HortScience 52(9):1307–1312. 2017. doi: 10.21273/HORTSCI11903-17

Optimal Rate of Organic Fertilizer during the Vegetative-stage for Cannabis Grown in Two Coir-based Substrates

Deron Caplan, Mike Dixon, and Youbin Zheng[1]

School of Environmental Sciences, University of Guelph, Guelph, ON N1G 2W1, Canada

Additional index words. Cannabis sativa, cannabis growth, floral dry weight, marijuana, THC, CBN

Abstract. **Cannabis producers, especially those with organic operations, lack reliable information on the fertilization requirements for their crops. To determine the optimal organic fertilizer rate for vegetative-stage cannabis (*Cannabis sativa* L.), five rates that supplied 117, 234, 351, 468, and 585 mg N/L of a liquid organic fertilizer (4.0N–1.3P–1.7K) were applied to container-grown plants with one of two coir-based organic substrates. The trial was conducted in a walk-in growth chamber and the two substrates used were ABcann UNIMIX 1-HP with lower water-holding capacity (WHC) and ABcann UNIMIX 1 with higher WHC. No differences in growth or floral dry weight (yield) were found between the two substrates. Pooled data from both substrates showed that the highest yield was achieved at a rate that supplied 389 mg N/L (interpolated from yield-fertilizer responses) which was 1.8 times higher than that of the lowest fertilizer rate. The concentration of Δ^9-tetrahydrocannabinol (THC) in dry floral material was maximized at a rate that supplied 418 mg N/L, and no fertilizer rate effects were observed on Δ^9-tetrahydrocannabidiolic acid (THCA) or cannabinol (CBN). The highest yield, cannabinoid content, and plant growth were achieved around an organic fertilizer rate that supplied 389 mg N/L during the vegetative growth stage when using the two coir-based organic substrates.**

Cannabis (*Cannabis sativa* L.) legislation in North America continues to move rapidly toward liberalization and in some instances legalization, shifting cultivation from a largely illicit practice to one that is not only legal, but in high demand. In the United States, with only a handful of states having legalized recreational cannabis as of 2017, the market for legal cannabis was estimated at $2.7 billion USD in 2014, and it is expected to reach $11 billion by 2019 (ArcView Market Research and New Frontier, 2014). The current Canadian government has pledged to follow suit and pass legislation to legalize cannabis for recreational purposes beginning in spring of 2017. Until then, current legislation allows a limited number of private, licensed facilities to produce and distribute cannabis for medicinal purposes as well as conduct scientific research (Canada Gazette, 2016).

Cannabis is an annual dioecious species, producing separate male and female plants. Archeological evidence of cultivation dates to 10,000 BCE in China where cannabis was used primarily for fiber. Later, the medicinal use of cannabis became widespread, with evidence of cultivation and use in ancient Egypt around 2800 BCE and in China around 2000 BCE (Russo, 2007). The medicinal value of cannabis is attributed primarily to a group of secondary metabolites called cannabinoids which are concentrated mostly in the essential oils of unfertilized female flowers (Potter, 2014).

More than 100 unique cannabinoids have been identified (Ahmed et al., 2008, 2015; ElSohly and Slade, 2005), although Δ^9-THC and cannabidiol (CBD) are considered the primary psychoactive and medicinal components (Elzinga et al., 2015; Mechoulam et al., 1970). In live plants, cannabinoids exist primarily as carboxylic acids such as Δ^9-THCA and cannabidiolic acid (CBDA) (Muntendam et al., 2012). These acids undergo decarboxylation during storage (Ross and ElSohly, 1997; Taschwer and Schmid, 2015) and upon heating (Kimura and Okamoto, 1970) to become neutral cannabinoids such as THC and CBD. Varieties of cannabis with low THC and high CBD are termed hemp or fiber-type cannabis, whereas those with high THC and low CBD are termed marijuana or drug-type cannabis, hereafter referred to as cannabis (van Bakel et al., 2011; Vollner et al., 1986). Selective breeding has produced hundreds of varieties of cannabis with varying chemical compositions and growth characteristics (Vollner et al., 1986). Selection has mostly been for high floral THC concentration, but the medicinal effects of CBD have recently been identified (Russo, 2011) leading some breeders to select for high CBD. Most indoor production of cannabis occurs in two growth stages, vegetative and flowering, which are controlled by photoperiod (Farag and Kayser, 2015). Modern day cultivation of cannabis takes place almost exclusively indoors under artificial lighting using either solution culture systems or soilless growing substrates (Leggett, 2006; Potter, 2014). In addition, many cannabis growers favor organic production practices because consumers and regulating bodies often demand pesticide-free cannabis (Canada Gazette, 2016).

Online horticultural resources are available for cannabis production; however, limited information is available in peer-reviewed scientific literature. Furthermore, there is scant published scientific research on any aspect of organic cannabis production. Because of a lack of systematic horticultural research, current cannabis producers rely on cultivation methods derived largely from anecdotal information. Information on fiber-type cannabis cultivation techniques allows for some parallels to be drawn; however, fiber-type cannabis is field-grown and has been selectively bred for fiber production rather than for essential oil content (Amaducci et al., 2015). A chemotaxonomic study found low gene flow between drug- and fiber-type cannabis (Hillig and Mahlberg, 2004) and was supported by a recent genomic study comparing fiber and drug-type cannabis (van Bakel et al., 2011). This makes it difficult to relate cultivation techniques between the two crops (Amaducci et al., 2015).

Fertilization is one of the most important factors for indoor organic cannabis production. For fiber-type cannabis, the suggested fertilization rate is around 50–200 kg N/ha (Aubin et al., 2015; Ehrensing, 1998; Vera et al., 2004), which is similar to other high-yielding field crops such as wheat (*Triticum* spp.; Baxter and Scheifele, 2008). It is difficult, however, to estimate fertilizer requirements of drug-type cannabis based on fiber-type cannabis or other crops because of the differences in species and growing conditions (Wright and Niemiera, 1987). Furthermore, it is well-known that different growth stages of the same species have varying nutrient demand; when the demand is met, plant performance is improved (Raviv and Lieth, 2007; Wang, 2000). Most studies on fertilizer application in other crops have been conducted using conventional fertilizers, and there are few on the use of organic fertilizers for container crops. Fertigation rates of 190–400 mg N/L have been reported for container production of organic greenhouse-grown tomatoes (*Solanum lycopersicum* L.; Surrage et al., 2010; Zhai et al., 2009). To our knowledge, neither organic nor conventional fertilizer application rates have been published for indoor cannabis production in scientific literature.

Appropriate choice of a growing substrate is essential for soilless crop production because it directly affects root zone water, air,

We thank ABcann Medicinals Inc. for providing funding as well as materials, expertise, and ground-level support. We would also like to thank Millenniumsoils Coir and EZ-GRO Inc. for providing materials and technical support.

[1]Corresponding author. E-mail: yzheng@uoguelph.ca.

and nutrient availability and balance (Zheng, 2016). While there are no experimental data on growing substrates for cannabis, the information we collected from the industry indicates that many North American cannabis producers are using either coir- or peat-based substrates, or inert substrates such as rockwool. Different substrates have different physical and chemical properties; therefore, it is essential to fertigate plants accordingly to ensure an adequate root zone environment (Zheng, 2016).

The objective of this study was to determine the optimal organic fertilizer rates for growing vegetative-stage cannabis plants in two coir-based organic growing substrates in a controlled environment growth chamber.

Materials and Methods

Plant culture and treatments. Seventeen-day-old rooted cuttings (≈10 cm high with ≈6 leaves) of cannabis 'OG Kush × Grizzly' were transplanted into round peat-based pots (9.5 cm diameter × 10.2 cm high) with one plant per pot. Pots were filled with one of two growing substrates, ABcann UNIMIX 1-HP (U1-HP) or ABcann UNIMIX 1 (U1) (Physical and chemical properties presented in Tables 1 and 2, respectively; ABcann Medicinals Inc., Napanee, Canada). The two organic substrates were coir-based and with two distinct WHCs: U1-HP with lower WHC and more drainage than U1.

Pots were randomly arranged in a growth chamber at a density of 97 plants/m^2. The growth chamber was set at 22 °C, 85% RH, 500 ppm CO_2 (day and night), and a photosynthetically active radiation (*PAR*) of 250 ± 50 µmol·m^{-2}·s^{-1} at canopy level with an 18-h photoperiod under fluorescent lighting. Beginning 3 d after transplant, plants were hand-fertigated with corresponding nutrient solution to saturation with a 20% leaching fraction when mean substrate moisture was ≈30%, measured using a WET-2 soil moisture sensor (Delta-T Devices Ltd., Cambridge, UK). This was considered the first day of treatment application. Plants were rerandomized after each irrigation event.

The experiment was a completely randomized design with two factors: five fertilizer rates and two substrate types, with 10 replicates for each factor combination. Each potted plant was an experimental unit. Plants were fertilized at one of the five rates of Nutri Plus Organic Grow liquid organic fertilizer (4.0N–1.3P–1.7K; Nutri Plus Grow; EZ-GRO Inc., Kingston, ON, Canada), supplying 117, 234, 351, 468, or 585 mg N/L, diluted with reverse osmosis (RO) water. Other nutrient element concentrations of Nutri Plus Grow were (in mg·L^{-1}): 0.0 Ca, 0.0 Mg, 14.5 Zn, 0.0 Mn, 12.0 B, 2.6 Mo, 2.1 Cu, and 8.5 Fe.

At the end of the vegetative growth period (21 d after transplanting), six plants with representative height and canopy size from each treatment were selected and transferred into a growth chamber for the flowering stage. Plants were potted into 6 L blow-molded black pots (22 cm diameter × 22 cm height) containing a custom blended organic growing substrate (60% sphagnum peat and 40% bulk coconut coir; Premier Tech, Rivière-du-Loup, QC, Canada). Agricultural dolomitic lime (Premier Tech) was incorporated at a rate of 3.0 kg·m^{-3} of substrate. Plants were spaced on tables to a density of 6.5 plants/m^2. The *PAR* was maintained at 500 ± 50 µmol·m^{-2}·s^{-1} with a 12-h photoperiod. Irrigation was administered with one emitter per plant. During the first 11 d in the flowering stage, plants were irrigated whenever the substrate moisture content reached 30% with Nutri Plus Grow at a recommended rate of 140 mg N/L and from then on with a flowering specific fertilizer, Nutri Plus Organic Bloom (2.00N–0.87P–3.32K; EZ-GRO Inc.). Other nutrient element concentrations in Nutri Plus Organic Bloom were (in mg·L^{-1}): 0.0 Ca, 100.0 Mg, 10.0 Zn, 0.0 Mn, 12.8 B, 0.1 Mo, 2.3 Cu, and 6.8 Fe. Nutri Plus Organic Bloom was administered at the following manufacturer-recommended rates: 77 mg N/L from day 12 to 19 in the flowering stage, 103 mg N/L from day 20 to 27, and 129 mg N/L from day 28 to 39. Both vegetative and flowering fertilizers were amended with 2 mL·L^{-1} of an organic calcium-magnesium supplement (3.0N–0.0P–0.0K–3.0Ca–1.6Mg; EZ-GRO Inc.). Between days 39 and 47 in the flowering stage, no fertilizer was applied and the substrates were flushed, as per current industry practice, with RO water: 10 L per pot at 7 d before harvest and 6 L at 5 d before harvest.

Growth and yield measurements. During the vegetative stage, leaf number, canopy area, and plant height were measured every ≈7 d on five randomly selected plants from each treatment. Repeated measurements were made on these same plants throughout the vegetative stage. During the flowering stage, branch number, canopy area, and plant height were measured on all plants every ≈10 d. Growth index for each plant was calculated as height (cm) × length (cm) × width (cm) × 300^{-1} (Ruter, 1992). Plants were harvested after 47 d in the flowering stage on 14 Dec. 2015, when floral resin on most plants had ≈50% amber coloration. Stems were cut at soil level, floral material was cut from stems, and leaves were trimmed thereafter. Floral fresh weight was measured before the floral material was placed in paper bags for drying at 21 °C and 40% RH for 5 d until moisture content reached 11 ± 1%. Dry material was then cured at 18 °C and 60% RH for 14 d before determining the floral dry weight (yield).

Substrate electrical conductivity and pH measurement. Substrate pH and electrical conductivity (EC) were determined weekly using the pour-through method (Wright, 1986) during the vegetative stage and at 4 and 5 weeks of the flowering stage. Pour-through solutions were measured for pH and EC using a HI991300 portable pH/EC/TDS/Temperature Meter (Hanna Instruments, Woonsocket, RI).

Floral cannabinoid analysis. Dried, cured floral material was stored in dark and cool conditions according to United Nations Office on Drugs and Crime (2009) before being analyzed by an independent laboratory (RPC Science and Engineering, Fredericton, NB, Canada). Cannabinoid analysis was performed on the floral material of plants grown in U1 substrate. Analysis of the neutral cannabinoids THC, CBD, and CBN as well as acid forms, THCA and CBDA were conducted by high-performance liquid chromatography as described in section 5.4.8 of United Nations Office on Drugs and Crime (2009).

Statistical analysis. Data were analyzed using JMP Statistical Discovery Version 13.0 (SAS Institute Inc., Cary, NC) at a Type 1 error rate of ≤0.05. Full-factorial ANOVA with repeated measures was used to determine the effects of substrate, fertilizer, and their interaction on substrate EC and pH as well as growth index, leaf number, and branch number over time. Differences among means were tested with Tukey's multiple means comparison test. Two-way ANOVA was used to determine the effects of substrate, fertilizer, and their interaction on yield and the effects of fertilizer on cannabinoid concentrations.

Pearson correlation coefficients were calculated to determine if there is a relationship between growth attributes and final yield. Orthogonal partition and regression analysis (Bowley, 1999) were used to relate substrate EC, pH, plant growth, yield, and cannabinoid concentrations with fertilizer rate and/or yield. If the partitioning variance analysis indicated a significant treatment effect, then the treatment effects were partitioned into one or more regression effects followed by an estimation of regression parameters for the best-fit regression. In all analyses, if there

Table 1. Physical properties of growing substrates ABcann UNIMIX 1-HP (U1-HP) and ABcann UNIMIX 1 (U1).

Growing substrate	Total porosity[z]	CC[z]	Air space[z]	Bulk density[z] (g·cm^{-3})
	(%)			
U1-HP	93 ± 0.4	61 ± 1.2	31 ± 1.3	0.09 ± 0.001
U1	91 ± 0.3	72 ± 0.2	19 ± 0.3	0.10 ± 0.001

[z]Data are means ± SE (n = 3).
CC = container capacity.

Table 2. EC, pH, and nutrient content measured using the saturated paste method for growing substrates ABcann UNIMIX 1-HP (U1-HP) and ABcann UNIMIX 1 (U1).

Growing substrate	EC[z] (mS·cm^{-1})	pH[z]	Nitrate N	P	K	Ca	Mg	SO_4^{2-}	Na	Cl^-	Zn	Mn	Cu	Fe	B	Mo
			(mg·L^{-1})													
U1-HP	1.8 ± 0.07	6.30 ± 0.01	5	9.2	338.1	<1	2.7	31.2	104.5	413	<0.01	<0.01	<0.01	0.12	0.09	<0.01
U1	2.3 ± 0.12	6.28 ± 0.01	8	10.4	431.2	2.3	5.3	41.3	136.3	724	<0.01	0.01	<0.01	0.84	0.13	<0.01

[z]Data are means ± SE (n = 3).
EC = electrical conductivity; N = nitrogen; P = phosphorus; K = potassium; Ca = calcium; Mg = magnesium; SO_4^{2-} = sulfate; Na = sodium; Cl^- = chloride; Zn = zinc; Mn = manganese; Cu = cupper; Fe = iron; B = boron; Mo = molybdenum.

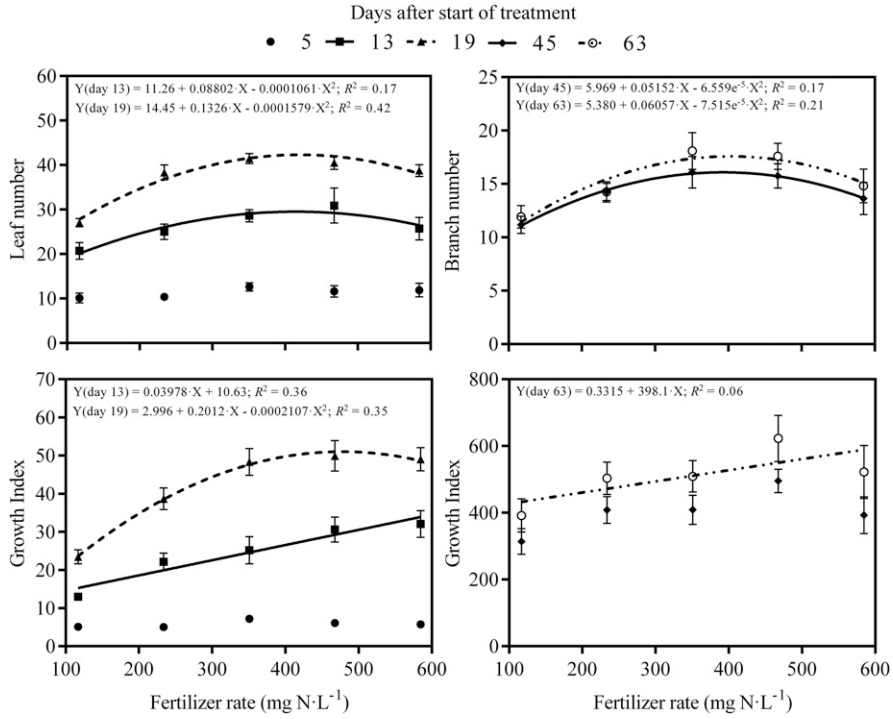

Fig. 1. Response of cannabis growth attributes to organic fertilizer (4.0N–1.3P–1.7K) rate [indicated by nitrogen (N) concentration] applied during the vegetative stage. Values are means ± SE and lines are the best fit regression relationships at $P < 0.05$. For days 5 and 13, $n = 10$; for day 19, $n = 20$ (vegetative stage; left); for days 45 and 63 (flowering stage; right) at rates that supplied 117, 234, and 468 mg N/L, $n = 12$; at rates that supplied 351 and 585 mg N/L, $n = 11$.

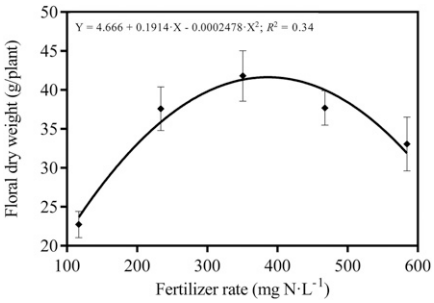

Fig. 2. Response of cannabis yield to organic fertilizer (4.0N–1.3P–1.7K) rate [indicated by nitrogen (N) concentration] applied during the vegetative stage. Values are means ± SE. The curve is the best fit regression relationship with $P < 0.05$ ($n = 12$ at rates that supplied 117, 234, and 468 mg N/L; $n = 11$ for rates that supplied 351 and 585 mg N/L).

was no significant treatment effect, then data were presented as the average of all the treatments (pooled). If cannabinoid concentrations were below the detection limit (<0.05%), the values were excluded from the analysis. The residuals of the above analyses were tested for normality and equality of variance using the Shapiro–Wilk test and Bartlett's test, respectively.

Results

Growth. There were no observed signs of nutrient toxicity or deficiency at any fertilizer rate during the vegetative or flowering stage. Identifying nutrient disorders based on visible foliar symptoms became difficult at 6 weeks of the flowering stage when all plants began showing signs of foliar senescence. Older leaves started to become chlorotic in week 6 and eventually necrotic before harvest. Substrate and substrate × fertilizer rate had no effect on leaf number, branch number, or growth index. During the vegetative stage, both leaf number and growth index responded similarly to fertilizer rate (Fig. 1). Growth attributes did not respond to fertilizer rate at 5 d after the first treatment application (DAT), although growth index responded to fertilizer rate linearly at 13 DAT and quadratically at 19 DAT, and leaf number responded to fertilizer rate quadratically at 13 and 19 DAT. At 19 DAT, maximum leaf number was 42, achieved at a rate that supplied 420 mg N/L and maximum growth index was 51 at a rate that supplied 477 mg N/L. Treatment effects carried forward into the flowering stage in which branch number responded quadratically to vegetative-stage fertilizer rate (maximum of 17.6 at a rate that supplied 403 mg N/L) and growth index increased linearly with increasing vegetative-stage fertilizer rate.

Yield. There was no yield difference between substrates, and no substrate × fertilizer rate effect on yield. Based on the pooled data from both substrates, yield responded to fertilizer rate quadratically with the highest yield at a rate that supplied 389 mg N/L (Fig. 2). Yield at this fertilizer rate was interpolated to be 41.6 g/plant which is 1.8 times higher than that at the lowest which supplied 117 mg N/L. The yield was positively correlated with growth index ($r = 0.45$, $P < 0.001$), leaf number ($r = 0.39$, $P = 0.0027$), and branch number ($r = 0.53$, $P < 0.0001$) measured at the end of the vegetative stage (19 DAT; $n = 58$).

Cannabinoids. Of the analyzed cannabinoids, only THC, THCA, and CBN were above the detection limit (0.05%). Floral THC concentration responded quadratically to increasing fertilizer rate, reaching a maximum of 0.31% a rate supplying 418 mg N/L (Fig. 3). There was no fertilizer rate effect on the floral THCA concentration (mean ± SE of 10.6% ± 0.31%) or CBN concentration (mean ± SE of 0.08% ± 0.018%). Cannabinoid concentrations also varied with yield. THC and CBN were positively correlated with yield, whereas THCA was not correlated with yield (Fig. 4).

Substrate EC and pH. Substrate pH decreased over time for all fertilizer rates during the vegetative stage (Fig. 5), decreasing linearly or responding quadratically to increasing fertilizer rate. The lowest mean pH was 6.19 at the 351 mg N/L rate, measured at 17 DAT. Substrate EC, measured at 5, 13, and 17 DAT, increased linearly over time and with increasing fertilizer rate. Mean EC ranged from 0.9 to 3.9 mS·cm^{-1} from the lowest to the highest fertilizer rate at 17 DAT. In the flowering stage, pH (measured at 47 and 59 DAT) increased linearly with increasing vegetative-stage fertilizer rate with means ranging from 6.74 to 7.16 (Fig. 6). No difference was observed in EC among vegetative stage fertilizer rates during the

flowering stage with substrate EC at 1.3 ± 0.03 mS·cm^{-1} and 1.6 ± 0.02 mS·cm^{-1} (mean ± SE) at 47 and 59 DAT, respectively. No differences in substrate EC or pH were observed between the two tested substrates in both the vegetative and flowering stages.

Discussion

No visual signs of nutrient disorders were observed in this trial which suggests that the fertilizers used had nutrient elements and ratios within an acceptable range. Both growth attributes and yield of the cannabis plants exhibited a typical response to varying fertilizer application rates. Yield increased with increasing fertilizer until reaching a maximum at a rate supplying 389 mg N/L. Optimal organic fertilizer application rates in this experiment were higher than synthetic fertilizer recommendations for most conventional crops (Raviv and Lieth, 2007). Organic fertilizers contain slower releasing and less soluble forms of nitrogen and phosphorus compared with most synthetic fertilizers and may release only 25% to 60% of their nitrogen content (Prasad et al., 2004). Therefore, it is important to establish organic-specific fertilizer rates rather than using conventional standards as guidelines. Results from our present study showed a fertilizer rate supplying 389 mg N/L provided the highest yield while increasing or having no effect on the concentration of the cannabinoids measured.

Substrate EC increased over time during the vegetative stage, and the increase was more apparent at higher fertilizer rates. Suboptimal yields were seen at fertilizer rates that supplied 468 and 585 mg N/L under which substrate EC was 3.0 ± 0.13 and 3.8 ± 0.13 mS·cm^{-1}, respectively. These yield reductions may have been caused by high substrate salinity. High salinity can damage crops through increased ψ_S, depressing the external water potential in the root zone. In greenhouse-grown flowering crops, salinity thresholds vary dramatically among species, ranging in EC from of 1.0 to >4.2 mS·cm^{-1} (Sonneveld et al., 1999). In the current study, cannabis tolerated substrate EC up to 3.0 mS·cm^{-1} without reduction in yield.

In all fertilizer rates, pH decreased gradually during the vegetative stage; and the highest yielding rates, which supplied 234, 351 and 468 mg N/L, exhibited the lowest pH values. In most organic fertilizers, nitrogen exists primarily as NH_4^+ (i.e., high NH_4^+-N/NO_3^--N ratio; Gül et al., 2007) and can be taken up by plants directly or as other forms after being converted by microorganisms in the substrate via ammonification and nitrification (Shinohara et al., 2011). Reductions in pH under organic fertilization can be caused by NH_4^+ nitrification and the excretion of protons by the roots after NH_4^+ uptake (Johnson et al., 2011; Silber et al., 2004). It is possible that larger plants, those fertilized at rates identified as, or close to, optimal in this study, had higher rates of NH_4^+ uptake which decreased root zone pH. There are no experimental data in the literature on ideal growing substrate pH range for cannabis in soilless production system; however, information we collected from the industry and gray resources (Cervantes, 2006) suggest a range of 5.8–6.8 to avoid causing nutrient disorders. In the current study, there were no visual signs of pH-induced disorder in plants within the pH ranges measured (means of 6.2 to 7.1 in the vegetative stage and 6.7 and 7.2 in the flowering stage) suggesting that these ranges are suitable for container production of organic cannabis. More research is needed to determine the optimal growing substrate pH ranges for cannabis.

Around the optimal fertilizer rate, both growing substrates tested in the current study demonstrated acceptable qualities for the growth of cannabis in the vegetative stage. There were no growth or yield differences observed between plants grown in the lower WHC (drier) substrate (U1-HP) and the higher WHC (wetter) substrate (U1) with fertigation administered when substrate moisture content dropped to 30%. This indicated that both substrates were appropriate for container production of organic cannabis.

The positive correlations between growth attributes in the vegetative stage and final yield may indicate that growing larger plants during the vegetative stage will increase yield. Because larger plants, those fertilized at rates around the optimal fertilizer rate, had increased THC concentration in floral material (maximized at the rate supplying 418 mg N/L) and the concentrations of other cannabinoids were unaffected, it may be concluded that to optimize the yield and total THC content, cultivation techniques to increase vegetative growth, specifically branching, should be used. Besides fertigation, other cultural practices such as topping (Tanaka and Fujita, 1974) may also be used to increase branching.

The highest yielding plants, those fertilized around the optimal rate, had higher concentrations of THC and CBN. In fact, as yield increased, so did the concentration of

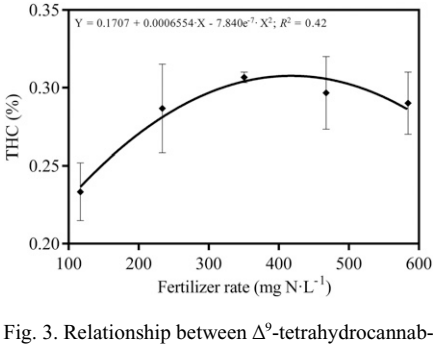

Fig. 3. Relationship between Δ^9-tetrahydrocannabinol (THC) concentration in dry floral material of cannabis and organic fertilizer (4.0N–1.3P–1.7K) rate [indicated by nitrogen (N) concentration] applied during the vegetative stage. Values are means ± SE. The curve is the best fit regression relationship with $P < 0.05$ ($n = 3$).

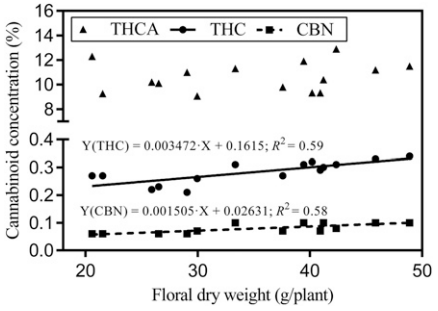

Fig. 4. Relationships between cannabinoid concentrations in dry floral material of cannabis and dry floral weight. Values are means ± SE ($n = 15$ for THCA and THC; $n = 13$ for CBN). Lines are the best fit regression relationships with $P < 0.05$. THCA = Δ^9-tetrahydrocannabidiolic acid; THC = Δ^9-tetrahydrocannabinol; CBN = cannabinol.

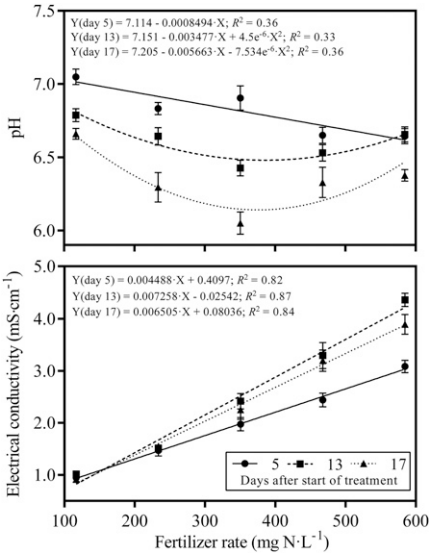

Fig. 5. Response of substrate pH and electrical conductivity to organic fertilizer (4.0N–1.3P–1.7K) rate [indicated by nitrogen (N) concentration] applied during the vegetative stage. Data are means ± SE ($n = 5$ for pH at the 585 mg N/L rate on day 17 and $n = 10$ for all other means) and lines are the best fit regression relationships with $P < 0.05$.

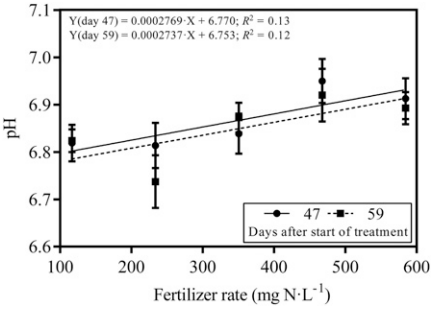

Fig. 6. Response of substrate pH during the flowering stage to organic fertilizer (4.0N–1.3P–1.7K) rate [indicated by nitrogen (N) concentration] applied during the vegetative stage. Data are means ± SE ($n = 8$), and lines are the best fit regression relationships with $P < 0.05$.

these neutral cannabinoids. During the flowering stage, THCA transcription in floral material slows between weeks 1 and 3 whereas total cannabinoid concentration continues to increase until weeks 3 and 4, as THCA breaks down into neutral cannabinoids such as THC and CBN (Muntendam et al., 2012). This leads to an accumulation of neutral cannabinoids as plants mature through the flowering stage. It is estimated that higher concentrations of neutral cannabinoids, as seen in plants fertilized around the optimal rate, would be observed in plants which mature early. Optimal fertigation during the vegetative stage may, therefore, reduce maturation time in cannabis. Early maturation is desirable as it could decrease time to harvest and result in more frequent crop turnover. To evaluate whether fertigation can, in fact, reduce maturation time, further study is required with cannabinoid analyses throughout the flowering stage.

In the current study, treatments were applied only in the vegetative stage whereas cannabinoid production occurs primarily during flowering because of an increase in glandular trichome development in the flowering stage (Muntendam et al., 2012; Vogelmann et al., 1988). Treatment effects carried forward to some final floral cannabinoid concentrations; however, effects may have been more apparent with variable fertilizer treatments during the flowering stage. Further research is needed to evaluate the effects of fertilizer rate on flowering-stage cannabis.

Yields in the current study were slightly lower than industry standards and reports from recent horticultural studies on cannabis (Potter and Duncombe, 2012; Vanhove et al., 2011, 2012). The 47-d flowering period in the current study was relatively short, compared with the 7–9-week range in these cited studies. A shorter flowering period is known to reduce yields (Potter, 2014). Other factors including cannabis variety and the use of organic fertilizer may have also played a role.

Conclusions

Our results demonstrated that to produce high-yielding, cannabinoid-rich plants, the optimal fertilizer rate was that supplying about 389 mg N/L, for Nutri Plus Organic Grow liquid organic fertilizer (4.0N–1.3P–1.7K) in the vegetative growth stage of cannabis using coir-based organic substrates. These recommendations should be acceptable for similar organic fertilizer and substrates; however, different cannabis varieties may have different fertilization requirements. To provide variety-specific fertilization requirements, further study may be needed. Both organic substrates ABcann UNIMIX 1-HP and ABcann UNIMIX 1 maintained suitable pH (between 6.2 and 7.1 in the vegetative stage and between 6.7 and 7.2 in the flowering stage) and were effective for vegetative-stage cannabis growth; however, U1-HP may require more frequent fertigation than U1. Growing substrate EC of up to 3.0 $mS \cdot cm^{-1}$ was tolerated without yield reductions. Furthermore, larger plants (e.g., higher growth index, branching and leaf number) generally had higher yield and floral THC concentrations which may indicate that plants should be grown as large as possible during the vegetative stage.

Literature Cited

Ahmed, S.A., S.A. Ross, D. Slade, M.M. Radwan, I.A. Khan, and M.A. ElSohly. 2015Minor oxygenated cannabinoids from high potency *Cannabis sativa* L. Phytochemistry 117:194–199.

Ahmed, S.A., S.A. Ross, D. Slade, M.M. Radwan, F. Zulfiqar, and M.A. ElSohly. 2008. Cannabinoid ester constituents from high potency *Cannabis sativa* L. Planta Med. 74:536–542.

Amaducci, S., D. Scordia, F.H. Liu, Q. Zhang, H. Guo, G. Testa, and S.L. Cosentino. 2015. Key cultivation techniques for hemp in Europe and China. Ind. Crops Prod. 68:2–16.

ArcView Market Research and New Frontier. 2014. The state of legal marijuana markets. 4th ed. ArcView Mkt. Res. New Frontier, San Francisco, CA.

Aubin, M.-P., P. Seguin, A. Vanasse, G.F. Tremblay, A.F. Mustafa, and J.-B. Charron. 2015. Industrial hemp response to nitrogen, phosphorus, and potassium fertilization. Crop Forage Turfgrass Mgt. 1:1–10.

Baxter, W.J. and G. Scheifele. 2008. Growing industrial hemp in Ontario. Ontario Ministry of Agriculture, Food and Rural Affairs. Queen's Printer for Ontario, Toronto, Canada.

Bowley, S. 1999. A hitchhiker's guide to statistics in plant biology. Any Old Subject Books, Guelph, Canada.

Canada Gazette. 2016. SOR/2016-230, s.2. Access to cannabis for medical purposes regulations. P.C. 2016-743 (5 Aug. 2016). Canada Gazette, Part II. 150(17) 3525. 23 Nov. 2016. <http://gazette.gc.ca/rp-pr/p2/2016/2016-08-24/html/sor-dors230-eng.php>. Archived 25 Aug. 2016.

Cervantes, J. 2006. Marijuana horticulture: The indoor/outdoor medical grower's bible. Van Patten Publishing, Vancouver, WA.

Ehrensing, D.T. 1998. Feasibility of industrial hemp production in the United States pacific northwest. Oregan State Univ. Agr. Expt. Sta. Bul. 681.

ElSohly, M.A. and D. Slade. 2005. Chemical constituents of marijuana: The complex mixture of natural cannabinoids. Life Sci. 78:539–548.

Elzinga, S., J. Fischedick, R. Podkolinski, and J.C. Raber. 2015. Cannabinoids and terpenes as chemotaxonomic markers in cannabis. Nat. Prod. Chem. Res. 3:1–9.

Farag, S. and O. Kayser. 2015. Cultivation and breeding of *Cannabis sativa* L. for preparation of standardized extracts for medicinal purposes. Medicinal and aromatic plants of the world. Springer Netherlands, Dordrecht, The Netherlands.

Gül, A., F. Kıdoğlu, and D. Anaç. 2007. Effect of nutrient sources on cucumber production in different substrates. Sci. Hort. 113:216–220.

Hillig, K.W. and P.G. Mahlberg. 2004. A chemotaxonomic analysis of cannabinoid variation in cannabis (*Cannabaceae*). Amer. J. Bot. 91: 966–975.

Johnson, C.N., P.R. Fisher, R.P. Vetanovetz, and W.R. Argo. 2011. Quantifying the acidity and basicity of fertilizers in container substrates. Acta Hort. 891:159–166.

Kimura, M. and K. Okamoto. 1970. Distribution of tetrahydrocannabinolic acid in fresh wild cannabis. Experientia 26:819–820.

Leggett, T. 2006. Review of the world cannabis situation. Bull. Narc. 58:1–155.

Mechoulam, R., A. Shani, H. Edery, and Y. Grunfeld. 1970. Chemical basis of hashish activity. Science 169:611–612.

Muntendam, R., N. Happyana, T. Erkelens, F. Bruining, and O. Kayser. 2012. Time dependent metabolomics and transcriptional analysis of cannabinoid biosynthesis in *Cannabis sativa* var. Bedrobinol and Bediol grown under standardized condition and with genetic homogeneity. Intl. J. Med. Plants Res. 1:31–40.

Potter, D.J. 2014. A review of the cultivation and processing of cannabis (*Cannabis sativa* L.) for production of prescription medicines in the UK. Drug Test. Anal. 6:31–38.

Potter, D.J. and P. Duncombe. 2012. The effect of electrical lighting power and irradiance on indoor-grown cannabis potency and yield. J. Forensic Sci. 57:618–622.

Prasad, M., P. Simmons, and M.J. Maher. 2004. Release characteristics of organic fertilizers. Acta Hort. 644:163–170.

Raviv, M. and J.H. Lieth. 2007. Soilless culture: Theory and practice. 1st ed. Elsevier B.V., Burlington, MA.

Ross, S.A. and M.A. ElSohly. 1997. CBN and Δ9-THC concentration ratio as an indicator of the age of stored marijuana samples. Bull. Narc. 49:139.

Russo, E.B. 2007. History of cannabis and its preparations in saga, science, and sobriquet. Chem. Biodivers. 4:1614–1648.

Russo, E.B. 2011. Taming THC: Potential cannabis synergy and phytocannabinoid-terpenoid entourage effects. Brit. J. Pharmacol. 163:1344–1364.

Ruter, J.M. 1992. Influence of source, rate, and method of applicating controlled release fertilizer on nutrient release and growth of ''Savannah'' holly. Fert. Res. 32:101–106.

Shinohara, M., C. Aoyama, K. Fujiwara, A. Watanabe, H. Ohmori, Y. Uehara, and M. Takano. 2011. Microbial mineralization of organic nitrogen into nitrate to allow the use of organic fertilizer in hydroponics. Soil Sci. Plant Nutr. 57:190–203.

Silber, A., L. Ben Yones, and I. Dori. 2004. Rhizosphere pH as a result of nitrogen levels and NH_4/NO_3 ratio and its effect on zinc availability and on growth of rice flower (*Ozothamnus diosmifolius*). Plant Soil 262:205–213.

Sonneveld, C., R. Baas, H.M.C. Nijssen, and J. de Hoog. 1999. Salt tolerance of flower crops grown in soilless culture. J. Plant Nutr. 22:1033–1048.

Surrage, V.A., C. Lafrenière, M. Dixon, and Y. Zheng. 2010. Benefits of vermicompost as a constituent of growing substrates used in the production of organic greenhouse tomatoes. HortScience 45:1510–1515.

Tanaka, A. and K. Fujita. 1974. Nutriophysiological studies on the tomato plant. Soil Sci. Plant Nutr. 20:305–315.

Taschwer, M. and M.G. Schmid. 2015. Determination of the relative percentage distribution of THCA and Δ(9)-THC in herbal cannabis seized in Austria - Impact of different storage temperatures on stability. Forensic Sci. Intl. 254:167–171.

United Nations Office on Drugs and Crime. 2009. Recommended methods for the identification and analysis of cannabis and cannabis products, manual for use by national drug analysis laboratories. Laboratory and Scientific Section, United Nations Office on Drugs and Crime, Vienna, Austria.

van Bakel, H., J.M. Stout, A.G. Cote, C.M. Tallon, A.G. Sharpe, T.R. Hughes, and J.E. Page. 2011. The draft genome and transcriptome of *Cannabis sativa*. Genome Biol. 12:1–17.

Vanhove, W., T. Surmont, P. Van Damme, and B. De Ruyver. 2012. Yield and turnover of illicit indoor cannabis (*Cannabis* spp.) plantations in Belgium. Forensic Sci. Intl. 220:265–270.

Vanhove, W., P. Van Damme, and N. Meert. 2011. Factors determining yield and quality of illicit indoor cannabis (*Cannabis* spp.) production. Forensic Sci. Intl. 212:158–163.

Vera, C.L., S.S. Malhi, J.P. Raney, and Z.H. Wang. 2004. The effect of N and P fertilization on growth, seed yield and quality of industrial hemp in the Parkland region of Saskatchewan. Can. J. Plant Sci. 84:939–947.

Vogelmann, A.F., J.C. Turner, and P.G. Mahlberg. 1988. Cannabinoid composition in seedlings compared to adult plants of *Cannabis sativa*. J. Nat. Prod. 51:1075–1079.

Vollner, L., D. Bieniek, and F. Korte. 1986. Review of analytical methods for identification and quantification of cannabis products. Regul. Toxicol. Pharmacol. 6:348–358.

Wang, Y.T. 2000. Impact of a high phosphorus fertilizer and timing of termination of fertilization on flowering of a hybrid moth orchid. HortScience 35:60–62.

Wright, R.D. 1986. The pour-through nutrient extraction procedure. HortScience 21:227–229.

Wright, R.D. and A.X. Niemiera. 1987. Nutrition of container-grown woody nursery crops. Hort. Rev. 9:75–101.

Zhai, Z., D.L. Ehret, T. Forge, T. Helmer, W. Lin, and A.P. Papadopoulos. 2009. Organic fertilizers for greenhouse tomatoes: Productivity and substrate microbiology. HortScience 44:800–809.

Zheng, Y. 2016. Root zone environment management in container crop production. Proc. for the Veg., potato, greenhouse, small fruit & Gen. session, Mid-Atlantic Fruit & Veg. Convention, Hershey, PA.