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ORIGINAL ARTICLE

Improving the quality of matured coconut (*Cocos nucifera* Linn.) water by low alcoholic fermentation with *Saccharomyces cerevisiae*: antioxidant and volatile profiles

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Abstract Matured coconut water (MCW) is a by-product in the coconut milk industry that is usually discarded due to its unpleasant flavor. In this study, low-alcohol coconut water (LACW) was fermented with *Saccharomyces cerevisiae* to improve the quality of MCW. Volatile components and nonvolatile flavor-related elements were estimated to compare the qualities of the MCW and LACW. Besides measuring the kinetic changes, the levels of fructose, glucose, sucrose and ethanol contents were also determined. The results of the organic acid assays showed that tartaric, pyruvic and succinic acids were the primary organic acids present in LACW and increased significantly with fermentation. The resulting volatile composition assay indicated that esters, alcohols and fatty acids were significantly influenced by fermentation and yeast strains. Moreover, 1,1-diphenyl-2-picrylhydrazyl (DPPH), 2,2′-azinobis-(3-ethylbenzothiazoline-6-sulfonic acid) (ABTS), cupric ion reducing antioxidant capacity and ferric reducing antioxidant power values increased significantly throughout the process, correlating with the enhancement of total phenolic content.

Keywords Matured coconut water · Quality improvement · *Saccharomyces cerevisiae* · Antioxidant capacity · Volatile component

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Introduction

Coconut is an important fruit in tropical regions and can be made into different foods and beverages. On average, a full coconut is composed of 33% husk, 16% shell, 33% kernel and 18% coconut water (Assa et al. 2010). Both the coconut meat and coconut water are edible parts (Yong et al. 2009). Coconut water is popular for containing nutrients such as salts, sugars, vitamins, amino acids and minerals. There is increasing scientific evidence that supports the role of coconut water in health and medicinal applications (Chowdhury et al. 2005), such as biologically functional roles in the human metabolic system and treatment for gastroenteritis.

Coconut water is classified into two categories according to the harvesting time: tender coconut water (TCW) and matured coconut water (MCW). The TCW can be drunk directly or processed into various kinds of beverages. However, due to the unpleasant flavor of the MCW, it is usually discarded as a by-product in the coconut milk industry.

In recent decades, low-alcohol fermented fruit juice has been a hot research topic (Pérez-Gregorio et al. 2011). Many kinds of fruit, such as apple, orange and carrot have been fermented to produce low-alcohol drinks (Rita et al. 2011; Escudero-López et al. 2015). A novel fermented beverage using coconut water as the main ingredient was developed by Prado et al. (2015). In addition, Watawana et al. (2016) used kombucha "tea fungus" to ferment king coconut water for 7 days. The results indicated that the pH value decreased significantly from approximately 5.1 to 4.0 after fermentation. However, the flavor of the fermented coconut water was not mentioned.

There is evidence that supports the many claims that excessive alcohol drinkers have a higher probability of death from injuries, violence, poisoning, liver cirrhosis, certain cancers, and hemorrhagic stroke compared with nondrinkers (Wang et al. 2008). Nevertheless, studies in recent years have presented a new point of view. There is a U-shaped relationship between the amount of alcohol consumed and cardiovascular risk. In addition, the evidence indicates that light to moderate drinkers are associated with lower cardiovascular death rates compared to heavier drinkers and abstainers (Gaziano et al. 2000; Markus et al. 2015). However, limited studies about fermented coconut water, especially MCW, have been conducted in the beverage industry.

The major objective of this study was to improve the flavor and bioactive properties of MCW using low alcoholic fermentation with three *Saccharomyces cerevisiae* strains (Lalvin R-HST, DV10 and D254). MCW with light to moderate levels of alcohol would be conducive to reducing the cardiovascular risk of drinkers. In addition to this objective, the changes in antioxidant activity (including 2,2′-azinobis-(3-ethylbenzothiazoline-6-sulfonic acid) (ABTS), 1,1-Diphenyl-2-picrylhydrazyl (DPPH), ferric reducing antioxidant power (FRAP) and cupric ion reducing antioxidant capacity (CUPRAC) values) were also estimated during the fermentation of LACW. Antioxidants are proven to contribute to inhibiting premature aging and dementia. In addition, the sucrose, fructose, glucose, ethanol, total phenol, organic acid and volatile compound contents of MCW during fermentation were analyzed.

Materials and methods

Materials

Active dry yeast strains of *Saccharomyces cerevisiae* (Lalvin R-HST, DV10, and D254) were obtained from Lallemand Inc. (Montreal, Quebec, Canada). Vietnam tall coconut (*Cocos nucifera L.*) water, a by-product of coconut milk from TwinCoco Inc. (Haikou, China), was sampled randomly from a large batch of discarded coconut water. DPPH, ABTS, 6-hydroxy-2,5,7,8-tetramethylchroman-2-carboxylic acid (Trolox), and 2,4,6-tris(2-pyridyl)-s-triazine (TPTZ) were acquired from Sigma-Aldrich Chemical Co. (Milwaukee, WI, USA). Citric acid, malic acid, tartaric acid, succinic acid, acetic acid, oxalic acid, pyruvic acid, ketoglutaric acid, and Folin-Ciocalteu phenol reagent were purchased from Merck company (Merck, Darmstadt, Germany). Fructose, glucose, sucrose, diatomite and macroporous resin D301R were obtained from the Aladdin Industrial Corporation (Aladdin, Shanghai, China). All other chemicals were of analytical grade unless otherwise noted.

Methods

Fermentation

Coconut water was first filtered with diatomite, and then, 0.02% sodium metabisulfite was added. The total soluble solids content was adjusted from an initial value of 6.54°Brix to approximately 10°Brix with sucrose. Next, 5% yeast cultures were inoculated in the coconut water to ferment. The fermentation was carried out in triplicate in three parallel conical flasks (2 L) at 25 °C for 48 h. Finally, samples were collected aseptically at 0, 8, 16, 24, 32, 40 and 48 h and immediately stored at − 80 °C. The supernatant of the samples was obtained by centrifugation ($4000\times g$, 10 min).

Determination of the total soluble solids and alcohol content

The °Brix values were measured by a portable refractometer (ATAGO PAL-1, Tokyo, Japan). The concentration of ethanol was quantified by a distillation method using a portable alcohol meter (KEM DA-130 N, Tokyo, Japan). In brief, the distilling compounds were distilled from a 20 mL sample and diluted to 20 mL by distilled water. The diluents were then injected to the portable alcohol meter to test the alcohol concentration.

Determination of soluble sugars

A 1.5 g pre-treated anion-exchange resin D301R (a weakly alkaline anion-exchange resin with larger pores and a structure that is a co-polymer of styrene–divinylbenzene with a tertiary amine group) and a 5 mL sample were added to a flask for adsorption. The adsorption flask was kept in a water bath with magnetic stirring for 2 h at 50 °C (Buran et al. 2014). The above mixture was subjected to centrifugation for 10 min at $4000\times g$, and the supernatant was filtered through a 0.45 μm nylon membrane for HPLC analyses.

The sugar content was measured according to the method of Morales et al. (2016) with some modifications. Separation was conducted using an Agilent 1200 system equipped with a ZORBAX carbohydrate analysis column (4.6 × 250 mm, 5 μm; Agilent, USA) and a G1362A RID detector. The HPLC analysis was conducted by isocratic elution using a mobile phase composed of ACN and H_2O (ACN: H_2O = 75: 25, v/v) at a flow rate of 1.0 mL/min. Standards and samples (5 μL) were injected in triplicate. The column was washed with the mobile phase for 5 min before the next injection.

Determination of the organic acid content

The organic acid content was determined using an HPLC method (Coelho et al. 2015) with an Agilent Technologies 1260 series machine including a G1312B pump and a G1314C diode-array detector (DAD). Standards and samples were filtered (0.22 μm) and injected on a ZORBAX SB-Aq StableBond analysis column (4.6 × 250 mm, 5 μm; Agilent, USA) in triplicate. The wavelength of the DAD was maintained at 210 nm with a run time of 15 min for analysis of oxalic, tartaric, succinic, malic, lactic, acetic, pyruvic and ketoglutaric acids. The HPLC analysis was conducted by isocratic elution using a mobile phase composed of a 0.02 M buffer solution of KH_2PO_4 (pH 2.65) and methyl alcohol (97:3) at a flow rate of 1.0 mL/min. In addition, after each analysis, the column was washed with the mobile phase for 5 min.

Determination of the total phenol content (TPC)

The TPC of samples was determined using the Folin–Ciocalteu colorimetric method of de Camargo et al. (2012) with some modifications. The sample (0.5 mL) was diluted (1:6) with distilled water in a test tube, and Folin–Ciocalteu reagent (0.2 mL) was added. The mixture was mixed thoroughly and kept in the dark for 5 min at room temperature. Then 1.3 mL of a 10% Na_2CO_3 saturated solution was added and kept in the dark for another 1.5 h. The absorbance of the above mixture was evaluated against a reagent blank at 765 nm using a TU-1810 spectrophotometer (Persee, Beijing, China). A calibration curve was measured using gallic acid (0–8.2 μg/mL) as a standard, and the results were expressed as microgram gallic acid equivalents per milliliter of sample.

Determination of the DPPH radical scavenging capacity

The DPPH assay was carried out according to a previous study (Escudero-LóPez et al. 2013) with slight modifications. The DPPH solution was diluted with methanol to achieve an absorbance of 1.2–1.3 at 517 nm. For the assay, 4 mL of DPPH solution was added to 1 mL of sample. The solution was mixed well and incubated in the dark at 30 °C for 45 min, and the DPPH absorbance was read at 517 nm using a TU-1810 spectrophotometer (Persee, Beijing, China). The DPPH radical scavenging capacities of the samples were calculated using the following equation:

$$\text{DPPH radical scavenging capacity (\%)} = [(A_0 - A_1)/A_0] \times 100$$

where A_0 is the absorbance of the control (methanol) and A_1 is the absorbance in the presence of the sample.

Determination of the ABTS scavenging capacity

The ABTS assay was performed using a modified method described by Koksal et al. (2011). The ABTS assay is based on the scavenging of 2,2-azinobis- (3-ethylbenzothiazoline-6-sulfonate) radical anions. A solution of ABTS radicals was produced by reacting a 7.4 mM ABTS solution with 2.6 mM of potassium persulfate solution and storing the mixture in a dark place at room temperature for 12–16 h prior to use. The ABTS solution was diluted to an absorbance of 0.70 ± 0.02 at 734 nm using 5 mM phosphate-buffered saline (pH 7.4). In addition, 3.8 mL of the dilution was added to 0.2 mL of each sample. The decrease in absorbance of the above mixtures was read at 734 nm after 6 min using a TU-1810 spectrophotometer (Persee, Beijing, China). The ABTS radical scavenging capacities of the samples were calculated using the following equation:

$$\text{ABTS radical scavenging capacity (\%)} = [(A_0 - A_1)/A_0] \times 100$$

where A_0 is the absorbance of the control (phosphate-buffered saline) and A_1 is the absorbance in the presence of the sample.

FRAP assay

The FRAP assay was conducted according to a method described by de Camargo et al. (2014). The FRAP reagent was composed of a 0.3 M acetate buffer (pH 3.6) solution, 20 mM $FeCl_3 \cdot 6H_2O$ solution, and 10 mM TPTZ solution (acetate buffer: $TFeCl_3 \cdot 6H_2O$: TPTZ = 10:1:1, v/v/v). Samples (0.5 mL) were reacted with a 3.0 mL FRAP solution for 30 min in dark conditions, and the absorbance was read at 593 nm. $FeSO_4 \cdot 7H_2O$ was used as the standard, and the standard curve was linear between 20 and 600 μM $FeSO_4 \cdot 7H_2O$. The results are expressed as μM FeSO4·7H2O equivalents.

CUPRAC assay

The CUPRAC assay was performed according to the method described by Magalhães et al. (2012). It is based on the reduction of a cupric neocuproine complex (Cu(II)–Nc) by antioxidants to the cuprous form (Cu(I)–Nc). For the assay, 1.0 mL of each sample was added to a solution containing 10 mM CuSO4, 7.5 mM cupric neocuproine and 1 M NH4Ac buffer. The solution was mixed well and stored in the dark at room temperature for 30 min. The absorbance of the above mixture at 450 nm was evaluated against a reagent blank using a TU-1810 spectrophotometer (Persee, Beijing, China). Trolox was used as the standard, and the standard curve was linear between 25 and

320 μM Trolox. The results are expressed as μM Trolox equivalents.

Determination of volatile compounds

The volatile compounds in the fresh and fermented coconut waters were analyzed using headspace (HS) solid-phase microextraction (SPME) sampling combined with a gas chromatography-mass spectrometer (GC–MS) (HS–SPME–GC–MS) according to the method described by Lee et al. (2012). Samples were added into a headspace bottle for balance at 45 °C. Then, the samples were extracted with HS-SPME at 45 °C for 30 min using an SPME autosampler (Supelco, Pennsylvania, USA) under magnetic stirring and thermally desorbed into the injector port at 240 °C for 3 min. Separation was performed on a Ptx-Wax capillary column (30 m × 0.25 mm, Shimadzu, Japan). In addition, the oven temperature increased from 50 °C (hold time: 3 min) to 120 °C at 10 °C/min and then increased to 230 °C at 8 °C/min (final hold time: 10 min). The eluted volatile compounds were identified by matching the mass spectrum against the NIST 11 libraries and confirmed with linear retention index (LRI) values.

Statistical analysis

Analyses of the variance were performed, and the mean values ± standard deviation were evaluated by Duncan's multiple range test ($p < 0.05$), using SPSS version 17.0 statistical software (SPSS Inc. Chicago., IL, USA) and DPS V6.55 statistical software (DPS Soft Inc., Tang, Hangzhou, China). Origin (Origin Lab Co., Pro.8.0, USA) software was used for data processing and to create charts.

Results and discussion

The variation of fructose, glucose, sucrose and ethanol content during fermentation

Changes in the concentrations of fructose, glucose, sucrose and ethanol during the fermentation of MCW with three yeast strains (D254, DV10 and R-HST) are shown in Fig. 1. Three medium cultures showed similar characteristics in terms of the °Brix value (Fig. 1a). The °Brix value decreased only slightly in the first 16 h but decreased significantly from 16 to 48 h. Theoretically, *Saccharomyces cerevisiae* ferments only glucose, fructose and sucrose to produce ethanol and CO_2 via the Entner-Doudoroff pathway (Musatti et al. 2015). Fructose and glucose in coconut water came primarily from two sources: the original content of the coconut water (13.57 g/L of glucose and 4.09 g/L of fructose) and the conversion from sucrose (Xiong et al. 2016).

Sucrose was added to the substrate as the initial carbon source. The content of sucrose in the substrate was 78.68 g/L and decreased quickly during the whole fermentation process by all three yeasts while ethanol increased simultaneously. Among the three yeast strains, the ethanol content generated by yeast D254 was the highest (Fig. 1c). Therefore, the fermentation ability of D254 was higher when using the ethanol content as an assessment index. In addition, the content of fructose (Fig. 1d) and glucose (Fig. 1e) generated by the three yeast strains increased steadily throughout the time of fermentation from 0 to 32 h but then decreased from 32 to 48 h for the yeast strains D254 and DV10, probably due to the limited sucrose (Fig. 1b) remaining in the substrate during this period (32–48 h); meanwhile, the content of ethanol (Fig. 1c) increased continuously during the 48 h fermentation. However, the yeast strain R-HST was an exception, with a continuing increase of fructose and glucose after 32 h, possibly because sucrose was transformed more slowly in the early stages, and its concentration remained high enough for conversion after 32 h. The changing glucose content showed similar trends with fructose during the whole fermentation process. However, Fig. 1d, e indicate that the glucose content (which decreased by 7.26 and 6.49 g/L in the fermentations with DV10 and D254, respectively) decreased significantly faster than the fructose content (which decreased by 5.14 and 4.07 g/L in the fermentations with DV10 and D254, respectively) in the last 8 h when the sucrose content was extremely low, indicating that glucose is the preferred energy source over fructose for *Saccharomyces cerevisiae* (Xiong et al. 2016). A similar result was reported by Lu et al. (2001) during cucumber juice fermentation with *Saccharomyces cerevisiae*.

Antioxidative properties of the coconut water during fermentation

As shown in Table 1, DPPH, ABTS, FRAP and CUPRAC assays were selected to evaluate the antioxidant capacity of LACW. During fermentation, a wide variety of antioxidant compounds were formed, and many biochemical reactions with antioxidants occurred in the coconut water. These assays have different properties and chemical mechanisms and thus provide a comprehensive evaluation of the antioxidant capacity of LACW (Escudero-LóPez et al. 2013).

Table 1 details the changes of the LACW antioxidant capacity during fermentation for 48 h. The antioxidant capacity assessed by the CUPRAC assay showed an increasing value in all three of the fermentation processes

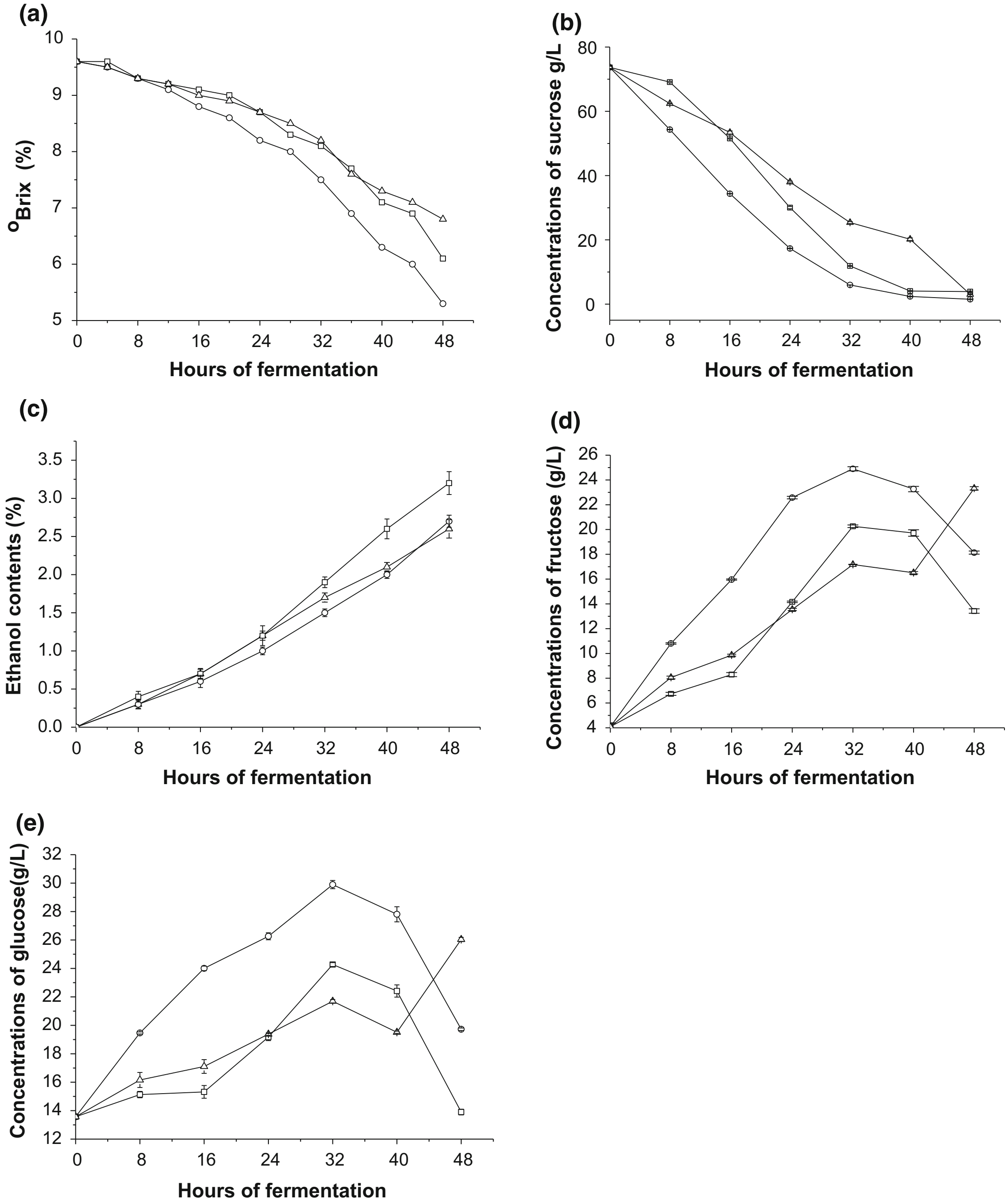

Fig. 1 Changes in the concentrations of fructose, glucose, sucrose and ethanol during fermentation of coconut water with three yeast strains: D254 (square), DV10 (circle) and R-HST (triangle)

(91.77 μM Trolox, R-HST; 93.10 μM Trolox, D254; 94.72 μM Trolox, DV10). This significant ($p < 0.05$) increase in the CUPRAC value was consistent with the results of Jiang and Zhang (2012) during the fermentation of Cabernet Sauvignon and Merlot wines, indicating that alcoholic fermentation had a positive effect on the antioxidant capacity of LACW when assessed by the CUPRAC assay.

The FRAP values of LACW fermented by all of the yeast strains changed slightly in the first 8 h (Table 1) but changed significantly ($p < 0.05$) from 8 to 48 h (from 325.11 to 609.07 μM $FeSO_4$ in R-HST; 326.04–598.10 in D254; 347.29–631.49 in DV10). These results were in accordance with those of Watawana et al. (2016), who reported a significantly ($p < 0.05$) increased FRAP value in king coconut water fermented with kombucha 'tea

Table 1 Antioxidant capacity of LACW fermented by three *Saccharomyces cerevisiae* strains (R-HST, D254 and DV10) during fermentation

Hours of fermentation	CUPRAC (μM Trolox)			FRAP (μM $FeSO_4$)		
	R-HST	D254	DV10	R-HST	D254	DV10
0	60.99 ± 1.57a	58.55 ± 0.51a	58.71 ± 0.88a	318.34 ± 1.46a	321.14 ± 2.83a	324.64 ± 7.42a
8	66.89 ± 0.50b	74.15 ± 2.26b	66.51 ± 0.64b	325.11 ± 2.83a	326.04 ± 4.77a	347.29 ± 20.3a
16	72.54 ± 0.50c	79.68 ± 0.42c	69.85 ± 0.78c	359.67 ± 4.67b	358.27 ± 14.4b	406.38 ± 5.53b
24	79.73 ± 2.12d	87.98 ± 1.06d	72.12 ± 1.71d	448.41 ± 3.46c	425.52 ± 15.9c	467.09 ± 4.59c
32	83.85 ± 0.37e	95.53 ± 1.60e	84.15 ± 0.78e	493.48 ± 1.07d	536.68 ± 10.3d	549.99 ± 3.16d
40	89.83 ± 1.06f	93.58 ± 1.94e	85.94 ± 0.70f	562.37 ± 1.85e	555.13 ± 5.17e	594.36 ± 11.6e
48	91.77 ± 0.96g	93.10 ± 2.17e	94.72 ± 1.13g	609.07 ± 3.53f	598.10 ± 3.90f	631.49 ± 9.96f

Hours of fermentation	DPPH (% Inh[a])			ABTS (% Inh)		
	R-HST	D254	DV10	R-HST	D254	DV10
0	27.55 ± 0.41a	27.63 ± 0.72a	27.67 ± 0.31a	41.24 ± 0.83a	40.82 ± 0.68a	41.17 ± 1.10a
8	32.83 ± 0.65b	35.78 ± 0.28b	34.57 ± 0.80b	55.46 ± 0.74b	47.55 ± 0.29b	57.40 ± 1.43b
16	39.25 ± 0.42c	45.18 ± 0.61c	45.26 ± 0.50c	55.52 ± 1.03b	50.14 ± 0.96c	65.34 ± 0.83c
24	47.81 ± 0.38d	56.03 ± 0.67d	54.30 ± 1.33d	66.06 ± 0.33c	54.88 ± 1.15d	67.39 ± 0.75c
32	54.43 ± 0.32e	67.00 ± 0.13e	56.18 ± 0.46e	72.98 ± 0.29d	64.81 ± 1.11e	76.29 ± 1.17d
40	59.51 ± 0.50f	68.02 ± 0.13e	61.47 ± 0.12f	81.11 ± 0.81e	74.11 ± 2.39f	84.00 ± 1.99e
48	65.16 ± 0.31g	71.53 ± 0.85f	68.81 ± 0.06g	84.79 ± 0.34f	78.46 ± 0.10g	85.68 ± 3.17e

Values are expressed as the mean ± SD. Values with different letters (a–g) in the same column are significantly different at $p < 0.05$

[a] *Inh* inhibition

fungus'. Other authors have also obtained the same results in the fermentation of sea buckthorn juice co-fermented by *Saccharomyces cerevisiae* and *Issatchenkia orientalis* and hawthorn wines fermented by wine yeasts (Negi and Dey 2013).

The DPPH radical scavenging capacity of all fermented samples showed similar trends of increase, from 27% to 65.2, 71.5 and 68.8% for R-HST, D254 and DV10, respectively. Analogously, Escudero-López et al. (2013) reported a significantly increasing trend ($p < 0.05$) for the DPPH value in the fermentation of orange juice. Ayed and Hamdi (2015) also obtained a similar result in cactus pear fermented with 'tea fungus'.

The ABTS radical scavenging capacity of the LACW was doubled by fermentation, reaching 84.79% (R-HST), 78.46% (D254) and 85.68% (DV10). These results agree with those reported by Peerajan et al. (2016), Wu et al. (2011) and Sandhu et al. (2016) in the fermentation of *Phyllanthus emblica* fruit juice, *Graptopetalum paraguayense* and wheat cultivars, respectively. Therefore, fermentation exerts a positive influence on the antioxidant capacity when assessed by the ABTS radical scavenging capacity assay.

The changes in the TPC of LACW during fermentation

The changes in the TPC of the LACW during fermentation are shown in Table 2. The TPC of the fermented samples increased significantly from 32.76 to 52.44 (R-HST), 55.59 (D254) and 51.13 (DV10) μg/mL GAE. These increases were induced by the fungal β-glucosidases, which make β-glucosidic linkages hydrolyzed to mobilizing phenols that are able to react with the Folin–Ciocalteu reagent (Dulf et al. 2016). Other authors have similarly reported that a richer TPC was obtained after alcoholic fermentation. Değirmencioğlu et al. (2016) assessed the phenolic compounds of watermelon, radish, red beet and black carrot juices fermented using *Saccharomyces cerevisiae* and *Saccharomyces boulardii* and concluded that there was an increase in TPC after fermentation. Moreover, the TPC in the fermented products was also influenced by the species of yeast, which was in agreement with the findings of Ivanova-Petropulos et al. (2015). As shown in Table 2, the LACW fermented by yeast D254 contained slightly lower TPC levels than those fermented by yeasts R-HST and DV10, possibly because fewer reducing substances were absorbed by this strain (Ivanova-Petropulos et al. 2015).

Table 2 Changes in the TPC and Correlation coefficients (r^2) between the TPC and antioxidant capacity of LACW fermented by three *Saccharomyces cerevisiae* strains (R-HST, D254 and DV10)

	TPC (μg/mL GAE)[a]							Correlation coefficients (r^2)			
	0 h	8 h	16 h	24 h	32 h	40 h	48 h	CUPRAC/TPC	FRAP/TPC	DPPH/TPC	ABTS/TPC
R-HST	32.77 ± 0.81a	33.2 ± 0.21a	33.94 ± 0.66a	37.26 ± 1.21b	42.52 ± 0.06c	46.24 ± 0.29d	52.44 ± 0.88e	0.924**	0.976**	0.954**	0.931**
D254	32.76 ± 0.21a	35.31 ± 1.62b	35.51 ± 0.26b	40.1 ± 0.06c	48.82 ± 0.71d	52.91 ± 0.41e	55.59 ± 0.15f	0.845*	0.991**	0.943**	0.989**
DV10	32.77 ± 1.11a	34.74 ± 0.88b	38.66 ± 0.80c	42.56 ± 0.06d	45.33 ± 0.51e	51.47 ± 0.58f	51.13 ± 0.63f	0.955**	0.989**	0.969**	0.981**

TPC total phenolic content; *LACW* low alcoholic coconut water

[a] Values are expressed as the mean ± SD. Values with different letters (a–f) are significantly different at $p < 0.05$

Correlation is significant at the following levels: * $p < 0.05$; ** $p < 0.01$

Correlation between the TPC and the antioxidant capacity of the LACW

The correlation between the TPC and antioxidant capacity of LACW is shown in Table 2. The antioxidant capacity of fermentation with R-HST positively and significantly ($p < 0.01$) correlated with the TPC, and the values of the correlation coefficients (r^2) were 0.924, 0.976, 0.954 and 0.931 for the CUPRAC, FRAP, DPPH and ABTS values, respectively. The same trend was also observed in the fermentations with yeast strains D254 ($r^2 = 0.845$, 0.991, 0.943 and 0.989, respectively) and DV10 ($r^2 = 0.955$, 0.989, 0.969 and 0.981, respectively). The increasing antioxidant capacity of the LACW depends on the enrichment of the TPC (Ivanova-Petropulos et al. 2015).

The results in Table 2 indicate that there is a high correlation between the TPC and DPPH capacity, which agrees with the results of Van Leeuw et al. (2014) in wines. Escudero-López et al. (2013) observed the same positive correlation between the TPC and FRAP capacity in fermented orange juice. Similar results of strong correlation between the TPC and ABTS capacity have also been reported by previous authors (Peerajan et al. 2016). In addition, there was also a strong correlation between the four methods for antioxidant capacity themselves, such as the correlation between the CUPRAC and DPPH methods ($r^2 = 0.994$, R-HST; $r^2 = 0.963$, D254; $r^2 = 0.950$, DV10, not shown in the table). This result indicated that the CUPRAC and DPPH methods were almost interchangeable for characterizing the antioxidant capacity (Jiang and Zhang 2012).

The changes in organic acid content in coconut water during fermentation

The changes in organic acids during fermentation are shown in Table 3. The main organic acids of the raw coconut water were oxalic, malic, ketoglutaric, citric and succinic acid. During fermentation, oxalic acid and citric acid were depleted quickly, while tartaric, pyruvic and succinic acid increased significantly. The yeast strains had an important influence on the variable quantity of these acids. Tartaric acid increased from 0.03 to 0.56 g/L (R-HST), 0.30 g/L (D254) and 0.41 g/L (DV10) respectively after fermentation. Pyruvic acid reached 1.02 g/L (yeast R-HST), 0.49 g/L (yeast D254) and 0.92 g/L (yeast DV10) after fermentation from a starting value of 0.02 g/L. Compared to the former two acids, the concentration of succinic acid only increased slightly. Other organic acids, such as malic acid and acetic acid, held relatively steady during fermentation, probably due to the dynamic balance between consumption and generation in the tricarboxylic acid cycle.

Table 3 Organic acids content of coconut water during different fermentation times

Hours of fermentation	Oxalic acid (mg/L)			Tartaric acid (g/L)		
	R-HST	D254	DV10	R-HST	D254	DV10
0	74.6 ± 0.01a	74.6 ± 0.13a	74.6 ± 0.11a	0.03 ± 0.00a	0.03 ± 0.00a	0.03 ± 0.001a
8	23.5 ± 0.20b	10.5 ± 0.11b	7.8 ± 0.09b	0.39 ± 0.00b	0.17 ± 0.00b	0.4 ± 0.001b
16	2.2 ± 0.2c	ND	ND	0.39 ± 0.00b	0.48 ± 0.00f	0.49 ± 0.00c
24	ND	ND	ND	0.41 ± 0.02c	0.34 ± 0.00d	0.73 ± 0.01f
32	ND	ND	ND	0.42 ± 0.00d	0.37 ± 0.00d	0.63 ± 0.021e
40	ND	ND	ND	0.44 ± 0.00e	0.43 ± 0.01e	0.59 ± 0.037d
48	ND	ND	ND	0.56 ± 0.00f	0.30 ± 0.00c	0.41 ± 0.015b
Hours of fermentation	**Pyruvic acid (g/L)**			**Malic acid (g/L)**		
	R-HST	D254	DV10	R-HST	D254	DV10
0	0.02 ± 0.00a	0.02 ± 0.0a	0.02 ± 0.00a	0.23 ± 0.00c	0.23 ± 0.00b	0.23 ± 0.00bc
8	0.02 ± 0.00a	0.02 ± 0.0b	0.10 ± 0.00b	0.29 ± 0.00a	0.10 ± 0.00d	0.27 ± 0.00abc
16	0.17 ± 0.00b	0.08 ± 0.0c	0.29 ± 0.00c	0.25 ± 0.00b	0.01 ± 0.00e	0.24 ± 0.00bc
24	0.36 ± 0.00c	0.21 ± 0.0d	0.54 ± 0.00d	0.20 ± 0.00e	0.20 ± 0.01c	0.34 ± 0.01a
32	0.59 ± 0.00d	0.29 ± 0.0e	0.77 ± 0.01e	0.22 ± 0.00d	0.21 ± 0.01c	0.35 ± 0.03a
40	0.76 ± 0.01e	0.46 ± 0.002f	0.93 ± 0.02f	0.22 ± 0.00d	0.30 ± 0.01a	0.30 ± 0.08ab
48	1.02 ± 0.01f	0.49 ± 0.001g	0.92 ± 0.01f	0.20 ± 0.00e	0.29 ± 0.02a	0.19 ± 0.00c
Hours of fermentation	**Ketoglutaric acid (g/L)**			**Acetic acid (mg/L)**		
	R-HST	D254	DV10	R-HST	D254	DV10
0	1.20 ± 0.00c	1.20 ± 0.00c	1.2 ± 0.00de	0.47 ± 0.01a	0.47 ± 0.01a	0.47 ± 0.00a
8	1.13 ± 0.02d	0.98 ± 0.01f	1.15 ± 0.01e	0.41 ± 0.01b	0.25 ± 0.01e	0.45 ± 0.00a
16	1.19 ± 0.02bc	1.11 ± 0.01d	1.01 ± 0.00f	0.39 ± 0.01c	0.33 ± 0.02b	0.43 ± 0.01a
24	1.06 ± 0.02e	1.17 ± 0.01c	1.22 ± 0.03c	0.29 ± 0.02e	0.30 ± 0.00c	0.42 ± 0.01a
32	1.21 ± 0.01b	1.00 ± 0.01e	1.30 ± 0.02b	0.30 ± 0.00e	0.22 ± 0.00 g	0.44 ± 0.08a
40	1.26 ± 0.02a	1.26 ± 0.01a	1.42 ± 0.01a	0.29 ± 0.00e	0.26 ± 0.00d	0.35 ± 0.00b
48	1.29 ± 0.02a	1.23 ± 0.02b	1.20 ± 0.02ed	0.35 ± 0.00d	0.23 ± 0.00f	0.26 ± 0.00c
Hours of fermentation	**Citric acid (g/L)**			**Succinic acid (g/L)**		
	R-HST	D254	DV10	R-HST	D254	DV10
0	0.23 ± 0.00a	0.23 ± 0.00a	0.23 ± 0.00a	0.28 ± 0.00b	0.28 ± 0.00c	0.28 ± 0.00d
8	0.23 ± 0.00a	0.23 ± 0.00b	0.01 ± 0.00e	0.23 ± 0.01c	0.18 ± 0.02e	0.22 ± 0.00e
16	0.17 ± 0.00b	0.07 ± 0.00c	0.01 ± 0.00de	0.14 ± 0.01d	0.07 ± 0.00f	0.28 ± 0.0d
24	ND	ND	0.01 ± 0.00 cd	0.29 ± 0.00b	0.36 ± 0.01b	0.34 ± 0.00c
32	ND	ND	0.02 ± 0.00c	0.40 ± 0.00a	0.27 ± 0.00d	0.36 ± 0.01b
40	ND	ND	0.06 ± 0.00b	0.43 ± 0.05a	0.35 ± 0.00b	0.38 ± 0.00a
48	ND	ND	ND	0.39 ± 0.00a	0.38 ± 0.00a	0.23 ± 0.02e

Values are expressed as the mean ± SD. Values with different letters (a–g) in the same column are significantly different at $p < 0.05$

Organic acids, except for vitamin C, are generally not related to bioactive properties. However, according to the study released by Tang et al. (2013), malic acid and citric acid help to protect the myocardium and prevent ischemic lesions. Moreover, organic acids also play an important role in promoting flavor, taste and color (Esteves et al. 2004). A small supply of malic acid can improve the fresh sour of fruit wine. During fermentation, tartaric acid can be generated by oxaloacetic acid and degraded into lactic acid and acetic acid by lactobacilli, increasing the content of the total volatile acids (Radin et al. 1994).

Analysis of the volatile quality and composition of coconut water during fermentation

A wide variety of volatile compounds were detected by GC–MS in MCW and LACW, and the results are shown in Table 4. The volatile compounds mainly included alcohols, fatty acids, esters, aldehydes, ketones, phenol and alkanes. There were significant differences in the type and quantity of volatile compounds between the fresh and fermented coconut water, and these differences were largely related to the yeast strains (Renouf et al. 2007). As shown in Table 4, some volatile compounds, such as 2,6,10,14-tetramethy1-hexadecane, 2,6,10,14-tetramethy1-pentadecane, heptadecane and volatile phenol, decreased dramatically ($p < 0.05$). In contrast, some increased significantly ($p < 0.05$), including ethanol, phenylethyl alcohol, octanoic acid and n-decanoic acid, and some remained unchanged, such as 9-octadecanone. These results are possible due to the variety and activity of the enzymes related to the yeast strains (Sumby et al. 2010).

Alcohols were the dominant product in LACW, with a range of 39.46–51.59% (RPA), while the major volatile compounds in MCW were alkanes (66.73%). Ethanol, 1-octanol and 1-hexanol all accounted for a high proportion of the volatile components and were the major production in all fermentations (Table 4). It was noted that small quantities of higher alcohols, such as 1-hexanol and 2-hexyl-1-dodecanol, were only found in the fresh coconut water, probably because these higher alcohols were not part of the fermentation production, or they could be utilized to generate esters with fatty acids, such as hexanoic acid 2-phenylethyl ester and dodecanoic acid ethyl ester. The results corresponded to those reported by Gamero et al. (2016). Moreover, higher alcohols were important precursors for creating esters (Gamero et al. 2016). Generally, the utilization of higher alcohols helped to weaken the negative influence on the volatile qualities of the coconut water; however, excess higher alcohols have been shown to have adverse effects on volatile qualities (Lee et al. 2012). Conversely, some fusel alcohols derived from the Ehrlich pathway during fermentation, such as phenylethyl alcohol, were detected in the fermented coconut water sample but not in the fresh sample (Dashko et al. 2015).

Esters, which depend on enzymes for formation, contributed significantly to the overall volatile profiles of the fermented products (Gamero et al. 2016). As shown in Table 4, fermentations with yeast strains R-HST, DV10, and D254 all showed similar change trends of esters, containing high levels of four ethyl esters (ethyl acetate, octanoic acid ethyl ester, dodecanoic acid ethyl ester and decanoic acid ethyl ester) that contributed to the fruity, pineapple notes, as well as two 2-phenylethyl esters (hexanoic acid 2-phenylethyl ester and octanoic acid 2-phenylethyl ester) that conferred banana, pear or other tropical fruit notes (Gamero et al. 2016). Moreover, the results also revealed that the amounts of ethyl acetate, octanoic acid ethyl ester, dodecanoic acid ethyl ester, decanoic acid ethyl ester and ethyl 9-decenoate were the five largest of the volatile ester group compounds, mainly because the abundant corresponding fatty acids (octanoic acid, dodecanoic acid, decanoic acid and decenoic acid) and alcohols in the fermentation product have the potential to generate these esters. In addition, the formation of these esters is dependent on three factors: acyl-CoA formed from fatty acid metabolism processes (Bardi et al. 1998), alcohols that are either ethanol or higher alcohols derived from amino acid metabolism processes (Saerens et al. 2008) and the corresponding enzymes (Saerens et al. 2006).

Lots of fatty acids are formed during fermentation (Torrens et al. 2008), with values ranging from 13.23 to 22.16% (RPA). Among all fatty acids, medium-chain fatty acids were the main components, including nonanoic acid, octanoic acid, dodecanoic acid, decanoic acid and decenoic acid. In fact, medium-chain fatty acids also contributed to the overall volatile profiles of the fermented products, conferring on them moderate and pleasant notes at lower concentrations (4–10 mg/L) (Liang et al. 2013). Products fermented by yeast strain R-HST produced the highest levels of medium-chain fatty acids, with a value of 22.13% (RPA), while the highest value of decanoic acid was fermented by D254.

Other volatile compounds including aldehydes, ketones, alkanes and volatile phenols were also detected. Some of them, such as ketones, were the products of metabolism during fermentation. In addition, most of them (aldehydes, volatile phenols and alkanes) were utilized.

As shown in Fig. 2, principal component analysis (PCA) was applied to obtain the volatile data of each LACW sample fermented by yeast strains R-HST, D254 and DV10 and to elucidate the differences between their typical volatile profiles. The variables were discriminated by the correlation matrix computed by SPSS 22.0. Thus, 22 volatile compounds with higher concentrations were selected as the parameters, including ethanol, 1-octanol, phenylethyl alcohol, nonanoic acid, octanoic acid, n-decanoic acid, 9-decenoic acid, ethyl acetate and ethyl esters of octanoic acid, nonanoic acid, dodecanoic acid, and decanoic acid.

The PCA of the volatile data showed that the first two dimensions were elected and that the cumulative contribution rate was 100%, indicating that most information from the original data can be explained by the two principal components. Principal component 1 (PC1) accounted for 55.95% of the total sample variance and was closely related to 2-methyl-heptadecane, ethanol, 2-nonanone,

Table 4 Major volatile compounds (peak area × 10^6) and relative peak area (RPA) in LACW fermented by *S. cerevisiae*

	RI	0 h		R-HST (48 h)		DV10 (48 h)		D254 (48 h)	
		Peak area	RPA (%)	Peak area	RPA (%)	Peak area	RPA (%)	Peak area	RPA (%)
Alcohols									
Ethanol	463	1.69 ± 0.05a	1.25	322.43 ± 9.84b	37.38	631.45 ± 12.98d	49.87	551.87 ± 12.52c	44.88
1-Octanol	1059	0.69 ± 0.02a	0.51	1.55 ± 0.01b	0.18	1.98 ± 0.22c	0.16	0.56 ± 0.05a	0.04
Phenylethyl Alcohol	1136	0.00 ± 0.00a	0.00	16.41 ± 1.72b	1.90	19.51 ± 0.89c	1.54	21.41 ± 0.42d	1.72
1-Hexanol	860	0.15 ± 0.03a	0.11	ND	0.00	0.12 ± 0.05b	0.01	0.00 ± 0.00b	0.00
2-hexyl-1-Dodecanol	1989	1.65 ± 0.18a	1.22	ND	0.00	ND	0.00	ND	0.00
n-Nonadecanol	2153	0.68 ± 0.01a	0.50	ND	0.00	0.05 ± 0.04b	0.01	ND	0.00
Subtotal		4.86	3.59	340.39	39.46	653.12	51.59	573.85	46.64
Acids									
Nonanoic acid	1272	0.53 ± 0.01a	0.39	2.13 ± 0.10c	0.25	1.71 ± 0.16b	0.13	1.92 ± 0.12bc	0.16
Octanoic acid	1183	0.18 ± 0.02a	0.13	82.99 ± 3.19b	9.61	94.00 ± 4.04c	7.42	79.99 ± 7.25b	6.51
n-Decanoic acid	1372	0.61 ± 0.03a	0.45	68.63 ± 4.60c	7.95	49.46 ± 1.41b	3.90	105.54 ± 4.67d	8.60
Dodecanoic acid	1580	3.80 ± 0.14b	2.78	4.04 ± 0.14b	0.46	2.13 ± 0.15a	0.17	7.21 ± 0.16c	0.59
9-Decenoic acid	1362	NDa	0.00	33.41 ± 2.91c	3.89	20.62 ± 1.43b	1.61	33.40 ± 1.28c	2.71
subtotal		5.12	3.75	191.20	22.16	167.92	13.23	228.06	18.57
Aldehydes									
Dodecanal	1402	0.13 ± 0.01a	0.10	ND	0.00	ND	0.00	ND	0.00
Decanal	1204	0.67 ± 0.02abc	0.50	0.60 ± 0.02ac	0.06	0.72 ± 0.06a	0.06	0.71 ± 0.04ab	0.06
Undecanal	1303	0.27 ± 0.04a	0.20	0.09 ± 0.02b	0.01	ND	0.00	ND	0.00
Tridecanal	1502	0.57 ± 0.04a	0.42	ND	0.00	ND	0.00	ND	0.00
Subtotal		1.64	1.22	0.69	0.07	0.72	0.06	0.71	0.06
Esters									
Ethyl acetate	586	NDa	0.00	85.59 ± 2.77b	9.94	123.85 ± 8.81c	9.78	77.34 ± 0b	6.29
Octanoic acid, ethyl ester	1183	0.07 ± 0.01a	0.04	30.66 ± 1.21c	3.57	21.78 ± 1.45b	1.72	23.15 ± 0.00b	1.88
Acetic acid, 3,7,11,15-tetramethyl-hexadecyl ester	2119	2.18 ± 0.07a	1.61	0.96 ± 0.04b	0.11	2.29 ± 0.22a	0.18	ND	0.00
Propanoic acid,2-hydroxy,ethyl ester	848	0.55 ± 0.03a	0.40	ND	0.00	ND	0.00	ND	0.00
Methyl 6-methyloctanoate	1118	0.48 ± 0.11a	0.35	ND	0.00	ND	0.00	ND	0.00
Nonanoic acid, ethyl ester	1282	NDa	0.00	0.86 ± 0.07c	0.10	0.31 ± 0,00b	0.02	NDa	0.00
Oxalic acid monoamide N-(2-phenylethyl)-isohexyl ester	2192	NDa	0.00	5.89 ± 1.01c	0.68	NDa	0.00	0.17 ± 0.04b	0.01
Dodecanoic acid ethyl ester	1580	NDa	0.00	1.97 ± 0.20b	0.23	1.95 ± 0.03b	0.15	3.93 ± 0.20c	0.32
Ethyl 9-decenoate	1371	NDa	0.00	16.46 ± 0.40c	1.91	9.47 ± 0.49b	0.74	18.04 ± 0.29d	1.47
Decanoic acid ethyl ester	1381	NDa	0.00	14.26 ± 0.41c	1.65	8.84 ± 0.68b	0.70	24.88 ± 1.26d	2.03
Subtotal		3.27	2.4	156.64	18.19	168.28	13.29	147.91	12
Ketones									
9-Heptadecanone	1847	0.36 ± 0.01a	0.27	0.31 ± 0.02a	0.04	0.93 ± 0.03c	0.07	0.74 ± 0.04b	0.06
9-Octadecanone	1946	0.26 ± 0.00a	0.19	0.25 ± 0.03a	0.04	0.36 ± 0.03c	0.03	0.32 ± 0.02b	0.03
4-Hydroxy-2-methylacetophenone	1363	NDa	0.00	1.93 ± 0.11c	0.22	2.45 ± 0.39d	0.19	1.31 ± 0.08b	0.11
2-Nonanone	1052	NDa	0.00	0.26 ± 0.06b	0.03	0.5 ± 0.04d	0.04	0.46 ± 0.09c	0.04
2-Isobutoxyethyl benzoate	1569	1.48 ± 0.16a	1.09	ND	0.00	ND	0.00	ND	0.00
8-Octadecanone	1946	0.52 ± 0.09a	0.38	ND	0.00	ND	0.00	ND	0.00
Subtotal		2.45	1.93	2.75	0.33	4.25	0.33	2.82	0.24

Table 4 continued

	RI	0 h		R-HST (48 h)		DV10 (48 h)		D254 (48 h)	
		Peak area	RPA (%)	Peak area	RPA (%)	Peak area	RPA (%)	Peak area	RPA (%)
Phenol									
2,4-bis(1,1-dimethylethyl)-Phenol	1555	10.27 ± 0.23a	7.64	3.20 ± 0.16c	0.37	4.78 ± 0.27b	0.38	4.29 ± 0.24b	0.35
Subtotal		10.27	7.64	3.20	0.37	4.78	0.38	4.29	0.35
Alkanes									
Hexadecane	1612	4.44 ± 0.09b	3.27	2.58 ± 0.41a	0.30	5.22 ± 0.19bc	0.41	4.83 ± 0.14c	0.39
2-Methyl-Heptadecane	1746	3.2 ± 0.09b	2.36	0.81 ± 0.09a	0.09	4.73 ± 0.2c	0.37	3.37 ± 0.22b	0.26
2,6,10,14-tetramethy1-Hexadecane	1753	24.32 ± 0.91b	17.92	0.71 ± 0.04a	0.08	30.69 ± 1.66c	2.39	37.99 ± 0.44d	3.09
Heneicosane	2109	6.67 ± 0.09a	4.92	0.72 ± 0.14d	0.08	1.47 ± 0.11c	0.11	2.64 ± 0.11d	0.22
2,6,10,14-tetramethy1-Pentadecane	1653	30.71 ± 2.05b	22.62	15.77 ± 0.11a	1.81	35.41 ± 0.87c	2.80	37.48 ± 0.41c	3.05
Heptadecane	1711	9.45 ± 0.75c	6.97	NDa	0.00	12.11 ± 1.55d	0.96	2.46 ± 0.12b	0.20
Octadecane	1810	1.88 ± 0.46b	1.19	NDa	0.00	1.85 ± 0.04b	0.15	11.65 ± 1.14c	0.95
Nonadecane	1910	1.12 ± 0.02a	0.83	ND	0.00	0.21 ± 0.08b	0.02	ND	0.00
3-methyl-Heptadecane	1746	1.52 ± 0.01c	0.82	0.56 ± 0.04b	0.06	NDa	0.00	1.96 ± 0.22d	0.16
8-hexyl-Pentadecane	2045	1.3 ± 0.02a	0.96	0.12 ± 0.1b	0.01	ND	0.00	ND	0.00
1-pentyl-2-propyl-Cyclopentane	1318	1.14 ± 0.22a	0.84	ND	0.00	ND	0.00	ND	0.00
Subtotal		85.78	62.7	21.72	2.43	91.70	7.21	102.37	8.32

The data were analyzed by DPS V6.55. Values are expressed as the mean ± SD. Values with different letters (a–d) in the same column are significantly different at $p < 0.05$

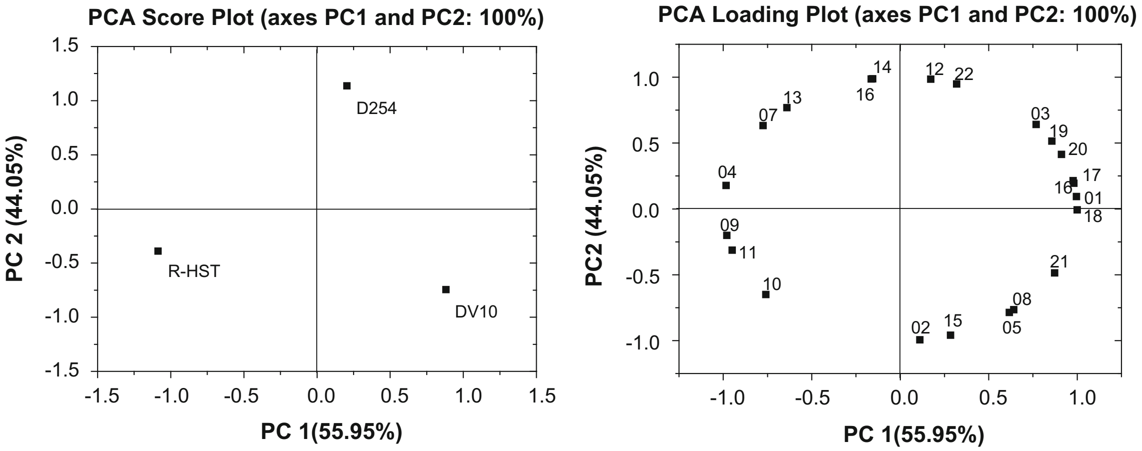

Fig. 2 Bi-plot of principal components analysis of selected volatiles of fresh coconut water and low alcoholic fermentation with *Saccharomyces cerevisiae.* 01) Ethanol; 02) 1-Octanol; 03) Phenylethyl Alcohol; 04) Nonanoic acid; 05) Octanoic acid; 06) n-Decanoic acid; 07) 9-Decenoic acid; 08) Ethyl Acetate; 09) Octanoic acid ethyl ester; 10) Nonanoic acid ethyl ester; 11) Oxalic acid monoamide N-(2-phenylethyl)-isohexyl ester; 12) Dodecanoic acid ethyl ester; 13) Ethyl 9-decenoate; 14) Decanoic acid, ethyl ester; 15) 4-Hydroxy-2-methylacetophenone; 16) 2-Nonanone; 17) Hexadecane; 18) 2-methyl-Heptadecane; 19) 2,6,10,14-tetramethy1-Hexadecane; 20) 2,6,10,14-tetramethy1-Pentadecane; 21) Heptadecane; and 22) Octadecane

hexadecane, 2,6,10,14-tetramethy1-pentadecane, heptadecane and 2,6,10,14-tetramethy1-hexadecane with high positive loading, while nonanoic acid, 9-decenoic acid, octanoic acid ethyl ester, oxalic acid, and monoamide N-(2-phenylethyl)-isohexyl ester showed high negative loading. PC2 accounted for 44.05% of the total sample variance and showed a high correlation with 2-nonanone, dodecanoic acid ethyl ester, octadecane, and 1-octanol (high positive loading), as well as 4-hydroxy-2-methylacetophenone, octanoic acid, ethyl acetate and 9-decenoic acid (high negative loading). As shown from the PCA score plot of Fig. 2, PC1 differentiated the fermentation product of yeast R-HST from those of DV10 and D254 because of the richer concentrations of nonanoic acid, 9-decenoic acid, octanoic acid ethyl ester and oxalic acid, and monoamide N-(2-phenylethyl)-isohexyl ester present in the product from R-HST. The fermentation product from yeast D254 was differentiated by PC2 from that of DV10 because of its richer concentrations of 2-nonanone, dodecanoic acid ethyl ester, octadecane, and 1-octanol.

Conclusion

It is clear that low alcohol fermentation is conducive to improving the quality of MCW, both in aromatic and bioactive properties. The results of the antioxidant capacity assays (DPPH, ABTS, FRAP, CUPRAC) showed that organic acids and total phenols all have a higher value after the fermentation of MCW by *Saccharomyces cerevisiae* R-HST, DV10 and D254.

Comparing the changes among the three yeast strains, fermentation by yeast R-HST had smaller values for TPC and antioxidant capacity and weaker volatile complexity and intensity. Nevertheless, antioxidant capacity and volatile compound profiles showed similar trends in all three fermentations. Therefore, yeast R-HST was comparatively the least suitable for coconut water fermentation.

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Compliance with ethical standards

Conflict of interest The authors report no conflicts of interest. The authors alone are responsible for the content and writing of the paper.

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