Reprinted from the *Soil Science Society of America Journal*
Volume 59, no. 1, January–February 1995
677 South Segoe Rd., Madison, WI 53711 USA

CONSTRUCTION AND INSTALLATION OF ACRYLIC MINIRHIZOTRON TUBES IN FOREST ECOSYSTEMS

BRIAN D. KLOEPPEL* AND STITH T. GOWER

Abstract

Glass tubing has been the traditional material used when constructing minirhizotrons for studying fine root dynamics in managed and natural terrestrial ecosystems. However, we have found that rocky soils, swelling smectite clays, large structural roots, and freezing soils frequently have broken glass minirhizotrons. We have developed a system to construct and install durable acrylic minirhizotrons. Acrylic tubing costs 42% less than glass and requires only 13% of the preparation time, resulting in a 121% savings vs. glass. The major time-saving device is a unique spring-loaded etching trough that places accurate, evenly spaced circles along the length of the tube in minutes. The design can be easily modified for many applications. Preliminary results in a mixed western larch (*Larix occidentalis* Nutt.)-lodgepole pine (*Pinus contorta* var. *latifolia* Engelm.) forest in western Montana indicate that the acrylic minirhizotrons are durable, the etching is visible, and the system can be used to examine fine root phenology.

MINIRHIZOTRONS have become a common tool to observe growth, morphology, and dynamic changes in fine roots since Bates (1937) introduced them more than a half century ago (Upchurch and Ritchie, 1984; Lussenhop et al., 1991; Hendrick and Pregitzer, 1992). They allow frequent measurements of fine root growth and morphology without destructive root coring and laborious sorting procedures (cf. Santantonio and Hermann, 1985), making fine root sampling feasible for many types of studies (Böhm et al., 1977). To adequately characterize root growth, a large number of minirhizotron tubes are necessary, but minirhizotron tubes are expensive. Glass minirhizotrons are less expensive than commercially available polybutyrate tubes. However, the installation of glass minirhizotrons in many forest ecosystems can be difficult and result in fractured tubes if large roots and rocks are encountered in the soil profile. In addition, freeze–thaw action and the shrink–swell action of smectite clay soils can also cause tube breakage.

We present a system for fabricating minirhizotron tubes from inexpensive acrylic tubing, marking them with an easily constructed etcher, and installing them with a tree seedling auger. This system has enabled us to monitor fine root dynamics of a mixed western larch–lodgepole pine forest growing on a volcanic ash soil in western Montana.

Department of Forestry, Univ. of Wisconsin, 1630 Linden Drive, Madison, WI 53706. *Corresponding author (kloeppel@calshp.cals.wisc.edu).
Received 2 Feb. 1994.

Published in Soil Sci. Soc. Am. J. 59:241–243 (1995).

Materials and Methods

The mixed western larch–lodgepole pine research site is near Troy, MT (N 48°25′10″, W 115°48′30″). This forest stand was naturally regenerated after clear-felling in 1958 and is located on a deep (>1-m) silt loam volcanic ash soil that is classified as an Entic Eutrandept.

Because a large fraction of glass minirhizotrons broke over winter in a ponderosa pine (*Pinus ponderosa* Dougl. ex P. Lawson & Lawson) forest in a separate study in western Montana and because of the high cost of glass, we decided to examine the suitability of more durable materials. Acrylic is durable and relatively inexpensive; therefore, it is a logical choice (Waddington, 1971). Transparent acrylic lasts longer than glass because it can withstand greater soil pressure (McMichael and Taylor, 1987). Ready-made plastic minirhizotron tubes are available from J.R.D. Merrill Co., Logan, UT; however, the cost per 1.2-m tube is high ($22.50). Acrylic tubing (44-mm o.d., 38-mm i.d.) was readily available in 1-m lengths from a Madison, WI, plastics supplier.

The construction time of the minirhizotrons was also a consideration because glass tubes require a significant amount of time to mark circles around the tube with either permanent markers or acid pens. We developed an automated etcher to quickly and precisely mark acrylic minirhizotron tubes. The device consists of a bench-mounted trough equipped with 50 spring-loaded etchers located at 2-cm intervals in an acrylic and wooden frame (Fig. 1). The frame can be built to any length, but is best constructed of hard, rigid material that will not bend when the acrylic tubes are rotated in the trough. The spring-loaded etchers were fabricated from sharpened double-head 10d nails fitted with no. 2 springs and mounted between two sheets of acrylic drilled with the desired spacing (Fig. 1). The spring loading was necessary to ensure that all etchers were pressed equally against the tube, thereby accounting for anomalies in the acrylic tubing. Without the springs, the etcher would cause deep cuts in the acrylic tubes in some locations, but not etch at other locations vertically along the tube. All components of the etcher were fastened with steel bolts every 15 cm to minimize frame bending because the etchers are under pressure when the acrylic is rotated.

After the tubes were etched, a no. 9 rubber stopper was placed in one end and secured with vinyl electrical tape. To make sure that the minirhizotron was water tight, we applied a silicon caulking around the stopper and tube. To increase the visibility of the etched lines under both high and low soil moisture conditions, we applied black, non-water-soluble ink to the etchings on one-half of the tube along the vertical axis with a small sponge and wiped off the excess with a dry towel. Finally, we labeled the etched tubes every 10 cm with a permanent marker; this enabled the user to quickly reference the depth of the roots.

Because forested ecosystems are relatively inaccessible to tractor- and truck-mounted augers for drilling holes to install minirhizotrons (cf. Brown and Upchurch, 1987), we were limited to portable systems. We used a chain-saw-mounted Cannon DH-2WM tree planter (Cannon Machine Works, Ltd., Burnaby, BC, Canada) equipped with a 5 by 90 cm carbide-tipped auger. The auger easily drilled through compacted soil, small tree roots, and small rocks and could be held at a 45° angle, which is required to minimize root growth along the length of the tube (Sanders and Brown, 1978; Upchurch and

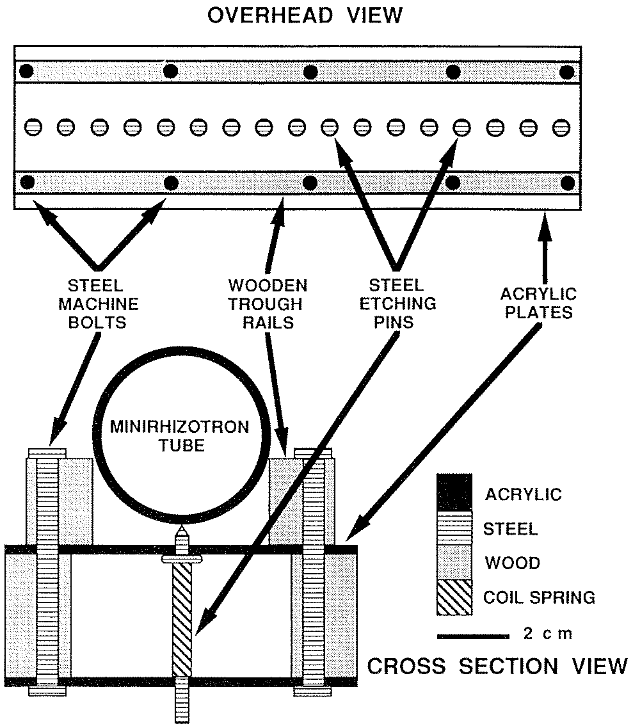

Fig. 1. Overhead and cross-section views of the acrylic etcher used to mark minirhizotron tubes at 2-cm intervals. The spring-loaded etcher corrects for inconsistent pressure when etching and anomalies in acrylic tubes. The etching pins are sharpened 10d double-head nails. Spring tension is generated from a no. 2 spring fit over each etching pin. The pins are held between two sheets of acrylic, 3.2 mm thick. The acrylic sheets are separated by two wooden rails (19 mm thick by 35 mm wide) along with two additional rails (19 mm thick by 25 mm wide) that guide the minirhizotron tube. The acrylic sheets and wooden rails are fastened together with machine bolts (6.4-mm diam. by 70 mm long).

Ritchie, 1983). After each minirhizotron was placed in the hole, the hole was back-filled with sieved soil from the same hole. A small wooden dowel was used to gently pack soil around the tube to minimize air pockets. The top of the minirhizotron above the soil surface was spray painted with a coat of black to prevent light penetration into the tube and then with a coat of white to minimize solar radiative heating.

We installed 80 acrylic minirhizotron tubes at the western Montana research site on 16 May 1992 and monitored root growth along the length of the minirhizotrons during the remainder of the growing season. Observations were made with a portable, battery-powered root periscope (J.R.D. Merrill Specialty Company, Logan, UT). A line-intercept method was used to estimate root elongation (Newman, 1966). Root intercepts on the evenly spaced circles were monitored along a 1-cm-wide strip on the upper surface of the minirhizotron. Root intercepts with each horizontal etch mark on the tube were tallied along with root diameter class (<0.5, 0.5–1.0, 1.0–1.5, and 1.5–2 mm) and color.

In October 1992, we made a complete baseline reading for all tubes for the upcoming growing season because it appeared that all root growth had ceased for the year. We verified this by again reading all tubes in March 1993 and observed no changes in the number or location of fine ($\leq$2-mm) diameter roots. Though we observed a slightly darker color on many of the new root tips that grew in 1992, no root mortality was observed during the first winter of installation in 1992–1993.

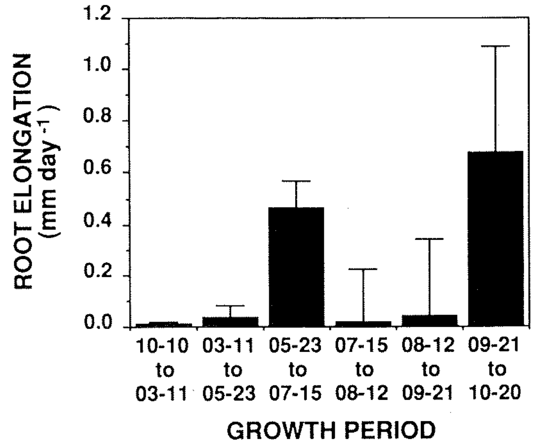

Fig. 2. Root elongation per day (mean ± standard error) in a mixed western larch–lodgepole pine forest in western Montana. The growth periods (mm-dd to mm-dd) begin in October 1992 and follow continuously through October 1993 with 28- to 53-d increments between growing season sampling dates.

Observations were then made approximately every 6 wk from March to October 1993.

Results and Discussion

Tube clarity was high throughout the 1992 and 1993 growing seasons despite large diurnal and seasonal temperature changes that typically cause condensation on the tubes. The dark ink that was placed on one-half of each etched circle did not significantly increase its visibility. Nonetheless, the contrast between the etched circles and the homogeneous light brown color of the ash soil was high throughout all moisture contents during the 12 mo of observations. The labels that indicated every 10 cm of depth along the tube were persistent throughout the 17 mo that the minirhizotrons were installed.

Results from six growth periods show the dramatic difference in root length during the year of observations (Fig. 2). Fine root elongation rates varied, with the greatest rates occurring during the early summer (23 May to 15 July) and fall (21 September to 20 October) while fine root growth was small during the summer. The periods of greater fine root growth coincided with high soil moisture and environmental conditions favorable for photosynthesis (Kloeppel et al., 1995). These results corroborate those from an earlier study that examined ectomycorrhizal growth for a Douglas fir–western larch forest in western Montana (Harvey et al., 1978), suggesting that the acrylic minirhizotron and line-intercept recording system can be used to characterize fine root growth (Waddington, 1971; Buckland et al., 1993).

In March 1993, we shoveled approximately 40 cm of snow away from each minirhizotron and found no broken tubes even though there was commonly 3 cm of ice around the minirhizotrons at the soil surface. In May 1994, 2 yr after installation, we still observed no damaged tubes. Furthermore, no tubes contained water, indicating that seepage through the lower rubber stopper was not

Table 1. Comparison of the materials cost, construction time, total cost, and relative durability of glass vs. acrylic minirhizotron tubes. Acrylic tubes that are ready to install or etched with the system illustrated in Fig. 1 are compared and the cost and preparation time to build the etcher system illustrated in Fig. 1 are also shown.

Material	Material cost†	Construction time	Total cost‡	Relative durability
	$/m	min/tube	$/tube	
Acrylic, etched	3.37	5	3.83	high
Glass	4.78	40	8.45	low
Acrylic, ready to install	18.75	0	18.75	high
Acrylic etching trough§	22.00	300	49.50	–

† Discount price when ordering 100 1-m-long tubes.
‡ Costs based on a manual labor rate of $5.50/h.
§ Costs and construction time are a one-time investment for acrylic etching trough.

a problem. These results are in contrast to other experiments using glass minirhizotron tubes. In a red pine (*Pinus resinosa* Aiton) forest on a coarse sand in northern Wisconsin, 3% of the tubes were broken after one winter (B.E. Haynes and S.T. Gower, 1991, unpublished data). In a ponderosa pine forest on a loam soil with 40% coarse fragments near Missoula, MT, 15% of the tubes were broken after one winter (E.R. Hunt, 1991, unpublished data). Finally, in a mixed oak (*Quercus* spp.) forest on a clay loam soil in southwestern Wisconsin, 90% of the tubes were broken after one winter (E.L. Kruger, 1993, unpublished data). Judging from the durability of the acrylic tubes 2 yr after installation, they may be reusable after the current 3-yr project is complete, thereby resulting in further financial savings.

An analysis of the construction costs of the acrylic minirhizotrons from this study and glass minirhizotrons from previous studies indicated that the glass tubes were 121% more expensive than the acrylic when materials and construction costs were considered (Table 1). Ready-to-install acrylic minirhizotrons are commercially available, but cost almost five times more than the total cost of etched tubes. The automated etcher significantly reduced the time required to prepare each tube, thereby contributing to the large cost difference between materials. Overall, the significantly lower price and greater durability of acrylic indicates that it is better than glass for use in minirhizotrons installed in forest soils.

Acknowledgments

We acknowledge financial support from National Science Foundation Grant BSR-9107419. We thank Susan Wiegrefe, who assisted with development of the acrylic tube etcher, and George Erickson, who assisted with field measurement of root intercepts.

References

Bates, G.H. 1937. A device for the observation of root growth in the soil. Nature (London) 139:966–967.

Böhm, W., H. Maduakor, and H.M. Taylor. 1977. Comparison of five methods for characterizing soybean rooting density and development. Agron. J. 69:415–419.

Brown, D.A., and D.R. Upchurch. 1987. Minirhizotrons: A summary of methods and instruments in current use. p. 15–30. *In* H.M. Taylor (ed.) Minirhizotron observation tubes: Methods and applications for measuring rhizosphere dynamics. ASA Spec. Publ. 50. ASA, CSSA, and SSSA, Madison, WI.

Buckland, S.T., C.D. Campbell, L.A. Mackie-Dawson, G.W. Horgan, and E.I. Duff. 1993. A method for counting roots observed in minirhizotrons and their theoretical conversion to root length density. Plant Soil 153:1–9.

Harvey, A.E., M.F. Jurgensen, and M.J. Larsen. 1978. Seasonal distribution of ectomycorrhizae in a mature Douglas-fir/larch forest soil in western Montana. For. Sci. 24:203–208.

Hendrick, R.L., and K.S. Pregitzer. 1992. The demography of fine roots in a northern hardwood forest. Ecology 73:1094–1104.

Kloeppel, B.D., S.T. Gower, and P.B. Reich. 1995. Net photosynthesis of western larch and sympatric evergreen conifers along a precipitation gradient in western Montana. *In* Proc. Int. Symp. Ecol. Manage. *Larix* forests: A look ahead, Whitefish, MT. 5–9 Oct. 1992. U.S. For. Serv. Gen. Tech. Rep. INT-000. Intermountain Res. Stn., Ogden, UT (in press).

Lussenhop, J., R. Fogel, and K. Pregitzer. 1991. A new dawn for soil biology: Video analysis of root–soil–microbial–faunal interactions. Agric. Ecosyst. Environ. 34:235–249.

McMichael, B.L., and H.M. Taylor. 1987. Applications and limitations of rhizotrons and minirhizotrons. p. 1–13. *In* H.M. Taylor (ed.) Minirhizotron observation tubes: Methods and applications for measuring rhizosphere dynamics. ASA Spec. Publ. 50. ASA, CSSA, and SSSA, Madison, WI.

Newman, E.I. 1966. A method of estimating the total length of root in a sample. J. Appl. Ecol. 3:139–145.

Sanders, J.L., and D.A. Brown. 1978. A new fiber optic technique for measuring root growth of soybeans under field conditions. Agron. J. 70:1073–1076.

Santantonio, D., and R.K. Hermann. 1985. Standing crop, production, and turnover of fine roots on dry, moderate, and wet sites of mature Douglas-fir in western Oregon. Ann. Sci. For. 42:113–142.

Upchurch, D.R., and J.T. Ritchie. 1983. Root observations using a video recording system in minirhizotrons. Agron. J. 75:1009–1015.

Upchurch, D.R., and J.T. Ritchie. 1984. Battery-operated color video camera for root observations in mini-rhizotrons. Agron. J. 76:1015–1017.

Waddington, J. 1971. Observation of plant roots in situ. Can. J. Bot. 49:1850–1852.