. (1997) noted that EC was higher in the middle and bottom region of the pot when plants were irrigated by HI versus SI (ebb and flow) or DI. Drip-tube irrigation is a reduced-labor irrigation method versus HI. Similar to HI, salts can move to the bottom of the container via mass flow and displacement. However, there is a significant amount of capillary movement of water with DI and solutes due to a lack of dominant bulk-head water movement. This promotes movement of water and solutes towards the middle and upper 1/3 of the container as noted by Morvant et al. (1997) who observed higher EC values in the top region of the pot for

DI versus HI. Additionally, DI does not provide the high rate of irrigation to uniformly wet the top of the substrate. Over time, the salts tend to accumulate due to evaporation of water from the substrate surface (Argo and Biernbaum, 1995). This salt accumulation and dry substrate limit root growth; thus, enhancing salt accumulation from lack of nutrient uptake.

Subirrigation systems allow automation of large areas and are commonly used in large greenhouse-floriculture operations. Benefits of SI are reduced foliar-disease spread, reduced labor, and prevention of nutrient run-off (Lieth, 1996; Morvant et al., 1997). Water movement within a container that is subirrigated is via capillary action. Therefore, solutes may accumulate in the top 1/3 of the substrate because there is no bulk water head movement to force the solutes towards the lower portion of the container. Over time, the salts tend to accumulate due to evaporation of water from the substrate surface and root growth is inhibited (Argo and Biernbaum, 1995).

Compared to the 1:2 (v/v) volumetric slurry (1:2 VS) or the saturated media extract (SME), in which the nutrient samples can be altered dependent upon where the substrate sample is taken from the container, the PourThru extraction (an in-situ method) does not allow for user control of where the nutrient extraction sample is taken. The ability to alter values with 1:2 VS and SME has allowed greenhouse producers to use a single set of nutrient standard values or producers acknowledged and compensated for the differences regardless of irrigation system used. Producers may collect root substrate from the middle of the container or remove a small volume of substrate from the entire container profile (Cavins et al., 2001).

Consequently, potential variable substrate sample locations causes confusion (Argo and Biernbaum, 1995).

PourThru procedures may need to be altered or multiple standard values may need to be established dependent upon crop and irrigation systems used. PourThru works by displacing the substrate solution out of the container (Wright, 1986). Bilderback and Fonteno (1987) noted that a majority of the substrate solution resides in the lower 1/3 of the container. Therefore, PourThru nutrient values likely only represent pH and EC values from the lower portion of the rootball of which nutrient status can be influenced by irrigation method. Handreck (1994) noted that pH values are similar to SME values obtained from the substrate removed from the basal portion of the container.

Due to the anomalous salt distribution among irrigation systems, PourThru pH and EC values that represent only the lower 1/3 of the rootball would not represent the entire rootball pH or EC and would likely differ among irrigation systems. For DI and SI production systems, PourThru may underestimate EC values for the entire rootball, as EC values tend to be higher in the top of the container versus the bottom (Argo and Biernbaum, 1995, Morvant et al., 1997). The objective of this study was to determine the effects of HI, DI, and SI systems on PourThru pH and EC values.

Materials and Methods

Geraniums (*Pelargonium xhortorum* L.H. Bail.) 'Rocky Mountain Salmon' were grown in 15.4 cm (6 inch) plastic containers using Fafard 4P root substrate (Conrad Fafard Inc., Anderson, S.C.). All containers were placed in hydroponic troughs (Bato Trading B.V., The Netherlands) with drainage holes and plugs for leachate collection to determine leaching fraction and fertigated by hand [333 mL/pot (11.3 oz) by watering can with breaker], drip tube [350 mL/pot (11.8 oz) with pressure compensated emitters rated at $95 \text{ mL} \cdot \text{s}^{-1}$ (1.5 gal/min)], or sub-irrigation [troughs were filled with fertigation solution to approximately 2.5 cm (1 inch) up the pot side, allowed to equilibrate for 12 minutes and drained] methods with $200 \text{ mg} \cdot \text{L}^{-1}$ (ppm) N from 13N-0.88P-10.8K (13-2-13) fertilizer (The Scotts Co., Marysville, Ohio). Plants were irrigated when three randomly selected plants from each irrigation treatment averaged 40% of the available water content (Morvant et al., 1997). PourThru extractions were performed at 2, 4, 6, and 8 weeks after planting of rooted cuttings.

Data collected included PourThru pH and EC, leaching fraction, plant height and diameter, and plant dry weight. This study was a completely randomized design with three irrigation methods and six replications with six subsamples for each treatment. Data were analyzed using procedure general linear models and protected interactive LSD (SAS Institute, Cary, N.C.).

Results and Discussion

An irrigation system by sampling date interaction affected pH, EC, and leachate volume. The pH values for HI plants were generally lower than DI and SI in corresponding weeks except for SI during week eight, when pH values were similar to HI values (Fig. 1A). Morvant et al. (1997) found similar results such that HI produced lower pH values than SI when comparing only the lower portion of the substrate. Due to the nature of displacement, PourThru only samples nutrient solution from the lowest portion of the container (Bilderback and Fonteno, 1987; Handreck, 1994; Wright, 1986). The lower pH is likely a result of higher water content of HI plants (577.79 g, average weight of container, substrate, plant and water prior to irrigation) versus the DI and SI plants (552.93 and 529.86 g, respectively). A moist substrate condition can promote an increase in N reduction (mineralization) and organic acid release from the substrate, which may account for the lower pH values with HI plants (Morvant et al., 1997; Sparks, 1995).

The pH values were highest for DI during week eight and a pH increase was observed with all irrigation treatments during weeks four to six (Fig. 1A). The higher pH values observed at the end of the crop are a result of the basic residues of the 13N-0.88P-10.8K fertilizer, which has a potential basicity of 155 kg/907 kg calcium carbonate (342 lb/ton).

Hand irrigation produced the highest EC values and the values increased from week two to four and then from six to eight (Fig. 1B). Electrical conductivity values were higher for HI due to the downward movement of solutes within the substrate from the bulkhead of water formed when overhead-hand irrigating and the

low leaching fraction (9.9%) compared to DI (20.5%), which has limited bulk head water movement (Argo and Biernbaum, 1995, Morvant et al., 1997). A majority of the substrate solution resides in the lower 1/3 of the container (Bilderback and Fonteno, 1987); therefore, the higher EC values were reflected in the displaced PourThru sample from HI plants.

Values obtained from HI plants were within the EC range (2.0 to 3.5 mS/cm) for geraniums recommended by Whipker (2000). Hand irrigation, DI, and SI had similar heights (12.35, 15.72, and 15.22 cm, respectively), diameters (22.6, 24.8, and 28.27 cm, respectively) and shoot dry weights (22.6, 24.8, and 24.2 g, respectively) (values collected at termination of study, week 8). These similarities indicate that all three irrigation methods provided adequate nutrition despite variation in EC values.

Drip tube and SI treatments had similar trends such that EC decreased from weeks two to six and then increased from week six to eight (Fig. 1B). The depletion of nutrients from plants in the rapid vegetative state of growth led to the initial decline in EC values (Whipker and Hammer, 1997). Albeit adequate nutrients were supplied, the increased stratification of solutes due to evaporation of water from the surface of DI and SI containers compared to the HI containers (which were sufficiently rewetted every irrigation) resulted in lower EC values in the lower 1/3 of the containers where PourThru displacement occurs (Argo and Biernbaum, 1995, Morvant et al, 1997).

The observed EC increase from week six to eight is due to salt accumulation (Fig. 1B). Over time, the solute build up in the top of the container presumably migrated downward. Therefore, the high EC values were detected in the PourThru

leachate as it was obtained from the lower 1/3 of the container. The EC values obtained for DI and SI were below the recommended values (2.0 to 3.5 mS/cm) noted by Whipker (2000) for actively growing geraniums.

PourThru leachate volumes were generally greatest for HI containers, except during week six (Fig. 1C). The higher leachate volumes were due to more uniform moisture contents for the HI containers versus the DI and SI containers (Argo and Biernbaum, 1995; Morvant et al, 1997). In our study, HI containers generally had higher moisture volumes (577.8 g, weight of container, substrate, plant, and water) prior to irrigation versus DI or SI irrigated plants (552.9 and 529.9 g, respectively).

Sub-irrigated containers appeared to be the most consistent for leachate volumes. A decrease in PourThru leachate volume was observed between weeks two and four; however, weeks four, six, and eight were similar. This indicates that SI was the most uniform irrigation treatment in our study.

Conclusion

Irrigation system did affect pH values; however, for the corresponding week, the greatest difference between irrigation treatment pH values was 0.27, which is within the breadth of soilless substrate pH recommendation (0.6 pH units) (Bailey, 1996). Therefore, it was not necessary to develop alternative pH recommendations based upon irrigation systems. Morvant et al. (1997) noted a 0.46 pH variation among HI, DI, and SI when sample were taken from the lower 1/3 of the container.

Electrical conductivity values differed among the irrigation systems. This was consistent with findings by Argo and Biernbaum (1995) and Morvant et al. (1997) and was attributed to the stratification of salts due to the physics of water movement, which was influenced by the irrigation treatment. For HI Morvant et al. (1997) noted salt stratification was greatest in the lower 1/3 of the container, the portion of the container sampled by PourThru displacement, and the HI containers in our study were generally within the recommended EC levels (2.0 to 3.5 mS/cm) (Whipker, 2000). Salt stratification with DI and SI containers was greatest in the upper 1/3 of the container, the portion not sampled by PourThru displacement (Morvant et al., 1997). This resulted in EC values with DI and SI containers that were below recommended values; albeit, provided the same fertilizer concentration. Even with different PourThru EC values, plant heights, diameters and dry weights were similar for all three irrigation treatments.

Consequently, alternative PourThru EC recommendations were developed for DI and SI systems as HI EC values were approximately two times higher than DI and SI containers (pooled data of DI and SI treatments) (Table 1). In contrast, with

SME or 1:2 VS extractions one recommendation can be made for all irrigation systems since samples can be taken excluding the upper 1/3 of the container (Argo and Biernbaum, 1995). Because PourThru is a non-destructive technique that displaces the bulk solution from only the lower 1/3 of the container, customized EC interpretation values were required to account for the EC differences among irrigation methods. Further studies should be conducted to evaluate if altering PourThru procedures (i.e. overhead-hand irrigating prior to PourThru analysis, regardless of normal irrigation treatment) are viable alternatives to separate EC recommendations.

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Table 1. PourThru electrical conductivity – recommended values for geraniums based upon irrigation systems and non-irrigation specific SME and 1:2 VS values.

Irrigation system	Electrical conductivity (mS/cm)
^z Overhead / hand	2.00 - 3.50
^y Drip tube or subirrigation	1.00 – 1.70
^z SME	1.50 – 3.00
^x 1:2 VS	0.75 – 1.25

^z Whipker, 2000.

^y Overhead / hand or subirrigation EC x 0.49.

^x Dole and Wilkins, 1999.

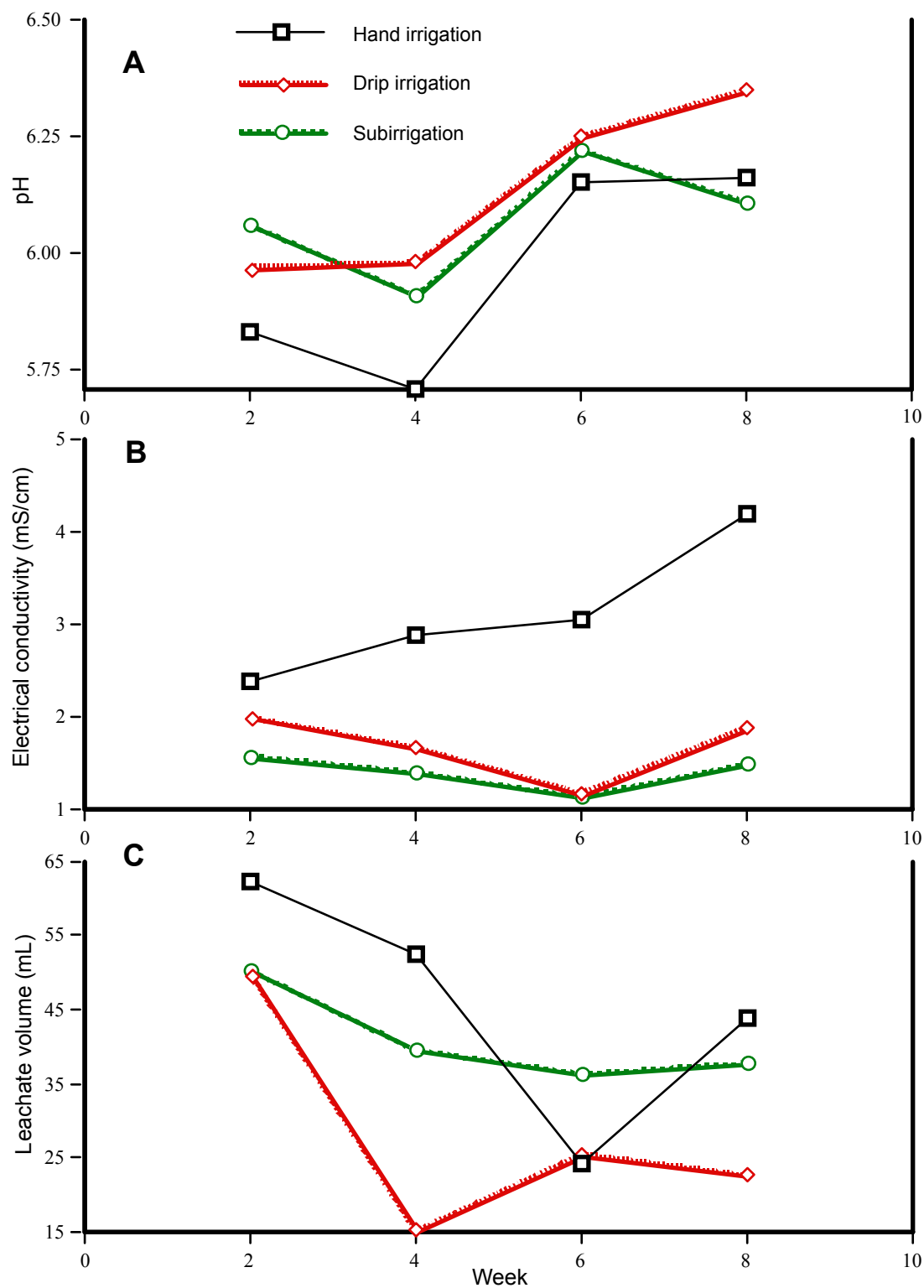


Fig. 1. Irrigation system effect on (A) pH ($LSD_{0.05} = 0.09$), (B) EC ($LSD_{0.05} = 0.17$), and (C) leachate volume ($LSD_{0.05} = 7.44$). Data points represent means of six replications with six subsamples each.

Chapter 4.

POURTHRU TIMING EFFECTS ON pH, ELECTRICAL CONDUCTIVITY, AND LEACHATE VOLUME¹

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POURTHRU TIMING EFFECTS ON pH, ELECTRICAL CONDUCTIVITY, AND LEACHATE VOLUME

Abstract. The time between irrigation and PourThru sampling may affect pH, EC, and leachate volume and has not been examined for greenhouse production. A greenhouse study with varying pot sizes and substrates was implemented and PourThru leachates were tested for pH, EC, and volume at 15, 30, 60, 120, and 240 minutes after irrigation. Substrates and pot sizes affected PourThru pH and EC; however, timing did not affect values in our study. Leachate volumes and mass wetness of the substrate were dependent upon elapsed time between irrigation and sampling. Timing of the PourThru was important to ensure adequate sample volume for analysis and timing may be more critical in smaller volume containers than used in our study.

Introduction

PourThru is a bulk solution displacement method that is increasing in popularity (1-4). This simple substrate solution extraction can be easily performed in the greenhouse without special equipment to obtain pH and electrical conductivity (EC) and sent to commercial or university laboratories for complete nutritional analysis. The ability of horticultural producers, consultants, and researchers to save valuable time for nutrient analysis can vastly influence the profitability of a crop. On-site analysis allows fertility regime adjustments in a timely manner.

The time between irrigation and PourThru testing may affect pH, EC, and leachate volume and has not been examined for greenhouse production systems. Dependent upon the time interval, rapid changes of water content in greenhouse containers may occur. Water content of the substrate at the time of PourThru did not affect PourThru values when large nursery containers (3 L) with a sand bark substrate were investigated (5). However, typical floriculture production uses smaller containers than those investigated in the previous study (5). Therefore, water content may be important due to the small volume of substrate and the substrate components (i.e. peatmoss and vermiculite) used in floriculture production systems. Reitemeier (6) noted that water content affects nutrient availability.

Wright (7) noted that containers must be near “water holding capacity” when the PourThru extractions are performed. “Near water holding capacity” would likely only exist immediately after irrigation in typical greenhouse conditions (high light and warm temperatures). However, if PourThru extractions were performed immediately after irrigation non-representative values may be attained. Compton and Nelson (8)

noted that substrate testing should be performed one to two hours after fertilization to allow for nutrient equilibration and to obtain values that represent nutrient availability.

Wright et al. (9) waited two hours between irrigation and completion of the PourThru extraction. Two hours may be sufficient time for the water content to change drastically from container capacity due to plant uptake or evaporation; thus, affecting nutrient concentration in the bulk solution (6). Wright et al. (9) also noted that 100 mL of displacement solution was used to obtain the leachate from a 15 cm wide container, but did not report the volume of leachate collected. Leachate volume is important to ensure adequate solution for testing and a representative sample. A volume of 50 mL has been suggested as the target volume of leachate to extract from a 1 to 12 L container (7). The objective of this study was to determine the affect of PourThru timing and substrate moisture status on pH, EC, and leachate volume.

Materials and Methods

Helianthus annuus L. 'Teddy Bear' seed was sown 15 April 2002 and transplanted on 29 April 2002 into 16.5 (1700 cm³) or 19.1 cm (2650 cm³) containers with Fafard 4P (Conrad Fafard Inc., Anderson, S.C.) or Scotts Metro Mix 320 high lime with coir (The Scotts Co., Marysville, Ohio). Glass covered greenhouses were set at 18/24 °C, night/day, respectively and plants were fertigated with a 150 mg·L⁻¹ N from a 20N-4.4P-16.6 K fertilizer (The Scotts Co., Marysville, Ohio). PourThru extractions using 100 mL for the 16.5 cm containers or 125 mL for the 19.1 cm

containers of distilled water were performed 15, 30, 60, 120, and 240 minutes after irrigation at 4, 5, and 6 weeks after transplanting of seedling.

Extractions were analyzed for pH, EC, and volume. Data collected also included mass wetness (g of water/g dry substrate) of the substrate [(whole container wet weight – shoot fresh weight – substrate dry weight – container weight)/substrate dry weight]. Additionally, soil moisture tensions were recorded by placing a vertically oriented porous cup (length = 2.9 cm) attached to a mercury manometer tensiometer at the substrate vertical midpoint. Soil moisture release curves were developed for the respective substrates to predict the relationship of substrate moisture content and substrate moisture tension (Fig. 1). The study was a split-split plot with a 2 x 2 x 5 x 3 factorial treatment combination. There were 2 substrates, 2 containers, 5 PourThru timings, and 3 sampling dates with 5 replications per treatment. Data were analyzed by general linear models and means separation was performed using a protected LSD ($P \geq 0.05$) (SAS Institute, Cary, N.C.).

Results and Discussion

A substrate by sampling date interaction affected pH (LSD = 0.06) (Fig. 2A). The Metro Mix 320 high lime substrate (MM320) had a higher pH than the Fafard 4P. The MM320 is a substrate that has an additional lime charge, versus conventional horticulture substrates, to counteract substrate component acidity (K. Santner, The Scotts Company, personal communication). The substrate pH generally decreased over crop time. This was due to the use of the 20N-4.4P-16.6 K

fertilizer, which has an acidic residue potential of 184.2 kg calcium carbonate (CaCO_3) equivalent per 1016 kg (10) due to plant uptake of NH_4^+ , which acidifies the root environment (11,12). The repeated fertilizer applications and plant NH_4^+ uptake counteracted the limestone charge in the substrate, resulting in a lower pH over time.

A container by sampling date interaction also affected pH ($\text{LSD} = 0.06$) (Fig. 3A) The 16.5 cm containers had higher pH values than the 19.1 cm containers except during week 6 and the pH decreased over time for the 16.5 cm containers. In contrast, the pH did not decrease over time for the 19.1 cm containers. The lack in pH decrease for the 19.1 cm containers was presumably due to the greater substrate volume (approximately 2000 cm^3 of substrate), which had sufficient buffering capacity with withstand pH shift compared to the smaller 16.5 cm container (approximately 1300 cm^3 of substrate).

Timing of the PourThru did not affect pH (data not shown). This was likely due to the high volume of substrate, which enhanced the buffering capacity of the system and prevented plant nutrient uptake from altering the soil solution within 240 minutes of irrigation. Therefore, timing was not the dominant factor affecting substrate pH. Conversely, Compton and Nelson (8) noted that EC and various nutrients decrease rapidly within 4 h of irrigation. However, their results were based upon smaller substrate volumes (6.5 cm^3 /plant) than ours (1300 or 2000 cm^3).

The EC was affected by the substrate such that the Fafard 4P had lower EC values than the MM320 (1.43 and 3.27 mS/cm , respectively; $\text{LSD}_{0.05} = 0.13$). The MM320 contained coir fiber as a component. Handreck (18) noted that coir fiber

yields higher EC values than conventional peatmoss substrates. The MM320 also did not drain as readily as Fafard 4P, which was indicated by its higher container capacity [75 versus 70 % (v/v), respectively], that led to more salt accumulation in the substrate. Container size, sampling date, or timing of PourThru did not affect the EC. Similar to pH, timing of the PourThru likely did not affect EC due to the large substrate volume.

Timing of the PourThru affected leachate volumes (LSD = 7.55)(Fig. 4). No differences were observed between the 15, 30, and 60-minute interval extractions (55.5, 58.3, and 54.7 mL, respectively); however, leachate volumes decreased when 120 (36.0 mL) or 240 (18.7 mL) minutes elapsed between irrigation and sampling. This indicates that samples should be collected 60 minutes after irrigation to ensure a sufficient volume of leachate extraction (7) and representative sample.

A substrate by sampling date interaction affected leachate volumes (LSD = 8.27)(Fig. 2B). The Fafard 4P produced less leachate than the MM320 and the volumes were lowest during week 6 sampling. The MM320 did not drain as readily compared to the Fafard 4P, as noted above. Leachate volumes during week 6 were presumably lower due to high plant water uptake versus the other sampling dates.

A container by sampling date interaction affected leachate volumes (LSD = 8.27)(Fig. 3B). The 16.5 cm containers produced less leachate than the 19.1 cm containers. This is due to a lower substrate volume, thus, a lower water holding capacity in the 16.5 cm container. Over time, leachate volumes generally decreased. The 19.1 cm containers during week 4 had the highest leachate volume and the least amount was obtained during week 6 for both containers. Leachate volumes

during week 6 were lower due to high plant water uptake versus the other sampling dates.

Mass wetness decreased after irrigation such that the moisture content was similar 15 to 60 minutes after irrigation and then decreased at 120 and 240 minutes (LSD = 0.23) (Fig. 4). This drop in moisture content beyond 60 minutes indicates that changes in nutrient status might be different with late testing (more than 60 minutes of elapsed time between irrigation and testing). In our study, we did not test specific nutrients, which may have reflected changes in concentration (8). Yeager et al. (5) noted that EC and NO_3^- values decrease in PourThru extracts when substrate volumes were dry and Compton and Nelson (8) noted testing should be done a minimum of 60 minutes after fertigation to obtain a representative sample of plant available nutrients.

Similar to leachate volumes, mass wetness was affected by a substrate by sampling date interaction (LSD = 0.25)(Fig. 2C). Fafard 4P had lower mass wetness values than the MM320. For MM320, week 4 had the lowest values and was similar to the Fafard 4P during the week 6 sampling date. The highest mass wetness values were obtained from the MM320 during weeks 5 and 6. Container size did not affect mass wetness. This was because mass wetness values are a representation of relative water content (mass of water per mass of dry substrate).

A substrate by sampling date interaction affected tensiometer values (LSD = 5.36)(Fig. 2D). Week 5 tensiometer values were low for both substrates. For sampling dates 4 and 6, MM320 had the lowest tensions indicating the substrate

held more water than the Fafard 4P, which is in agreement with the moisture release curve generated for the substrates (Fig. 1) and leachate volume findings.

A container by sampling date interaction affected tensiometer values ($LSD = 5.36$)(Fig. 3C). At week 4, the 19.1 cm container had higher tensiometer values than either the 19.1 or 16.5 cm containers during week 5. All other tensiometer values were similar. Leachate volume and mass wetness were also affected similarly to tensiometer values. This was expected as the tensiometer values reflect water holding capacity of a mix and the amount of water in the substrate would determine the amount of leachate displaced for a given influent volume.

Timing of the PourThru did not influence tensiometer values within 240 minutes of irrigation. This was due to the placement of the porous cup in the middle of the substrate. Most moisture loss was likely in the upper portion of the container due to evaporation and gravity (13).

Conclusion

Timing of PourThru affected the substrate mass wetness and leachate volume such that the leachate volume decreased when 120 or 240 minutes elapsed from irrigation to sampling. A correlation mass wetness and volume of leachate did occur indicating that moisture content of the substrate is important ($r^2 = 0.70$). However, timing did not affect pH or EC. This was likely due to overriding factors such as substrate components and container volume. In contrast, Compton and Nelson (8) noted EC and nutrients concentrations change within 240 minutes of irrigation in small substrate volume containers.

Based upon the fluctuations in leachate volumes and mass wetness values, we recommend 60 minutes elapse from time of irrigation to PourThru sampling. Sixty-minutes is sufficient time to allow for nutrient equilibration so the greenhouse crop producers can obtain a representative sample of the plant available nutrient status, yet maintain sufficient moisture status to prevent EC shifts due to moisture content variation leachate volumes from becoming too low (8,14).

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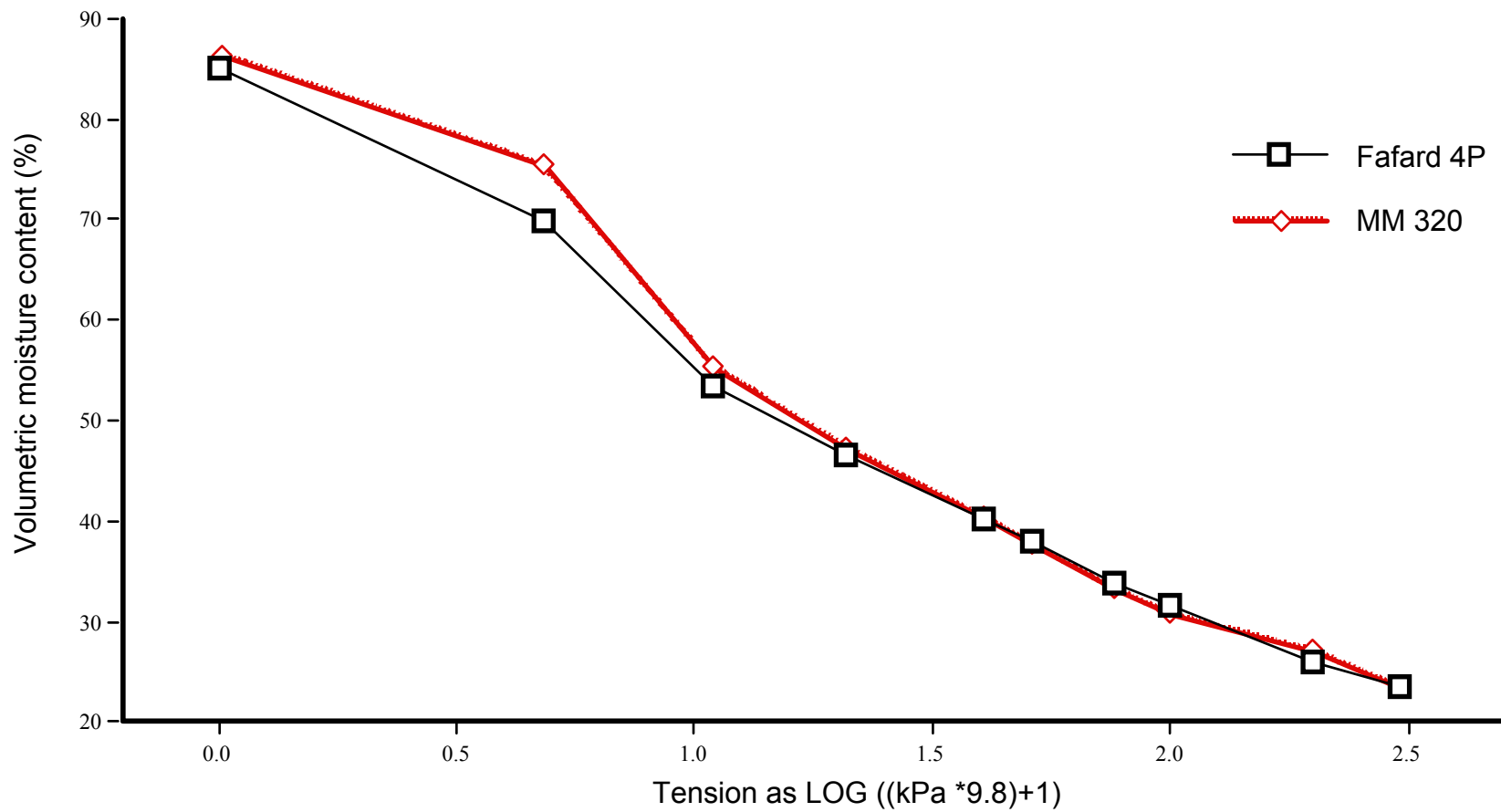


Fig. 1. Soil moisture tension curves and cubic regression equations used to describe them. Data points were experimentally determined (adapted from Karlovich and Fonteno, 1986).

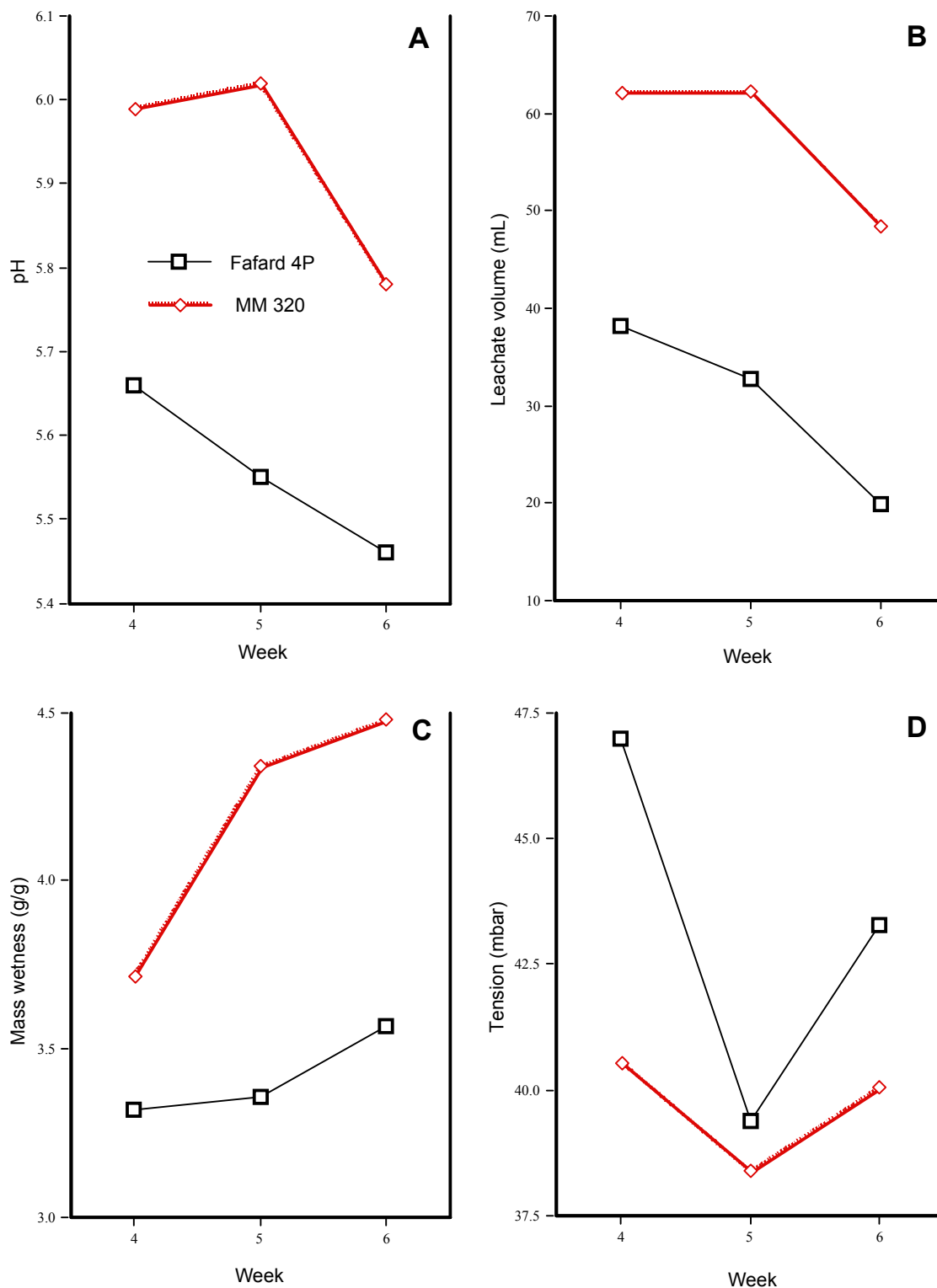


Fig. 2. Substrate by sampling date interaction effect on substrate pH, leachate volume, mass wetness, and soil moisture tension. Means are an average of five replications per treatment.

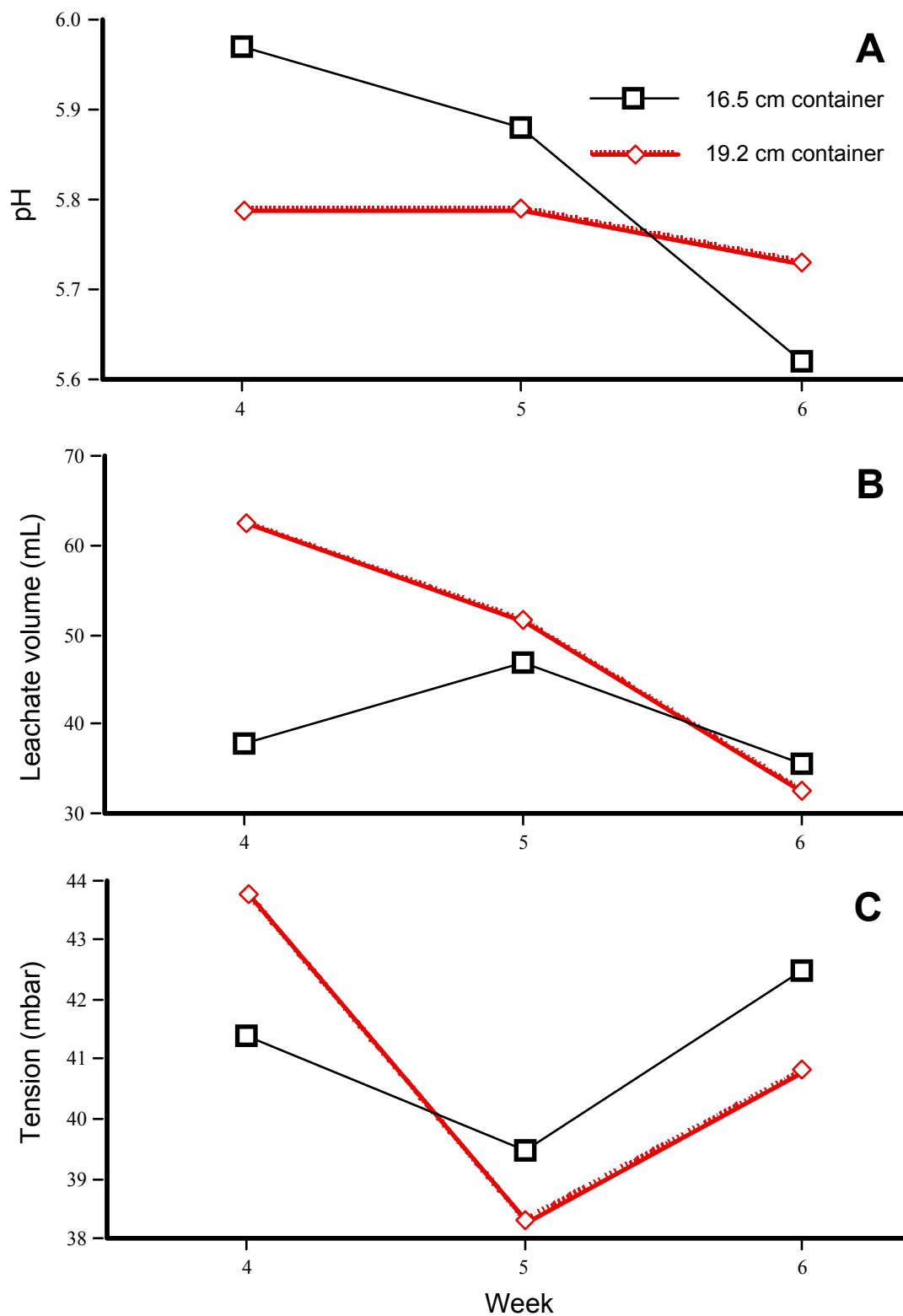


Fig. 3. Container by sampling date interaction effect on substrate pH, leachate volume, and soil moisture tension. Means are an average of five replications per treatment.

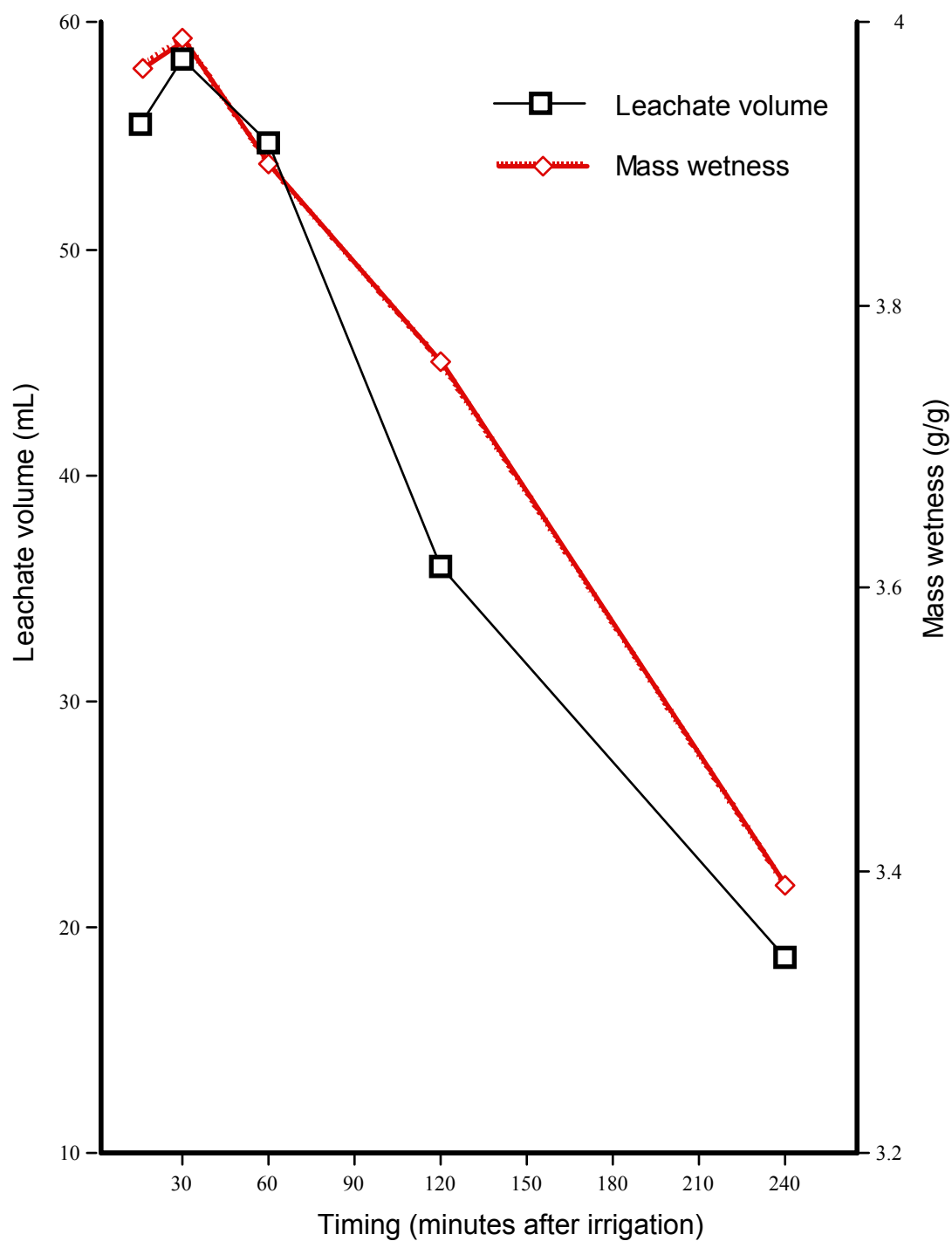


Fig. 4. PourThru timing affects leachate volume and mass wetness. Means are an average of five replications per treatment.

Chapter 5.

MIXING OF DISPLACEMENT SOLUTION AND BULK SOLUTION WITH POURTHRU EXTRACTION¹

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MIXING OF DISPLACEMENT SOLUTION AND BULK SOLUTION WITH POURTHRU EXTRACTION

Abstract. Two experiments were conducted to determine the effects of container size, substrate type, and crop time on PourThru substrate solution samples. Experiment 1-*Helianthus annuus* L. 'Pacino' was grown in 15.2 cm containers and various influent volumes were used to displace a substrate solution sample throughout the crop. Experiment 2-*Salvia farinacea* Benth. 'Reference' was grown in 1203 or 1801 bedding plant cell packs and 10.2 or 12.7 cm containers with either Fafard 4P commercial root substrate or 3 bark : 1peatmoss: 1 sand (v/v/v) substrate and various influent volumes were used to displace a substrate solution sample. Crop time affected the PourThru samples, however, the effects were likely due to environmental variations not controlled or recorded in this study. The bedding plant cell packs, small containers, and the highly porous 3:1:1 substrate were more susceptible to mixing of the influent and substrate solution versus the large containers and the less porous Fafard 4P. High influent volumes appeared to be the most influential factor such that PourThru leachate volumes and electrical conductivity increased with increasing influent volumes. For the 15.2 cm containers, we recommend 50 to 75 mL of distilled water for the influent volume. For bedding plant cell packs we recommend 15 mL for influent volume and for the 10.2 and 12.7 cm containers we suggest 30 mL.

Introduction

PourThru nutrient extraction is a simple and quick method for obtaining a substrate solution sample. PourThru works by using an influent (generally distilled water) evenly poured over a container of root substrate (1). The influent forms a bulkhead of water, which moves down due to gravity and pressure head that displaces the bulk substrate solution. The bulk solution (leachate) is collected and analyzed for nutritional content.

Bulk solution is analogous to soil solution, which is a quasi-equilibrium solution that contains a particular concentration of electrolytes under saturated conditions (2,3,4). The bulk solution contains plant available water and nutrients. Therefore, an unadulterated bulk solution sample is the ideal testing medium for plant substrate nutritional analysis (5,6).

Depending on influent volumes and other factors such as substrate physical properties, mixing may occur with the bulk solution and influent. Mixing indicates that an adulterated substrate solution was obtained; therefore, nutritional values may be non-representative. Yeager et al. (7) noted that varying the amount of displacement solution applied from 40 to 85 mL did not alter specific nutrient values of the leachate obtained from a 100% pine bark substrate in a 3000 cm³ container immediately following irrigation. However, the study did not address if mixing of the displacement and leachate occurred and the ample time for equilibration of nutrients was not allowed (8). The researchers did note that pH varied depending on influent volume. Mixing of the displacement and leachate solutions likely caused the pH to

be altered. The results indicate that mixing may depend upon the amount of displacing solution added for a given container or substrate volume.

The 40 to 85 mL of influent used by Yeager et al. (7) was a relatively small volume (1.9 to 4.0% of the substrate volume) versus the container and substrate volume used (3000 cm³ container with 2100 cm³ of pine bark) when compared to many containers used in floriculture crop production. To obtain sufficient leachate for analyses (approximately 50 mL per container from 75 mL of influent for a 15.2 cm container)(9), the quantity of influent needed would be a higher percentage (7.5%) of the container and substrate volume (a 15.2 cm container holds approximately 1000 cm³ of substrate under typical greenhouse production conditions).

Previous PourThru mixing studies were conducted without plants, in 100% pine bark substrate, and extractions were performed immediately after containers drained (7). The presence of plants may alter nutrient and pH values and one to two hours is recommended to allow for plant nutrient uptake after fertigation (7,8). Allowing one to two hours for equilibration facilitates a substrate test that measures plant available nutrients versus testing of the fertigation input nutrients. In addition, the 100% pine bark substrate is not representative of those typically used in floriculture substrates (coir, peat, perlite, pine bark, and/or vermiculite mixtures) (10,11).

Crop time may influence PourThru results, as the volume of roots would increase over time. The larger root volume may interfere with the displacement process, requiring an increased or decreased influent volume needed to produce a

given amount of leachate or increased water channeling. Jarrell et al. (6) noted that root development, which would increase over time, can affect substrate pH.

Information on influent volumes, container size, various mixes, and crop time are needed due to the small substrate volumes used in floriculture production, which increases the probability of influent and PourThru leachate mixing. The objective of this study was to determine if various influent volumes resulted in mixing with PourThru leachates in small containers and two substrates.

Materials and Methods

Experiment 1

Helianthus annuus 'Pacino' was sown on 24 Jan. 2001 and transplanted into 15.2 cm (1300 cm³) plastic containers on 7 Feb. 2001 with approximately 1000 cm³ of Fafard 4P commercial growing substrate (Conrad Fafard Inc., Anderson, S.C.) and fertigated with 150 mg·L⁻¹ N from a 13N-0.88P-10.8K (Scotts Co., Marysville, Ohio) fertilizer. Glass covered greenhouses were set at 20/24°C night/day temperatures, respectively. PourThru extractions using influents of 50, 75, 100, and 125 mL were performed one hour after irrigation at 2, 4, 6, and 8 weeks after the transplanting of 2 week old seedlings. The influent contained distilled water with aqueous 1.0% potassium thiocyanate (KSCN)(w/v). Extractions were analyzed for pH and electrical conductivity (EC) and for the presence of thiocyanate by adding 0.5 mL aqueous 0.5% ferric chloride (w/v), which reacts to form ferric thiocyanate (a dark red pigment), if the influent has mixed with leachate (adapted from 3).

This study was a completely randomized design with a 4 x 4 factorial treatment combination. There were four leachate volumes and four extraction dates and six replications per treatment. Results were analyzed using PROC ANOVA ($P \leq 0.001$) and protected LSD ($P \leq 0.05$) for means separation (SAS Institute, Cary, N.C.). Data analyses from the mixing of influent and PourThru leachate were based on arc-sin transformation, then converted to percent mixing (number of replications mixing per treatment divided by six).

Experiment 2

Salvia farinacea 'Reference' was sown on 22 Feb. 2002 and transplanted on 15 Mar. 2002 into 1203 bedding plant cell packs (348 cm³/cell with three plants), 1801 bedding plant cell packs (260 cm³/cell with one plant), 10.2 cm (365 cm³) plastic containers, or 12.7 cm (750 cm³) plastic containers in a 3 bark : 1 peat: 1 sand (v:v:v) or Fafard 4-P commercial rooting substrate (Conrad Fafard Inc., Anderson, S.C.). Plants were fertigated with 150 mg·L⁻¹ N from a 20N-4.4P-16.6 K fertilizer (Scotts Co., Marysville, OH). Polyethylene covered greenhouses were set at 20/24°C night/day temperatures, respectively. PourThru extractions using 1.0% potassium thiocyanate solution of 15, 30, 45, and 60 mL for the bedding plant cell packs and 30, 45, 60, and 75 mL for the 10.2 and 12.7 cm containers were performed one hour after irrigation at 0, 3, and 6 weeks after transplanting of 2 week old seedlings. Extraction volumes were recorded and samples were analyzed for pH and EC and for the presence of thiocyanate by adding 0.5 mL aqueous 0.5% ferric chloride (w/v) (adapted from 3).

Leachates from bedding plant cell packs were analyzed separately from the container plant leachates. The study consisted of two completely randomized designed experiments with a 2 x 2 x 4 factorial treatment combination with two containers as the main plots, and the two substrates and four displacement volumes as the subplot treatments. There were 5 replications per treatment per sampling date ($n = 15$). Results were analyzed using PROC ANOVA ($P \leq 0.05$) and protected LSD ($P \leq 0.05$) for means separation (SAS Institute, Cary, N.C.). Data analyses from the mixing of influent and PourThru leachate were based on arc-sin transformation, then converted to percent mixing (number of replications mixing per treatment divided by five).

Results and Discussion

Experiment 1

Substrate pH was not affected by the treatments in this study (mean pH = 6.28, data not shown). In contrast, Yeager et al. (7) noted that pH decreased with high influent volumes (100 mL for a 3000 cm³ container). Our differences in pH shift may have been due to the different substrates or high solute concentration of the KSCN solution (EC = 9.45 and pH = 6.9), which buffered the leachate. However, Yeager et al. (7) noted the use of distilled water, which should have had negligible effects on pH shift.

Crop time affected EC values (Table 1). During weeks two to six EC values were similar, then increased from week six to eight. This build up of salts was a

result of the sunflower plants entering reproductive growth and using fewer nutrients versus what was used earlier during a period of rapid vegetative growth (12, 13).

Electrical conductivity was not affected by influent volume for experiment one (data not presented). Evidently, when mixing of the influent and leachate occurred, the amount of mixing was not sufficient to alter the EC. Yeager et al. (7) observed similar trends, but our findings from year two disagree.

Crop time affected the volume of leachate such that leachate volumes collected during weeks two and six were similar and weeks four and eight were similar (Table 1). This variation was likely due to uncontrollable factors such as light intensity, which would affect evapotranspiration or inconsistent wetness of substrates that commonly develops in containers; thus, altering the amount of leachate volume.

Influent volume affected leachate volumes (Table 2). As expected, leachate volumes increased with increasing influent volumes (7). The higher volumes of influent satiated the substrate pores more readily than the low influent volumes, once satiated the pore water was displaced by the remaining influent.

Influent volume affected mixing (Table 2). As the influent volume increased, the amount of mixing increased. Little mixing occurred with 50 and 75 mL of influent. However, 100 and 125 mL resulted in increased mixing, respectively. The small amount of mixing with 50 and 75 mL of influent indicates that the volume range was acceptable to obtain an unadulterated substrate solution sample for 15.2 cm containers. Wright (1) suggested that 75 mL be added to a 1000 cm³ container. Crop time did not affect mixing of the influent and PourThru leachate (data not presented).

Experiment 2 -bedding plant cell packs

Substrate type affected pH such that the substrate pH was lower when plants were grown in Fafard 4P (5.76) versus the 3:1:1 substrate (6.74)(LSD = 0.13). The Fafard 4P substrate had a lower initial lime charge compared to the 3:1:1 (initial pH values were 5.58 and 6.53, respectively) and this difference in pH was persistent throughout the crop. We observed no differences in pH among container sizes or influent volumes. As noted above, Yeager et al. (7) observed differences in pH dependent upon influent volumes.

Container size affected substrate EC such that the EC was lower in the 1203 (1.38 mS/cm) versus the 1801 cell packs (1.79 mS/cm)(LSD = 0.25). The 1203 substrate EC may have been lower due to a higher density of plants per volume of substrate (3 plants/348 cm³) compared to the 1801 substrate (1 plant/260 cm³). The high plant density would likely increase fertilizer solution uptake; thus, decreasing EC.

The influent volume caused EC values to change (Fig. 1A). Electrical conductivity values increased with increasing influent volumes as more of the KSCN solution mixed with the leachate. The KSCN solution had an EC of 9.45 mS/cm. Thus, the increase in EC as influent volume increased was a result of the increased concentration of the KSCN solution in the leachate. Yeager et al. (7) generally did not observe a change in nutrient concentration in the leachate because they used distilled water. Therefore, if mixing occurred in their study the lack of solutes in the water would not have increased the EC as it did in our study, or in their study an insufficient volume of distilled water mixed with the leachate to dilute the sample.

The substrate also affected EC (LSD = 0.25). The 3:1:1 (1.85 mS/cm) substrate had a higher EC than the Fafard 4P (1.30 mS/cm) and the difference was presumably because the 3:1:1 substrate had more mixing of the influent and leachate.

Influent volume affected leachate volume (Fig. 1B). As expected, leachate volumes increased with increasing influent volumes (7). The high volume of influent satiated the substrate pores, once satiated the pore water was displaced by the remaining influent. Container size and substrate had no effect on leachate volume (data not presented).

In contrast, substrate type influenced the mixing of the influent and leachate (Fig. 2). Mixing occurred more readily in the 3:1:1 substrate due the lower container capacity and total porosity (Table 3). Since the 3:1:1 substrate was not able to hold as much substrate solution, the influent was more susceptible to leaching through the substrate.

Increasing influent volumes increased mixing of the influent and leachate (Fig. 1C). Similar to the results noted for leachate volume, the high volume of influent satiated the substrate pores, once satiated the pore water was displaced by the remaining influent, which eventually was leached. Container size did not affect the amount of mixing for the bedding plant cell packs.

Due to the small substrate volume in bedding plant cell packs, mixing of the influent and leachate readily occurs. To obtain representative substrate solution samples, only 15 mL on influent should be used. This small volume displacement

may warrant increased sampling to obtain representative values or the use of small volume pH and EC probes.

Experiment 2 -10.2 and 12.7 cm containers

Substrate type affected pH (LSD = 0.10), which was similar to that observed with the bedding plant cell packs such that the 3:1:1 pH (6.56) was higher than the Fafard 4P substrate pH (5.47). Container size and influent volumes did not affect the substrate pH for plants grown in the 10.2 and 12.7 cm containers (data not presented).

Electrical conductivity was affected by the substrate by influent volume interaction (Fig. 3). Substrate EC increased with increasing influent volumes and the 3:1:1 substrate had higher EC values than the Fafard 4P when 60 or 75 mL of influent was used for the PourThru extraction. The 3:1:1 substrate was a porous mix that had a lower container capacity compared to Fafard 4P (Table 3) and was more susceptible to mixing of the influent and leachate. Therefore, the high influent volumes (60 or 75 mL) resulted in mixing of the influent and PourThru leachate, which increased its EC due to the high solute (KSCN) content of the influent.

The EC values for the Fafard 4P were higher than the 3:1:1 substrate for plants provided 30 or 45 mL of influent and there was no difference between 45 or 60 mL of influent for the Fafard 4P (Fig. 3). When 30 or 45 mL of influent was added little mixing occurred. This indicates that the Fafard 4P actually had higher substrate EC values when the sample was unadulterated. Container size did not affect PourThru EC values (data not presented).

Container size affected leachate volumes such that the 10.2 cm containers produced higher volumes (42.9 mL) than the 12.7 cm containers (34.08 mL) (LSD = 1.86). The smaller 10.2 cm containers had less substrate volume, thus less pore space to hold the influent. Therefore, a higher volume of influent leached through the container.

Influent volume affected leachate volume (Fig. 4). The higher volumes of influent satiated the substrate pores, once satiated the pore water was displaced by the remaining influent versus substrate retention with the lower influent volumes; albeit, substrate did not affect leachate volumes.

Mixing of the influent and leachate was affected by the substrate by container by influent volume interaction (Table 4). Mixing was greatest with the 3:1:1 substrate in the 10.2 cm container and increasing influent volumes. As noted above, the 3:1:1 substrate has a lower container capacity and drains more readily than Fafard 4P. Mixing was affected by influent volume similarly to leachate volume. As influent volume increased, mixing increased. The smaller container (365 cm^3) held less substrate than the larger (750 cm^3); therefore, there was less pore volume to hold water.

To obtain representative substrate solution sample from 10.2 cm containers, 30 mL of influent (distilled water) should be used. Considerations must be made to the type of substrate being used as substrates similar to the 3:1:1 (less than or equal to 57.5 % container capacity by volume) would be more likely to yield adulterated PourThru leachates. For 12.7 cm containers, if a porous mix such as the 3:1:1 is used, then 30 mL of influent is recommended. If a mix such as Fafard 4P (greater

than or equal to 69.8% container capacity by volume) is used, then 30 to 45 mL of influent is acceptable.

Conclusion

Crop time influenced the PourThru results such that EC fluctuated among sampling dates. However, no parameters measured in this study were conclusive as to the cause and results were presumably due to natural variation in substrate moisture content and plant nutrient uptake as the plants grew. Influent volume caused differences in PourThru EC, leachate volumes, and mixing. The effect on EC and mixing warrant the recommendation of using distilled water to lessen the impact on these values if mixing of the influent and leachate occurs (1). The use of PourThru on small containers was more susceptible to adulterated samples versus large containers. The small containers have less substrate volume to serve as a buffer and prevent mixing during the displacement process. The substrate used was also influential. The greater porosity of the 3:1:1 mix made it more susceptible to leaching versus the Fafard 4P. To avoid mixing, greater care should be used when collecting substrate samples via the PourThru method in substrate with physical properties similar to the 3:1:1.

To obtain representative substrate solution samples in bedding plant cell packs, only 15 mL of influent should be used. When 10.2 or 12.7 cm containers are used with a typical floriculture substrate, 30 mL is recommended for the 10.2 cm containers and 30 to 45 mL is recommended for the 12.7 cm container. For larger

containers such as the 15.2 cm container, 50 to 75 mL of was acceptable to obtain an unadulterated substrate solution samples.

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Table 1. Main-effect of crop time on PourThru electrical conductivity and leachate volume for poinsettias grown in 15.2 cm containers. Means are an average of 24 samples per treatment. Expt. 1.

^z Crop time (weeks)	Electrical conductivity (mS/cm)	Leachate volume (mL)
2	2.43	69.0
4	2.51	60.8
6	2.29	70.0
8	3.31	56.7
Significance ($P > F$)	0.0001	0.0001
LSD _{0.05}	0.33	4.05

^z 0 representing transplanting of 2 week old seedling.

Table 2. Main-effect of influent volume on PourThru leachate volume and mixing for poinsettias grown in 15.2 cm containers. Means are an average of 24 samples per treatment. Expt. 1.

Influent	Leachate	
volume (mL)	volume (mL)	^z Mixing (%)
50	37.5	4.2
75	52.7	4.2
100	73.5	33.8
125	92.0	58.3
Significance ($P > F$)	0.0001	0.0001
LSD _{0.05}	4.04	23.5

^z Data analysis based upon arc-sin transformation.

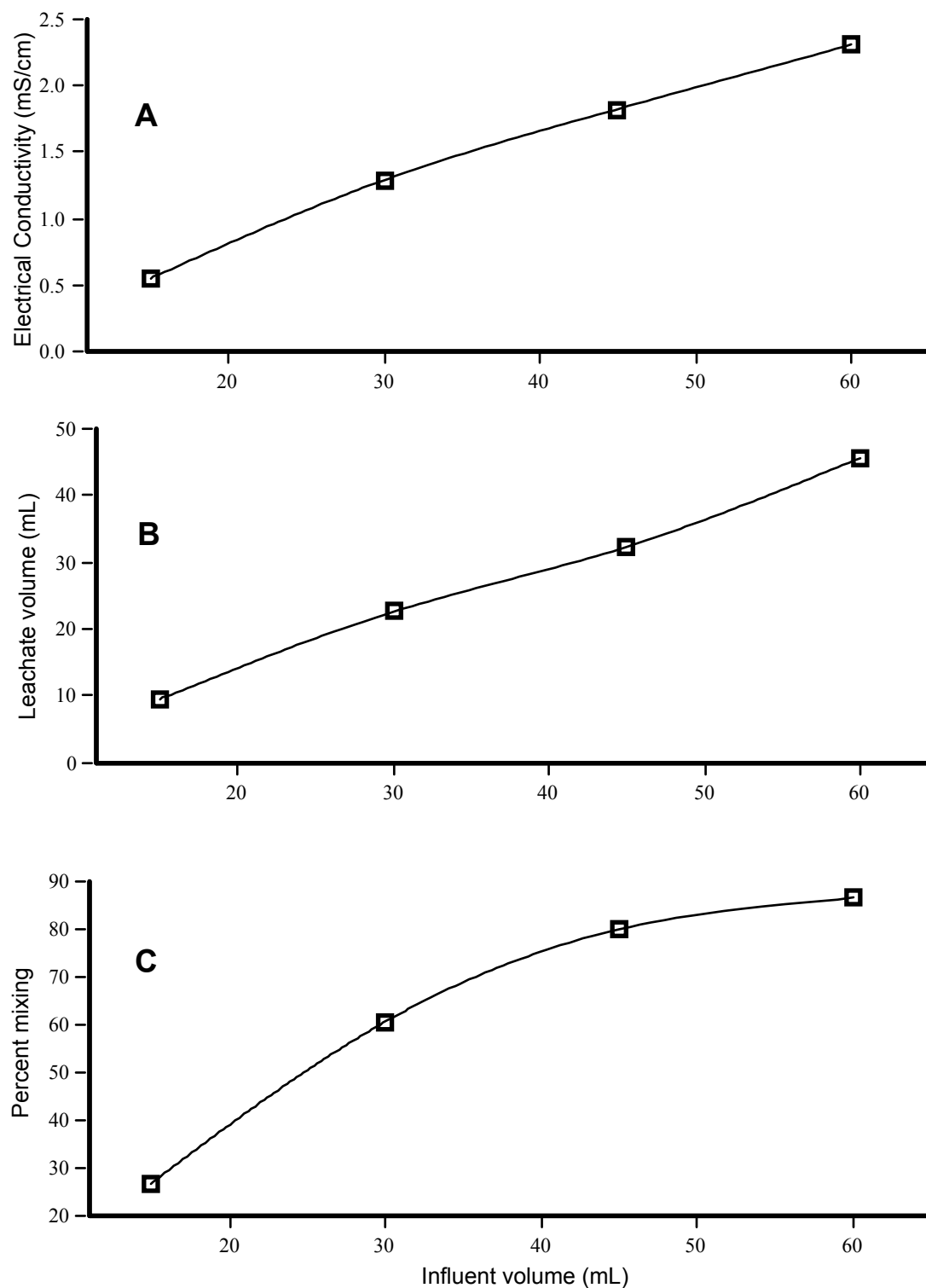


Fig. 1. Influent volume effect on (A) electrical conductivity (LSD = 0.36), (B) leachate volume (LSD = 3.07), and (C) mixing of influent and leachate (LSD = 16.8) for 1203 and 1801 cell packs. Data points represent a mean of 60 samples. Expt. 2.

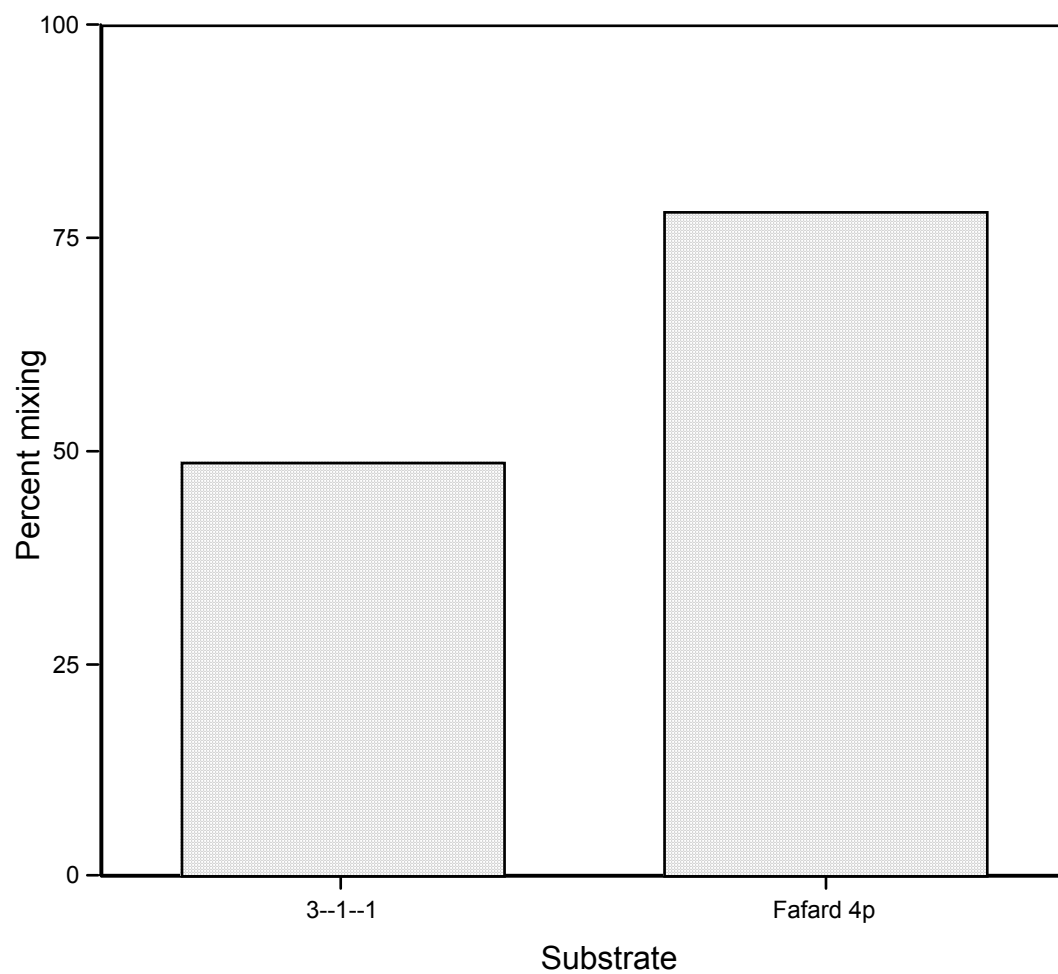


Fig. 2. Substrate type effect on mixing of influent and PourThru leachate for 1203 and 1801 cell packs ($LSD_{0.05} = 11.9\%$, $n = 120$). Expt. 2.

Table 3. Physical properties of substrates used for PourThru testing (n = 3).

Expt. 2.

Substrate	^z Total	^z Container	^x Bulk
	Porosity (%)	Capacity (%)	density (g/cm ³)
^w 3:1:1	66.4	57.5	0.53
Fafard 4P	85.0	69.8	0.13

^z Based on grams of water per 347.5 cm³ core.^x Based on gram of dry substrate per 347.5 cm³ core.^w Ratio of bark:peat:sand (v:v:v).

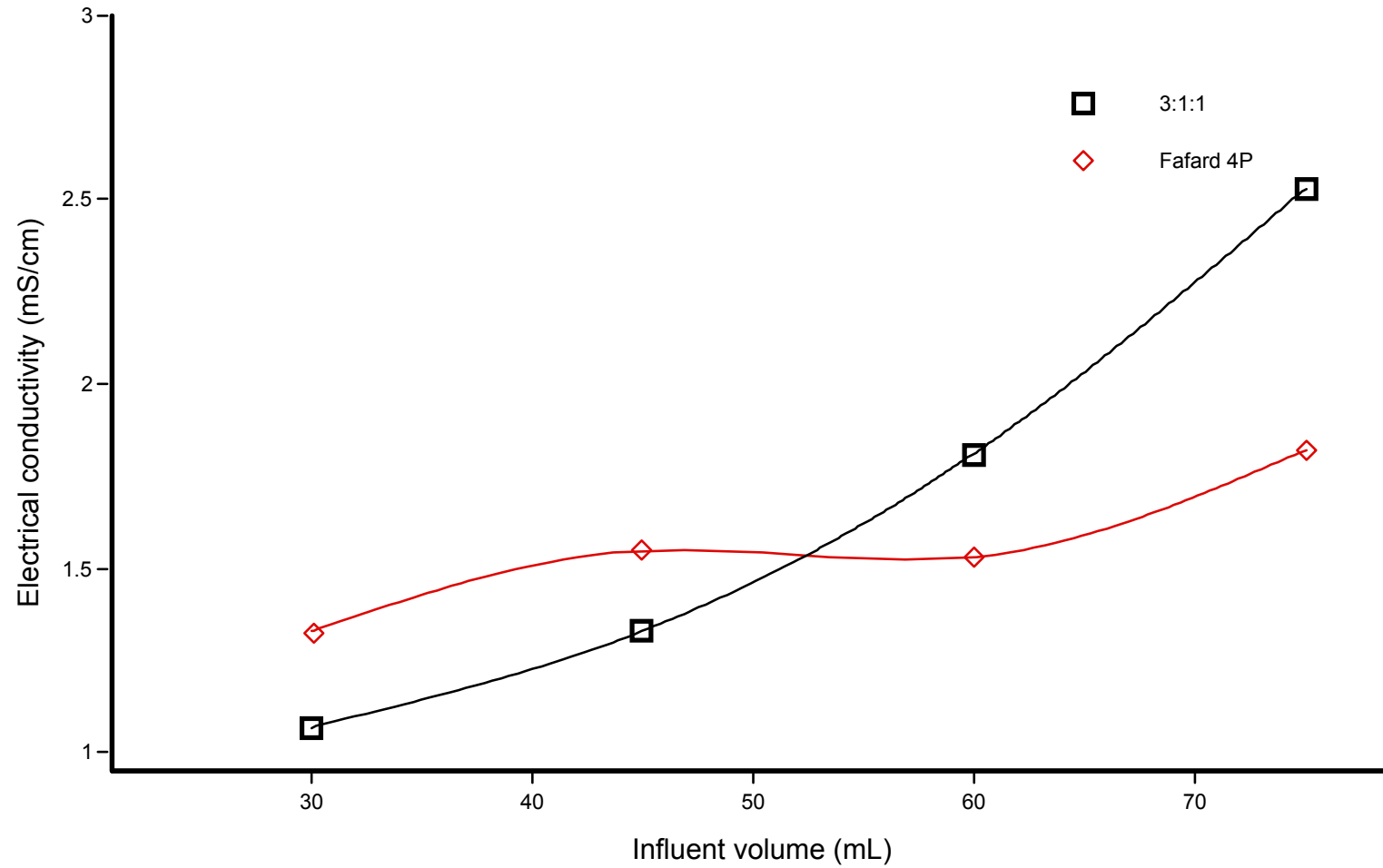


Fig. 3. Interaction of substrate and influent volume effect on electrical conductivity for 10.2 and 12.7 cm containers ($LSD_{0.05} = 0.27$, $n = 30$). Expt. 2.

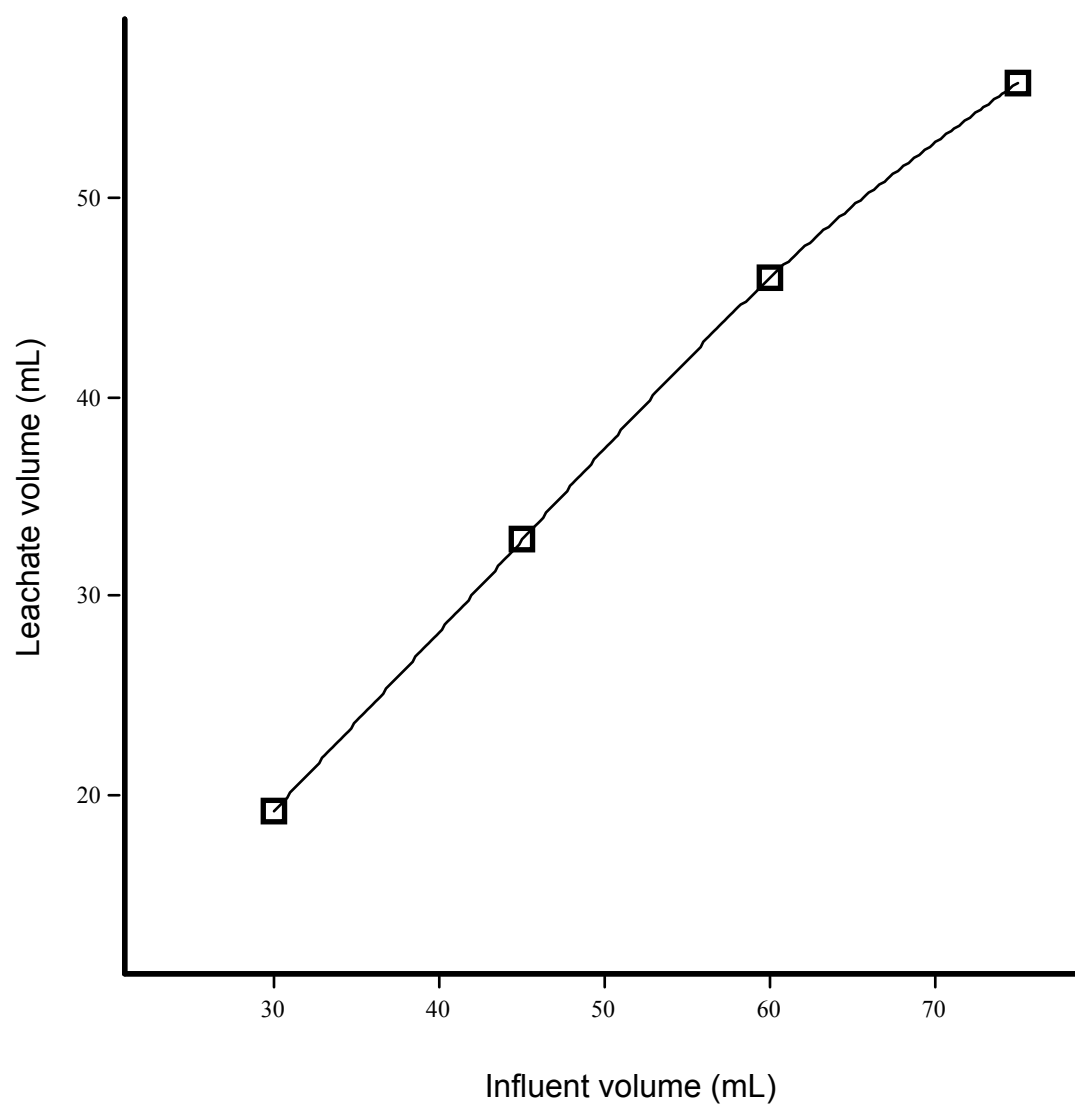


Fig. 4. Influent volume effect on PourThru leachate volume for 10.2 and 12.7 cm containers ($LSD_{0.05} = 3.5$, $n = 60$).

Table 4. Substrate, container size, and influent volume affect mixing of PourThru substrate samples. Means are an average of 15 samples per treatment. Expt. 2.

		Influent	
Substrate	Container	volume (mL)	^z Mixing (%)
^y 3-1-1	^x 10.2 cm	30	46.7
		45	73.3
		60	100.0
		75	100.0
	^w 12.7 cm	30	38.5
		45	86.7
		60	100.0
		75	100.0
Fafard 4P	10.2 cm	30	0.0
		45	13.3
		60	66.7
		75	100.0
	12.7 cm	30	14.3
		45	13.3
		60	13.3
		75	53.3
Significant interaction			
<i>Substrate * Container * influent volume (P > F)</i>			0.0105

LSD_{0.05}

38.7

^z Data analyses was based upon arc-sin transformation.

^y Substrate is 3 bark: 1 peat: 1 sand (v/v/v).

^x A 365 cm³ pot.

^w A 750 cm³ pot.

Chapter 6.

Evaluation of a Small – probe Time Domain Reflectometer in Small Volume Containers

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Evaluation of a Small – probe Time Domain Reflectometer in Small Volume Containers

Additional index words: TDR, volumetric moisture content, soil, peatmoss, bulk density, material specific calibration.

Abstract. The TRIME-FM TDR was evaluated with two probes [50 mm (1.97 in) and 110 mm (4.33 in) long] and various container sizes as well as soil moisture contents and bulk densities and compared to soil moisture contents obtained gravimetrically. Additionally, the TRIME-FM TDR was evaluated for use in peatmoss and material specific calibration. Probe size affected TDR measurements and the smallest probe [50 mm (1.97 in) length] provided values closest to those obtained gravimetrically. However, container size did not affect water content as measured by the probes in our study. Moisture status of the soil influenced the accuracy of the TDR readings such that values obtained in low moisture contents (10.3% by volume) were underestimated. Bulk density (ρ_b) greatly influenced the measured water contents, samples with ρ_b values lower than $1.2 \text{ g}\cdot\text{cm}^{-3}$ ($9.85 \text{ lb}\cdot\text{gal}^{-1}$) were drastically underestimated. Material specific calibration improved TDR values obtained in peatmoss from underestimating values approximately 60% to 4%. Time domain reflectometry may be useful to horticulturists if material specific calibrations are performed.

Introduction

Soil scientists have used time domain reflectometry (TDR) technology for many years to measure moisture content of field soils (Topp et al., 1980). However, limited information on TDR use is available for horticulture applications dealing with small containers (Anisko et al., 1994; Richardson et al., 1992). Time domain reflectometry works by using metal rods (probes) as wave-guides for the transmission of the TDR signal (electromagnetic wave). A high-frequency-pulse is generated, which propagates along the wave-guides generating an electromagnetic field around the TDR-probe. At the end of the wave-guides, the pulse is reflected back to its source. The resulting wave velocity or transit time is dependent on the dielectric constant, which is a function of water content, the sorbed matrix, and soluble salt content of the material (Fellner-Feldegg, 1969).

Time domain reflectometry could offer many advantages over conventional moisture analysis in horticultural substrates. Conventional analysis for gravimetric moisture content requires a destructive harvest of the substrate and extensive drying times so that substrate dry weights can be compared to wet weights. The instantaneous, non-destructive results associated with TDR moisture analysis would be advantageous. Other advantages include portability and data logger capabilities. Recent advances in TDR technology have yielded small, portable, and inexpensive units. These units show promise as becoming standard analytical tools for horticultural substrate analyses.

The TRIME-FM TDR (IMKO Micromodutechnik, GMBH, Ettlingen, Baden-Wurttemberg, Germany) operates based on the equation developed by Topp et al. (1980) and generates water content values in percent volumetric moisture content [$\text{cm}^3\text{cm}^{-3} * 100$ ($\text{in}^3\text{in}^{-3} * 100$)]. The TRIME-FM TDR is designed to be user-friendly; therefore, it is a self-contained unit with no need for a separate pulse generator and oscilloscope.

It has been noted that TDR deviates from gravimetric water content measures for organic substrates due to the large amount of bound H_2O and low bulk density (ρ_b) (Anisko et al., 1994; Dirksen and Dasberg, 1993; Pepin et al., 1992; and Stein and Kane, 1983). However, these discrepancies have been overcome by simple calibrations comparing the apparent wavelength to the wavelength ($L_a:L$) (Ledieu et al., 1986; Anisko et al., 1994) or by using the Maxwell – De Loor theoretical calibration equation (Dirksen and Dasberg, 1993).

Pepin et al. (1992), Roth et al. (1992), and Anisko et al. (1994) have noted that organic substrates, such as peatmoss, should be measured using an alternative calibration equation compared to those used for mineral soils (Topp et al., 1980), which is used in the TRIME-FM TDR. However, the TRIME-FM TDR allows for material specific calibration by use of computer software. This has been recommended for materials “significantly” greater or less than $1.4 \text{ g}\cdot\text{cm}^{-3}$ ($10.4 \text{ lb}\cdot\text{gal}^{-1}$). When densities are lower than $1.1 \text{ g}\cdot\text{cm}^{-3}$ ($8.3 \text{ lb}\cdot\text{gal}^{-1}$), the TRIME-FM TDR underestimates values by approximately 5% due to large air gaps (IMKO, personal communication). This material specific calibration may overcome anomalies associated with low-density materials such as peatmoss.

Previous studies on TDR in horticultural substrates have dealt primarily with larger containers than traditionally used in floriculture production (Anisko et al., 1994; da Silva et al., 1998; Murray et al., 2000; and Richardson et al., 1992). In addition, these studies also used TDR with 150 mm (5.9 in) or longer probes. The TRIME-FM is compatible with small probes [50 mm (1.97 in) long and 14 mm (0.55 in) apart] and probe size may be important dependent upon container size. Topp et al. (1982) noted that the sampling volume of the TDR probe is approximately the square of the spacing between the metal rods on the probe. Therefore, container size might be influential dependent upon the size of TDR probe being used.

The objective of our study was to determine if TDR was applicable to floriculture production using small probes and containers with a commercially available TRIME-FM TDR that has interchangeable probes and can be material specific calibrated. Probe size, container size, substrate moisture content, and substrate bulk density were evaluated to determine the effect on the measured moisture content as well as a comparison of material calibrated values of the TRIME-FM TDR.

Materials and Methods

Experiment 1

Soil [sandy loam (74% sand, 22% silt, 4% clay)] was sieved through a 6.4 mm (0.25 in) sieve and placed into 10.2 x 12 cm (4 x 4.7 in) [972 cm³ (0.25 gal)] or 15.2 x 12 cm (6 x 4.7 in) [2188 cm³ (0.58 gal)] poly vinyl chloride (PVC) cylinders that were covered by plastic window screen material on one end. Two TDR probes measuring 50 mm (1.97 in) long and spaced 14 mm (0.55 in) apart (P2D) or 110 mm (4.33 in) long and 20 mm (0.79 in) apart (P2) were used to measure percent volumetric moisture from the TRIME-FM TDR. The soil and cylinders were then weighed and placed in a drying oven [105 °C (221 °F)] for 48 h. Data collected included wet weight and dry weight to calculate volumetric moisture content [(wet weight - dry weight)/volume*100] and volumetric moisture content determined by the TDR probe. This study was a completely randomized design with a 2 x 2 factorial treatment combination with two probe sizes and two container volumes with 5 replications per treatment. Data were subjected to analysis by PROC GLM and means separation by protected LSD.

Experiment 2

Soil [sandy loam (74% sand, 22% silt, 4% clay)] was wetted and allowed to equilibrate for 24 h so that three relative moisture contents (ambient, moderately moist, and very moist) could be attained and placed into 10.2 x 10 cm (4 x 3.9 in) PVC cylinders [811 cm³ (0.21 gal)]. The TRIME-FM TDR probe (P2D) was used to evaluate the accuracy over the three moisture levels. Data collected included TDR

volumetric moisture content, soil wet weight and dry weight to obtain volumetric moisture content $[(\text{wet weight} - \text{dry weight})/\text{volume} \times 100]$. This study was a completely randomized design with three moisture levels and six replications per treatment. Data were subjected to analysis by PROC ANOVA and means separation by protected LSD.

Experiment 3

Peatmoss and sand were combined to obtain mixes with bulk densities (ρ_b) of 1.2, 0.96, 0.64, 0.39, and 0.04 $\text{g} \cdot \text{cm}^{-3}$ (9.85, 7.95, 5.30, 3.03, and 0.33 $\text{lb} \cdot \text{gal}^{-1}$, respectively) consisting of 20:80, 40:60, 60:40, 80:20, and 100:0, peat:sand (v:v) mixtures, respectively. The mixes were then placed into 10.2 x 12 cm (4 x 4.7 in) PVC cylinders [972 cm^3 (0.26 gal)]. The TDR probe (P2D) was used to evaluate each cylinder for volumetric moisture content and then cylinders were placed in a drying oven for 48 h at 105 °C (221 °F). Data collected included TDR volumetric moisture content and gravimetric moisture content converted to volumetric moisture content $[(\text{wet weight} - \text{dry weight})/\text{volume} \times 100]$. Percent deviation of measurement was reported as $\text{TDR measurement}/\text{gravimetric measurement} \times 100$. This study was a completely randomized design with five ρ_b levels. Data were subjected to analysis by PROC ANOVA and means separation by protected LSD.

Experiment 4

Peatmoss was wetted to obtain five moisture levels. The peatmoss was then placed into 10.2 x 12 cm (4 x 4.7 in) PVC cylinders [9722 cm³ (0.26 gal)] and the TDR probe (P2D) was used to obtain volumetric moisture content [cm³·cm⁻³ *100 (in³·in⁻³*100)] with the factory installed “universal” calibration. The samples were then weighed and placed in a drying oven for 48 h at 105 °C (221 °F). Dry weights were then obtained and volumetric moisture content was determined for each cylinder [(wet weight - dry weight)/volume*100]. The TDR and gravimetrically obtained volumetric moisture content values were then entered into the SMCAL calibration software for the TRIME-FM TDR and the calibration process was performed (IMKO Micromodutechnik, GMBH). After calibration, peatmoss samples of five moisture contents were then placed into the PVC cylinders. The “material specific calibrated” TRIME-FM TDR probe was then used to evaluate the volumetric water content and the drying procedures were repeated to obtain the gravimetrically obtained volumetric moisture contents. This study was a completely randomized design and data collected included the initial TDR and gravimetric moisture values and the “calibrated” TDR and gravimetric moisture values. The calibrated values were then analyzed by PROC ANOVA and PROC REG (SAS Institute, Cary, NC).

Results and Discussion

Experiment 1

The probe size by moisture content determination method interaction affected substrate moisture values (Table 1). The longer, wider spaced TDR probe (P2) underestimated the volumetric moisture content ($12.8 \text{ g} \cdot \text{cm}^{-3}$) compared to the smaller probe (P2D) ($18.6 \text{ g} \cdot \text{cm}^{-3}$). Values obtained from the smaller probe were similar to the gravimetrically obtained values (17.68 and $17.84 \text{ g} \cdot \text{cm}^{-3}$). Therefore, the P2D probe was the better choice for the soil cores used in this study when values were compared to the standard gravimetric moisture contents. Pepin et al. (1991) noted that a 3 cm spaced TDR probe yielded more reliable results than a 5 cm spaced probe presumably due to the decreased influence of air gaps in low moisture (less than $80\% \text{ cm}^3 \cdot \text{cm}^{-3}$) soils.

Topp et al. (1982) and Topp and Davis (1981) noted that the area of measurement of the TDR probe was the square of the distance between the rods. While this may hold true for the probes used in their study, an alternative area of measure likely occurred for our small probes. According to Topp et al.'s (1982) calculation, both container sizes used in our study had large enough volumes to accommodate both probe sizes as the area of influence would have extended to approximately 2 or 4 cm (0.79 or 1.57 in, respectively) beyond the probes, which is well within the radius of our 10.16 and 15.24 cm (4 and 6 in, respectively) wide containers. Due to the small probe size and small container volume, alternative areas of influence may need to be established.

Core size did not affect the TDR measurements (data not presented). As mentioned above, the soil core volumes used in our study were presumably sufficient to negate any influence of soil volume. Although height of the container was not evaluated in this study, we conducted previous studies that indicated depths of 10 to 14 cm (3.94 to 5.51 in) did not affect measurements of the P2D probe (unpublished data). However, Richardson et al. (1992) noted that TDR measurements were affected by measurement depths in a 100 cm tall root box.

Experiment 2

Moisture status affected the accuracy of TDR measurements ($P < 0.001$, $LSD_{0.05} = 1.72$). When soil moisture contents were high (34.7 and 18.1% by volume), the P2D probe measured similar moisture content values (35.5 and 16.8%, respectively) to those obtained gravimetrically. However, when soil moisture content was low (10.3%) the P2D probe underestimated the soil moisture content by 6.4%.

The P2D and other TDR probes have the ability to measure soil moisture values as low as 0 with $\pm 1\%$ error (IMKO, personal communication; Topp et al., 1980). However, our findings do not agree and it may have been due to the unique algorithms used by the TRIME-FM to determine the voltage at which time the amplitude measurements of the reflected pulse were taken. The TRIME-FM differs from conventional TDR probes as it operates at a higher frequency, has PVC coated metal rods, and measurements are taken at a specific voltage rather than a specific time (IMKO, personal communication). The TDR results may have been further

skewed by the perched water table effect that commonly occurs in container versus field conditions (Spomer, 1975).

Experiment 3

Altering the ratio of peatmoss to sand, thus, changing the bulk density affected TDR accuracy (Fig. 1). Deviation of the TDR and gravimetric values increased with decreasing bulk densities. The operation manual supplied with the TRIME-FM TDR noted that when bulk densities of the material being measured deviated “significantly” from $1.4 \text{ g}\cdot\text{cm}^{-3}$ ($10.4 \text{ lb}\cdot\text{gal}^{-1}$) that material specific calibrations should be performed; albeit “significant deviation” was not defined. Our findings are in agreement with the operator’s manual as well as Anisko et al. (1994), Dirksen and Dasberg (1993), Pepin et al. (1992), and Roth et al. (1992) that alternative calibration equations or material specific calibrations should be used.

Experiment 4

The material specific calibration (100% peatmoss) increased the accuracy of the TDR measurements. When the TDR and gravimetric values were evaluated over five moisture contents with the “universal” calibration, a linear relationship of $y = 0.60219x - 2.83024$, $r^2 = 0.98$ was established (Fig. 2). Therefore, TDR values were approximately 40% lower than values obtained gravimetrically. This underestimation is in agreement with the operator’s manual and Topp et al. (1984). However, these results differ from an observation by Pepin et al. (1992), who noted

TDR overestimated the water content for substrates with a ρ_b ranging from 0.06 to $0.25 \text{ g} \cdot \text{cm}^{-3}$ (0.49 to $2.08 \text{ lb} \cdot \text{gal}^{-1}$).

The linear relationship of the TDR to gravimetric values improved after material specific calibration (Fig. 2). The calibration yielded results that represent a 4% underestimation by the TDR versus gravimetric. While this was a vast improvement, the question of “how close is close enough” still remains. Topp et al. (1980) noted that TDR measurements in the field were within 1.3% of the gravimetric values and that the accuracy improved to 1% with material specific calibration. However, Topp et al. (1984) noted that slopes as low as 0.904 and correlation coefficients as low as 0.917 “indicate a strong functional relationship.” Considering this later statement, our material specific calibration produces acceptable results. However, one must consider that in small containers and with the moisture release characteristics of materials commonly used in floriculture that a small error may still have a significant impact.

Conclusion

Probe size (P2D: 50 mm long and 14 mm apart versus P2: 110 mm long and 20 mm apart) affected the TDR values obtained in our study and further research should be conducted to find the limitations of probe and container sizes. Moisture content affected the TDR accuracy; albeit, several researchers have reported that TDR was highly effective over wide moisture contents. These variations may be due to the perched water table effect that occurs in containers, which was accentuated by our small containers, versus many field situations.

Bulk density was critical for accurate measure of the TRIME-FM with the universal calibration. When ρ_b deviated from approximately $1.4 \text{ g}\cdot\text{cm}^{-3}$ ($10.4 \text{ lb}\cdot\text{gal}^{-1}$), the accuracy of the TDR decreased. However, material specific calibration greatly improved the TDR measurements to within 4%, which has been reported as an acceptable error (Topp et al., 1984). A concern with floriculture production is the changing ρ_b of the substrate over crop time. Further studies need to be conducted to determine if material specific calibrations would need to be made between the initial ρ_b and those that would be observed late in the crop.

Time domain reflectometry may have its future in horticulture substrates; however, several issues must first be resolved. The heterogeneity of floriculture mixes needs to be evaluated. Albeit several studies have been conducted using commercial mixes and reported acceptable results, the commercial mixes can be dramatically different and calibrations will presumably need to be developed for these varying substrate mix characteristics. This is supported by findings of da Silva et al. (1998) who noted perlite and organic media need different calibration equations. Further research is also needed to evaluate the reliability of small TDR probes and small containers. We observed favorable results for TDR use in horticulture production in this study, but have also observed inconsistencies in unpublished trials and feel that the results from TDR should closer emulate those obtained by gravimetric means.

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Table 1. Probe size effect on TRIME- FM TDR moisture content values. Means are an average of five replications per treatment (n = 40). Expt. 1.

TDR Probe	Method of moisture content determination	Moisture content z ($\text{cm}^3.\text{cm}^{-3} * 100$)
y P2D	TDR	18.63
	gravimetric	17.68
x P2	TDR	12.75
	gravimetric	17.84
Significance		
<i>Probe*method</i> ($P > F$)		0.0004
LSD _{0.05}		1.07

z in³.in⁻³*100

y probe has two 50 mm metal rods spaced 14 mm apart.

x probe has two 110 mm metal rods spaced 20 mm apart.

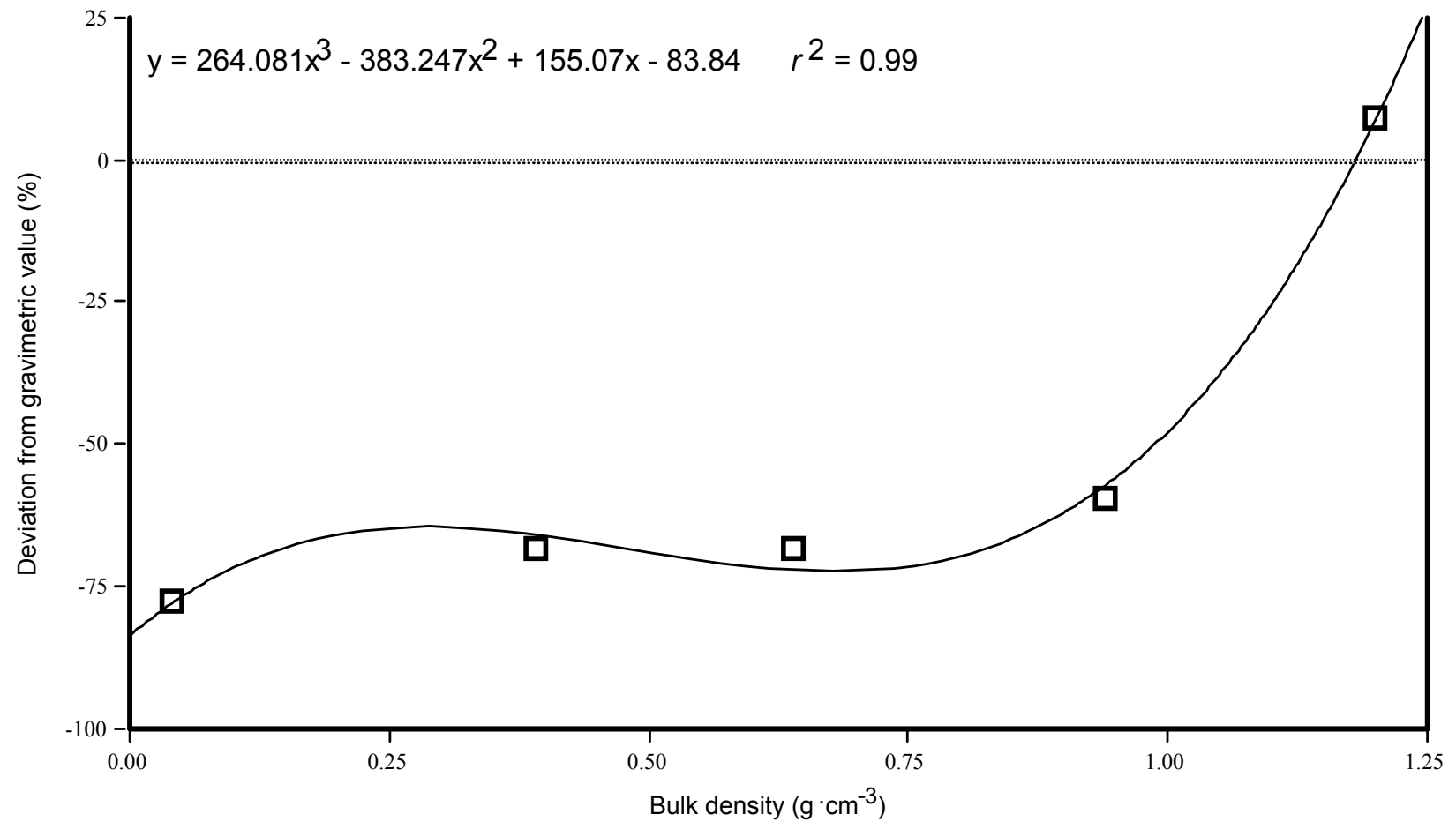


Fig. 1. Bulk density's effect on deviation of TRIME - FM TDR measurements from gravimetric measurements. Data points represent a mean of 5 replications per treatment ($n = 40$). Expt. 3.

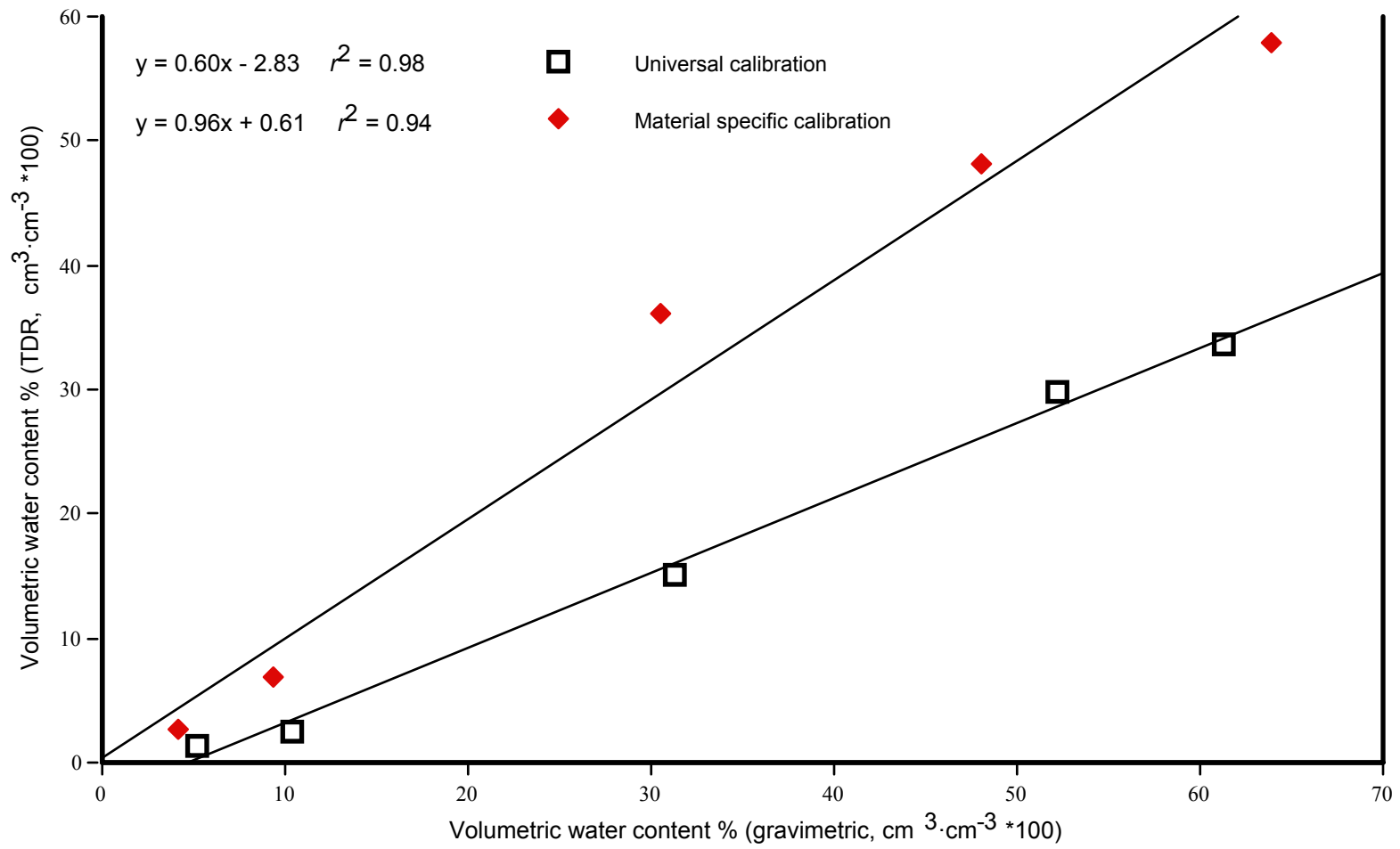


Fig. 2. Linear regression of gravimetric and TDR obtained volumetric moisture contents with "universal" and material specific calibration. Expt. 4.

Chapter 7.

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