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The PHENOLEO project or how to separate and add-value to phenolic compounds present in rapeseed and sunflower meals

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Abstract – Rapeseed and sunflower meal are mainly used as animal feed but they can also be considered as a potential source of bioactive phenolic compounds. However, the desolventization/toasting processes that are needed to produce these meals might influence concentration and chemical structure of phenolic compounds, and change their bioactive properties. Moreover, the recovery processes of these molecules from meals are based on the use of solvent that generates effluents and might affect the integrity of the other constituents of the meals. Knowing this, the PHENOLEO project, funded by the SAS PIVERT, was a research program based on the biorefinery of rapeseed and sunflower meals that aimed to develop new routes of valorization of these materials mostly by the separation and valorization of their simple phenolic compounds. Thus, we decided to focus this study on the impact of the desolventization process on the biochemical composition of meals, the separation process of their simple phenolic compounds, the production of phenolic acids from meals and the potential valorization routes of the phenolic fraction.

Keywords: rapeseed meal / sunflower meal / phenolic compounds / biorefinery

Résumé – Le projet PHENOLEO ou comment séparer et valoriser les composés phénoliques présents dans les farines de colza et tournesol. Les tourteaux de colza et de tournesol sont principalement utilisés pour l’alimentation animale, mais ils peuvent également être considérés comme une source potentielle de composés phénoliques aux propriétés bioactives. Cependant, les procédés de désolvantation/toastage qui sont nécessaires pour produire ces tourteaux pourraient influencer la concentration et la structure chimique des composés phénoliques induisant une possible modification de leurs propriétés bioactives. De plus, les procédés de récupération de ces molécules à partir de ces tourteaux reposent sur l’utilisation de solvants qui génèrent des effluents et pourraient affecter l’intégrité des autres constituants contenus dans ces matières premières. Dans ce contexte, le projet PHENOLEO, financé par la SAS PIVERT, était un programme de recherche basé sur la bioraffinerie des tourteaux de colza et tournesol visant à développer de nouvelles voies de valorisation de ces matières premières en réalisant la séparation et la valorisation de leurs composés phénoliques simples. Ainsi, cette étude a focalisé sur l’impact des procédés de désolvantation sur la composition biochimique des tourteaux, les procédés de séparation des composés

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phénoliques simples présents dans les tourteaux, la production d’acides phénoliques par voie enzymatique et leurs valorisations potentielles.

**Mots clés :** tourteau de colza / tourteau de tournesol / composés phénoliques / bioraffinage

### 1 Introduction

Rapeseed (*Brassica napus* L.) and sunflower (*Helianthus annuus* L.) are two oleaginous plants grown primarily to produce vegetable oil which is mainly used for human consumption or for biofuel production (Borredon *et al.*, 2011; Fine *et al.*, 2015). Moreover, the main co-products resulting from the crushing, de-oiling and desolventization/toasting processes of these seeds are their meals. In 2019, worldwide production of rapeseed meal (RSM) and sunflower meal (SFM) were estimated at 33.0 and 22.2 Mt, respectively (http://www.indexmundi.com/agriculture/). These heterogeneous raw materials are mainly used as animal feed because of their high protein content which is around 38% DM (dry matter) for RSM, 31% DM for SFM and 39.5% DM for dehulled SFM (González-Pérez and Vereijken, 2007; Ivanova *et al.*, 2016, Terres Inovia, 2019). However, RSM and SFM also contain interesting bioactive molecules such as phenolic compounds (PC) which are present as simple and extractable PC, mainly located in the kernel or as hardly extractable or bound polyphenols, concentrated in the hull (Amarowicz *et al.*, 2000; Weisz *et al.*, 2009; Pajak *et al.*, 2014; Laguna *et al.*, 2018, Fig. 1). The total simple phenolic compound (TSPC) content in RSM and SFM is around 2% DDM (defatted dry matter) and 4% DDM, respectively (Baumert *et al.*, 2005; Weisz *et al.*, 2009). Moreover, regarding the simple PC present in RSM, they are mainly sinapic acid esters with sinapine, the choline ester of sinapic acid, as the main ester that represents around 80% of the TSPC content (Baumert *et al.*, 2005; Milkowski and Strack, 2010; Siger *et al.*, 2013). Furthermore, the major bound PC in rapeseed hulls are mostly tannins and their content may exceed 2 g/100 g of defatted dry hulls (Amarowicz *et al.*, 2000). On the other hand, in SFM, simple PC are mainly caffeic acid esters with 5-cafeoylquinic acid (or chlorogenic acid, the quinic ester of caffeic acid) accounting for around 70% of the TSPC content (Weisz *et al.*, 2009; Szydlowska-Czerniak *et al.*, 2011; Guo *et al.*, 2013). There is no much literature data regarding the bounded PC in sunflower hulls but, in the whole seed, they are mainly flavonoids and they account for around 4.3 μg/g DM (Pajak *et al.*, 2014).

These naturally occurring molecules are known to be bioactive and multifunctional compounds exhibiting antioxidan (Dimitrios, 2006; Kreps *et al.*, 2014), anti-UV (Peyrot *et al.*, 2020) and antimicrobial activities (Ouerghemmi *et al.*, 2017) to name a few. Knowing this, they can find applications directly or after modifications in many fields such as in food, health and cosmetic products (Kikuzaki *et al.*, 2002; Shahidi and Ambigaipalan, 2015). For example, previous studies have shown the protective effect of ferulic acid for the preservation of some food matrices (e.g. oranges, biscuits, soybean oil, etc.) which is mainly associated with its antioxidant properties (Kumar and Pruthi, 2014). Furthermore, Lin *et al.* (2005) reported the antioxidant and anti-UV properties of ferulic acid and demonstrated that its association with vitamin E and vitamin C in a topical formulation provided about 4 to 8 fold higher protection against solar-simulated radiation damage. Regarding this matter, recent studies have also suggested that some sinapic acid derivatives, such as dehydrodiethylosinapate (Horbury *et al.*, 2020) or sinapic-acid-based octinoxate analogues (Peyrot *et al.*, 2020), are good candidates to be used as UV filters and antioxidants in sunscreen lotions. On the other hand, some phenolics can also be used as precursors of polymers for food packaging. Indeed, Reano *et al.* (2015), Diot-Néant *et al.* (2017) and Hollande *et al.* (2018) demonstrated that different biobased bis-aryl esters of dihydroferulic acid or dihydrosinapinic acid could be used as monomers in various types of polymerization systems with the resulting polymers displaying high antioxidant activities and high thermal stabilities. In addition, Aouf *et al.* (2016) showed that canolol (2,6-Dimethoxy-4-vinylphenol, the decarboxylation product of sinapic acid) could be used as the precursor of bio-based epoxy resins prepolymers.

In this context, RSM and SFM could be valorized and used as a source of bioactive phenolic compounds. However, the desolventization/toasting process that are needed to produce the meals might impact concentration and chemical structure of these molecules, and change their bioactive properties (Zago *et al.*, 2015b). Moreover, phenolic compounds can oxidize in RSM and SFM, enzymatically or in an alkaline medium, and form o-quinones and phenolic radicals that rapidly evolve towards complex oxidation products. Quinones and radicals can also react with the amino acids of the meal proteins, causing browning and changes in their organoleptic characteristics and applications, it is necessary to partially or totally separate the most possible natural compounds, plus the risks associated with the handling of formaldehyde during tanning favored the research for alternative tanning agents of vegetable origin. Thus, phenolic compounds present in RSM and SFM could also be valorized in the form of tanning agents.

Hence, in order to add-value to all the constituent present in these oleaginous meals either for food/feed or chemical applications, it is necessary to partially or totally separate the phenolic compounds beforehand. Thus, the PHENOLEO project, funded by the SAS PIVERT, was a research program based on the biorefinery of rapeseed and sunflower meals that aimed to develop new routes of valorization of these meals mostly by the separation and valorization of their simple phenolic compounds. Indeed, the separation of these molecules could allow increasing the nutritional qualities of...
meal’s proteins and concomitantly valorized the phenolic fraction. Hence, we decided to focus our study on (i) the impact of the desolventization process on the biochemical composition of the meals, (ii) the separation process of the simple phenolic compounds from the meals and (iii) the production of phenolic acids from the meals and (iv) the potential valorization routes of the phenolic fractions.

(i) Conventionally, toasting process using live steam allows the removal of the residual solvent in meals after the solvent assisted oil extraction but also induced the deactivation or elimination of some antinutritional factors such as endogenous enzymes and glucosinolates (Salazar-Villanea et al., 2016; Wildermuth et al., 2016). However, these thermal treatments can also reduce the nutritional qualities of proteins (Salazar-Villanea et al., 2016, 2017) and modified the bioactive properties and concentration of simple phenolic compounds (Zago et al., 2015b). Thus, during this project, we produced different rapeseed and sunflower samples (meals, kernels, hulls) under different desolventization/toasting conditions to study the impact of the process on different biochemical parameters (protein content, protein solubility, total simple phenolic compounds content (TSPC), complex polyphenolic content). (ii) Then, we were interested in the development of novel and green processes for the separation of simple phenolic compounds from the different meals, including novel dry fractionation processes or solvents assisted extractions with hydro-alcoholic mixtures. (iii) Afterwards, the enzymatic release of sinapic acid (SA) and caffeic acid (CA) from RSM and SFM, respectively, with different carboxylic esters hydrolases from Aspergillus niger was evaluated. (iv) Finally, the potential valorization routes of phenolic acids (either in pure form or after chemical modification) and some interesting fractions produced after the separation steps were evaluated to be used as antioxidants or tanning agents, respectively.

The results presented in the following sections are from several articles, either already published or in preparation for others. Readers are therefore invited to refer to the corresponding papers for more information and details.

2 Biochemical composition of the different raw materials used in the PHENOLEO project

During this project, different RSM and SFM samples were produced under different conditions (Laguna, 2019, Fig. 2). Firstly, the whole rapeseed and sunflower seeds were crushed and de-oiled at room temperature to produce a cake. Then, the residual lipids of the produced cakes were extracted by hexane under the conditions described in Figure 2A. Furthermore, to remove hexane in meals after the oil extraction, a desolventization step was performed under mild conditions (−0.87 bar, > 60 °C) to produce an untoasted rapeseed meal (U-RSM) and an untoasted sunflower meal (U-SFM) or, after desolventization, meals were toasted in
harsh conditions (114°C, 60 min with an injection steam for 5 min) similar to the ones used at industrial scale, to produce a toasted rapeseed meal (T-RSM) and a toasted sunflower meal (T-SFM). Afterwards, different biochemical parameters were determined to evaluate the impact of the different processes performed on meal samples.

2.1 Proteins, simple phenolic compounds and glucosinolate contents

The protein content obtained for all RSM and SFM samples were around 38% DDM and 32% DDM respectively (Tab. 1) and they were in good agreement with literature data (González-Pérez and Vereijken, 2007; Carré et al., 2016).
Thus, these results clearly demonstrated that extreme temperatures used during the toasting step did not change the protein content of the meals. Moreover, this recalcitrant aspect of proteins has already been observed in other studies where it was demonstrated that desolventization/toasting steps applied on RSM did not affect its protein content (Mosenthin et al., 2016; Salazar-Villanea et al., 2016).

However, either for RSM or SFM samples, a loss of protein solubility in soda (34.7 and 3.7% respectively) was observed after the toasting step (Tab. 1). This can be explained by the possible modification of techno-functional properties of proteins due to the harsh implemented conditions (Salazar-Villanea et al., 2016). Indeed, according to Salazar-Villanea et al. (2017), reduction of protein solubility in RSM is linked to physical (aggregation phenomenon) and chemical (Maillard reactions) modifications of their structures, which lead to a decrease of their digestibility and solubility. Furthermore, these modifications reduce the uses of these meals in feed applications. Regarding this matter, mild desolventization conditions are more suitable for the removing of residual solvent (after the solvent-assisted oil extraction step), while reducing the deterioration of protein quality in meals.

The total simple phenolic compound (TSPC) of RSM and SFM samples was determined by HPLC-DAD-MS, retention time of authentic standards or literature data as described by Laguna et al. (2018) and results can be seen in Tables 2 and 3. For U-RSM, nineteen different phenolic compounds were identified (Tab. 2). The main phenolic compound was sinapine (SNP), the choline ester of sinapic acid, accounting over 75% of TSPC content. The other compounds identified were trans-sinapinic acid (SA) and sinapic acid esters comprising sugar (glucose, gentobioside) and/or flavonoid (kaempferol) moieties. For U-SFM, eleven different phenolic compounds were identified (Tab. 3), the main being the 4- and 5-O-caffeoylquininic acids (4- and 5-CQA) which were co-eluted and quantified together (68% of all the phenolics). Furthermore, 3-O-caffeoylquinic and di-caffeoylquinic acid isomers were also detected. These results were in good agreement with literature data (Weisz et al., 2009; Fang et al., 2012; Carré et al., 2016).

### 2.2 Individual simple phenolic compounds

The individual phenolic compounds of U-RSM and U-SFM were identified by HPLC-DAD-MS, retention time of authentic standards or literature data as described by Laguna et al. (2018) and results can be seen in Tables 2 and 3. For U-RSM, nineteen different phenolic compounds were identified (Tab. 2). The main phenolic compound was sinapine (SNP), the choline ester of sinapic acid, accounting over 75% of TSPC content. The other compounds identified were trans-sinapinic acid (SA) and sinapic acid esters comprising sugar (glucose, gentobioside) and/or flavonoid (kaempferol) moieties. For U-SFM, eleven different phenolic compounds were identified (Tab. 3), the main being the 4- and 5-O-caffeoylquininic acids (4- and 5-CQA) which were co-eluted and quantified together (68% of all the phenolics). Furthermore, 3-O-caffeoylquinic and di-caffeoylquinic acid isomers were also detected. These results were in good agreement with literature data (Weisz et al., 2009; Khattab et al., 2010).

### 2.3 Complex bound polyphenolic compounds

Bound polyphenolic compounds still remain largely unexplored because they are difficult to extract in regular solvents. They likely correspond to oxidized condensed tannins in more or less polymerized forms and potentially exhibiting covalent linkages with macromolecules of the hulls (i.e. cell-wall complex polysaccharides and proteins) as it was mentioned in previous work (Auger et al., 2010). Knowing this, we performed the analysis of bound polyphenolic compounds in rapeseed and sunflower untoasted hulls.
Firstly, butanol-HCl assay was applied following the Porter’s reaction conditions (Porter et al., 1985). Considering sunflower hulls, no red coloration of the reaction medium was observed suggesting that this material do not contain any condensed tannins. In contrast, rapeseed hulls butanol-HCl reaction medium exhibited a pronounced red coloration that clearly indicated the presence of condensed tannins.

Secondly, rapeseed hulls were submitted to the phloroglucinolysis reaction coupled to reversed phase HPLC analysis according to the method of Kennedy and Jones (2001). The HPLC-UV-MS analysis of the pholoroglucinolysis medium unambiguously revealed the presence of the flavanyl epicatechin-phloroglucinol adduct at m/z 413 confirming the presence of condensed tannins (i.e. procyandin oligomers and/or polymers) as constituent of rapeseed hulls. In order to get optimized quantitative data related to those condensed tannins, the phloroglucinolysis reaction conditions were optimized for rapeseed hulls for incubation temperature, reaction duration and hydrochloric acid concentration applying an experimental design processed by response surface methodology. The response surface methodology showed that the maximum of 5.4 g of procyanidins/kg of rapeseed U-hulls was predicted with 0.8 M of HCl at 60°C during 30 min incubation (Unpublished results). Interestingly, the analysis of the phloroglucinolysis reaction media by LC/MS and LC/MS/MS allowed the identification of some oxidation markers of tannins. These molecules were detected on the full MS chromatogram by their molecular ions at m/z 699 and the corresponding product ions consistent with the presence of an intramolecular A-type bonds between two adjacent “epicatechin” units in the overall structure of the tannins as already detected in the case of oxidized apple procyanidins (Millet et al., 2019). On the other hand, the phloroglucinolysis method did not allow the identification of any polyphenolic compound in the U-hulls of SFM, indicating that phenolics are mainly present in the form of simple phenolic located in the kernel.

Finally, after the determination of the different biochemical parameters of the different meals, we were interested in the separation of their simple phenolic compounds by applying different separation methodologies, such as dry fractionation processes or solvent assisted extraction with hydro-alcoholic mixtures.

### Extraction of the simple phenolic compounds present in the untoasted meals

#### 3.1 Dry fractionation processes of the untoasted meals

Dry milling combined with dry fractionation technologies are new alternative processes that allow the separation of

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Table 2. MS data of phenolic compounds detected in U-RSM methanolic extract (from Laguna et al., 2018).

| Compound | λ max | [M−H]− m/z | [M+H]+ m/z | Name |
|----------|-------|------------|------------|------|
| 1        | 328   | 294        | 310        | Sinapine |
| 2        | 328   | 771        | n.d.       | Kaempferol-3-O-sophoroside-7-O-b-glucopyranoside |
| 3        | 332   | 385        | 207        | Sinapoyl glucopyranoside |
| 4        | 333   | 977        | 979        | Sinapoyl-Kaempferol derivative 1 |
| 5        | 327   | 520        | n.d.       | n.d. |
| 6        | 323   | 547        | 665        | Cyclic spermidine |
| 7        | 307   | 494        | 496        | Sinapoyl-Kaempferol derivative 2 |
| 8        | 328   | 237        | 207        | Trans-sinapic acid |
| 9        | 323   | 223        | 207        | Cis-sinapic acid |
| 10       | 323   | 223        | 207        | Sinapoyl-di-hexoside-sinapoyl-hexoside |
| 11       | 332   | 977        | 979        | Disinapoyl gentiobioside isomer 1 |
| 12       | 330   | 753        | 777        | Disinapoyl gentiobioside isomer 2 |
| 13       | 328   | 977        | 979        | Sinapoyl-Kaempferol derivative 3 |
| 14       | 330   | 591        | 369        | Disinapoyl glucopyranoside |
| 15       | 327   | n.d.       | n.d.       | n.d. |
| 16       | 326   | 959        | 737        | Trisinapoyl gentiobioside isomer 1 |
| 17       | 328   | 959        | 737        | Trisinapoyl gentiobioside isomer 2 |
| 18       | 328   | 959        | 737        | Trisinapoyl gentiobioside isomer 3 |

Note: U-RSM = untoasted rapeseed meal.

Table 3. Phenolic compounds detected in U-SFM methanolic extract (from Laguna et al., 2018).

| Compound | λ max | Name |
|----------|-------|------|
| 1        | 327   | 3-O-cafeoylquinic acid |
| 2        | 326   | 4-O- and 5-O-cafeoylquinic acid |
| 3        | 323   | Caffeic acid |
| 4        | 327   | n.d. |
| 5        | 327   | n.d. |
| 6        | 327   | 5-O-p-coumaroylquinic acid |
| 7        | 329   | 5-O-feruloylquinic acid |
| 8        | 326   | 3,4-di-O-cafeoylquinic acid |
| 9        | 328   | 3,5-di-O-cafeoylquinic acid |
| 10       | 327   | n.d. |
| 11       | 328   | 4,5-di-O-cafeoylquinic acid |

Note: U-SFM = untoasted sunflower meal.
certain macromolecules of interest from different agricultural materials without the use of solvents. These dry fractionation processes emerged as eco-friendly technologies suitable for the concentration of proteins, cellulose, lignin and polyphenols with retained (native) functionality. Knowing this, we tried to separate the simple phenolic compounds from proteins in U-RSM and U-SFM by performing dry fractionation processes (Laguna et al., 2018).

3.1.1 Effect of the ultrafine milling on the untoasted meals composition

Firstly, the untoasted meals were milled by shear and impact using an UPZ miller device. Furthermore, to obtain different particles sizes, three different grid were used: 0.5 mm, 0.3 mm and 0.1 mm (0.2 mm for the U-SFM) and the different ultrafine meals produced were named U-RSM-“size of the grid” and U-SFM-“size of the grid”. As expected, the effect of the milling step was different between U-RSM and U-SFM. Indeed, the reduction of the grid size was accompanied by a slight, but significantly, increase by 4% in the protein content in U-RSM-0.1 (39.5 ± 0.3% DDM, Tab. 4) but this increase was particularly higher in the case of U-SFM-0.2 (11% increase, 35.0 ± 0.0% DDM, Tab. 4). This was explained by a loss of hull particles during the milling step by a fouling effect. Besides, the loss of the hull fraction in U-SFM-0.2 also explained it low recovery yield (88.8 ± 0.1% w/w, Tab. 4) compared to the other meals (around 96% w/w, Tab. 4). Finally, it was noticed that the total simple phenolic compound (TSPC) values of the different U-RSM and U-SFM samples were not modified after the milling step (around 16 mg SAE/g DDM and 24 mg CAE/g DDM, respectively, Tab. 4).

3.1.2 Turbo-separation of the ultrafine untoasted rapeseed and sunflower meals

The samples previously milled were then subjected to a turbo-separation step which allows particles separation according to their size and density. Moreover, three different classifier wheel speeds (3000, 5000 and 7000 rpm) were tested and after each separation step, two different fractions were recovered: a fine fraction named FF and a coarse fraction named CF (Fig. 3A). Firstly, we observed, either for U-RSM or U-SFM samples, that proteins and phenolic compounds were separated together in the same fractions. However, in the case of U-RSM samples, we noticed that protein and TSPC content were similar in the different FF obtained, indicating that there was no qualitative influence of the particle size and the speed of the classifier wheel on the purity of protein/phenolics (Figs. 4A and 4B). However, the best overall recovery yields were obtained in FF fractions from U-RSM-0.1 (Fig. 4C) and the highest protein and TSPC content was obtained at 5000 rpm (48.7 ± 0.8% DDM and 21.4 ± 0.3 mg of sinapic acid equivalents (SAE)/g DDM, respectively, Tab. 5) with a recovery yield of 40.7 ± 0.6% w/w (Tab. 5). On the other hand, regarding the fine U-SFM, the higher protein and TSPC content were obtained in FF fractions of U-SFM-0.5 (Figs. 4D and 4E). Also, it is worth mentioning that turbo-separation of U-SFM-0.2 was ineffective because particles were too small and were all recovered in FF (data not shown). The best compromise between purity (in terms of protein/phenolic content) and recovery yield was achieved at 5000 rpm with an increase by 67.4% in proteins (51.2 ± 0.3 g/100 g DDM, Tab. 5), 53.3% in TSPC (39.9 ± 1.1 mg of chlorogenic acid equivalents (CAE)/g DDM /g DDM, Tab. 5), 53.3% in TSPC (39.9 ± 1.1 mg of chlorogenic acid equivalents (CAE)/g DDM /g DDM, Tab. 5), and a recovery yield of 30.0% (Tab. 5, Fig. 4F). Moreover, protein and TSPC content of FF of U-RSM-0.1 and U-SFM-0.5 were equivalent to the values determined in their respective U-kernels (Tab. 5), demonstrating that turbo-separation process allows the production of fine fractions concentrated in kernel particles. Hence, we assumed that in U-RSM-0.1 and U-SFM-0.5 particles enriched with protein/phenolic were dissociated from hull particles during the milling step and were more easily separated during turbo-separation.

3.1.3 Electrostatic sorting of the ultrafine untoasted rapeseed and sunflower meals

The fine meals were also subjected to a single step electrostatic sorting which allows the separation of differently charged or polarized particles under the action of an electric field. Moreover, at the end of the separation step, four different fractions were recovered: a positive fraction (PFe) and a negative fraction (NFe) recovered on the electrodes and a positive fraction (PFc) and a negative fraction (NFc) collected on the jars under the electrodes (Fig. 3B). Regarding proteins and phenolics contents, we noticed that they were again separated in the same fractions, indicating that electrostatic sorting was not able to segregate these molecules. However, regarding U-RSM, the highest increase of 50% in protein (59.1 ± 0.4 g/100 g DDM) and 55% in TSPC (25.8 ± 0.1 mg SAE/g DDM) content was obtained in PFe recovered from U-RSM-0.1 (Tab. 5, Figs. 5A and 5B). On the other hand,

Table 4. Size, yield and composition of U-RSM and U-SFM samples obtained by milling with different grids (from Laguna et al., 2018).

| Grid size (mm) | U-RSM | U-SFM |
|---------------|-------|-------|
|               |       |       |
| Recovery yield (% w/w) | 96.0 ± 0.6^a | 97.0 ± 0.2^a |
| Protein content (% DDM) | 37.9 ± 0.2^b | 31.3 ± 0.0^b |
| TSPC^c | 15.6 ± 0.6^d | 25.3 ± 0.4^e |
| D50 (μm) | 110.9 ± 9.1^f | 105.5 ± 8.3^g |

Note: TSPC: total simple phenolic compound. *: TSPC was expressed in mg of sinapic acid equivalent (SAE)/g of defatted dry matter (DDM) and mg of chlorogenic acid equivalent (CAE)/g DDM for U-RSM and U-SFM respectively. For each raw material, values followed by same superscript letters are not significantly different, p ≤ 0.05. Values are mean ± SD (n = 3).
for U-SFM, the highest increase of 96% in protein (61.1 ± 0.0 g/100 g DDM) and 80% in TSPC (45.8 ± 1.1 mg CAE/g DDM) content was obtained in PFe obtained from U-SFM-0.5 (Tab. 5, Figs. 5C and 5D). Moreover, it is worth mentioning that results obtained after U-SFM-0.2 electrostatic sorting were not taken into account because of the loss in hulls observed during the milling step (Tab. 4). Furthermore, the protein and TSPC content of PFe of U-RSM-0.1 and U-SFM-0.5 were higher than the values determined from their corresponding U-kernel (Tab. 5). Hence, we assumed that the protein bodies associated with phenolic compounds from kernels in U-RSM-0.1 and U-SFM-0.5 were more easily charged and concentrated in the respective PFe fraction after electrostatic sorting. On the other hand, to enhance the low recovery yield of U-RSM-PFe-0.1 and U-SFM-PFe-0.5 (around 9 and 18% respectively), multiple recycling steps of the collected fractions (PFc and NFe) were done. Finally, the cumulated recovery yield of U-RSM-PFe-0.1 and U-SFM-PFe-0.5 increases from 9 to 31% w/w after four cycles and from 18 to 32% w/w after three cycles, respectively (data not shown).

Here, we showed that electrostatic sorting allowed the recovery of fractions with higher purity in terms of proteins and phenolic compounds compared to turbo-separation. Nevertheless, it can be concluded that the dry fractionation
processes are not able to individually separate phenolic compounds from meals. However, dry fractionation processes can be used as a pre-purification step of protein and phenolic compounds from the other constituents of the meals. Finally, protein and phenolic compounds enriched fractions obtained in this work might also be used to different ends. Indeed, as already pointed out by other authors, the associations of proteins and phenolic compounds might act as emulsifiers displaying strong antioxidant activities.

### 3.2 Hydro-alcoholic assisted extraction of the simple phenolic compounds from the untoasted meals

Regarding the extraction of simple phenolic compounds from RSM and SFM, different studies have shown that methanol is the best solvent for their extraction (Pedrosa et al., 2000; Cai and Arntfield, 2001; Weisz et al., 2009). However, methanol is also considered as toxic and non-suitable for food and feed uses. Therefore, we decided to explore the use of more environmentally friendly mixtures such as water-ethanol and water-isopropanol for the extraction of simple phenolic compounds present in U-RSM and U-SFM (Laguna, 2019). Moreover, as U-RSM-0.1 and U-SFM-0.5 allowed the best separation in Section 2.1, these two materials were selected for this purpose.

Hence, simple phenolic compounds were extracted with mixtures of ethanol or isopropanol containing different amounts of water (5–25% v/v). Regarding U-RSM-0.1, either for ethanol or isopropanol, the extraction of simple phenolic compounds was enhanced with increasing the water concentration until a maximum of 15% was reached (Fig. 6A).
As a result, the TSPC values obtained with 85% ethanol and 85% isopropanol were of 16.3 ± 0.9 mg SAE/g DDM and 16.1 ± 1.6 mg SAE/g DDM respectively. Furthermore, these results were in the same range of the TSPC value obtained with pure methanol (16.6 ± 0.3 mg SAE/g DDM).

However, for U-SFM-0.5, the higher TSPC content of 23.7 ± 0.1 mg CAE/g DDM was obtained with ethanol at a water concentration of 20% (Fig. 6B) and this value was slightly lower than the TSPC content obtained with pure methanol (25.3 ± 0.4 mg CAE/g DDM, Tab. 4). Finally, regarding the extraction with isopropanol, a maximum was never reached whatever the water concentration tested in this study and the highest TSPC content of 21.5 ± 0.1 mg CAE/g DDM was obtained with 25% of water.

Thus, these results showed the efficiency of different hydro-alcoholic mixtures for the recovery of simple phenolic compounds present in U-RSM and U-SFM. Moreover, knowing that ethanol is a green bio-based solvent, their hydro-alcoholic mixtures could be considered as the best choice for the extraction of these bioactive molecules. However, adding water might also lead to the extraction of other undesirable compounds such as minerals, peptides or sugars, leading to tedious and complex purification steps for the recovery of pure phenolic compounds. Thus, it is of paramount importance to determine the specificity of these solvents towards other molecules present in the meals before concluding on their efficiency. Furthermore, knowing that the use of ethanol in RSM altered its protein solubility and amino acid composition (Kalaydzhiev et al., 2020), it is necessary to study the impact of the process presented in this work on the quality of the protein fraction in U-RSM and U-SFM.

4 Enzymatic treatments of RSM and SFM samples to produce phenolic acids

It has been well documented in numerous studies that phenolic acids have interesting bioactive properties, such as antioxidants, anti-UV and antimicrobial activities (Shahidi and Ambigaipalan, 2015; Ouerghemmi et al., 2017; Gullón et al., 2018). Moreover, phenolic acids could also be used for the production of new additives or polymers precursors (Aouf et al., 2012; Zago et al., 2015a; Hollande et al., 2018).

Consequently, rapeseed and sunflower meals could be advantageously used as sources of phenolic acids. Regarding this matter, we aimed to produce sinapic acid (SA) and caffeic acid (CA) from RSM and SFM samples, respectively, by performing enzymatic treatments with various fungal carboxyl ester hydrolases from recombinant Aspergillus niger strains (Laguna, 2019; Laguna et al., 2019). For SFM samples, we tested an A. niger type-B recombinant feruloyl esterase (AnFaeB) and a recombinant A. niger chlorogenate esterase (ChlE) because of their hydrolytic activity on caffeic acid esters (CAEsters) (Ramos-de-la-Peña and Contreras-Esquível, 2016). On the other hand, A. niger type-A recombinant feruloyl esterase (AnFaeA) was used for the hydrolysis of sinapic acid esters (SAEsters) in RSM samples because of their hydrolytic activity regarding these molecules (Faulds, 2010; Gopalan et al., 2015). Moreover, enzymatic treatments were done either directly on crude meals or on their dry methanolic extract, mainly composed of simple phenolics compounds, minerals and carbohydrates (Sripad and Narasininga Rao, 1987) but devoid of proteins. Furthermore, we used both toasted and untoasted meals because, as pointed out before, untoasted meals have a higher phenolic content than the corresponding toasted ones (Tab. 1), while thermal treatments performed during the toasting step might have deactivated endogenous enzymes that could disturb enzymatic hydrolysis.

4.1 Release of free caffeic acid from toasted SFM (T-SFM), untoasted SFM (U-SFM) and its methanolic dry extract with AnFaeB and ChlE

The enzymatic treatments were performed under optimal pH (6.0), temperature (50°C) and enzyme concentration (30 nkat/g and 10 nkat/g for AnFaeB and ChlE, respectively) for the complete hydrolysis of all the CAEsters present in sunflower samples. Moreover, the total simple phenolic content was expressed as total caffeic acid derivatives (TCAD) content since 95% of SFM phenolics were found as caffeic acid esters (Weisz et al., 2009; Karamać et al., 2012).

Regarding the use of AnFaeB in T-SFM, it allowed an 100% hydrolysis yield after 3 h incubation (Tab. 6, Supplementary Material Fig. S1A). On the other hand, the highest hydrolysis yield obtained with ChlE was 90% after 4
incubation (Tab. 6). Compared to AnFaeB, this lower value of free CA released by ChlE was mainly due to the incomplete hydrolysis of 4-CQA and 5-CQA and the complete inability to hydrolyse 3-CQA (Supplementary Material Fig. S1B). Finally, either with AnFaeB or ChlE, the TCAD content was unchanged during the different incubation times (around 58 μmol of caffeic acid derivatives (CaAE)/g DDM, data not shown).

Surprisingly, a strong decrease in the TCAD content over time (>30%) was observed with or without enzyme (control) in U-SFM (data not shown). This decrease was attributed to the presence of endogenous polyphenol oxidases (PPO, 1.02 ± 0.08 nkat/mL dipheno oxidase activity and 0.43 nkat/mL monophenol oxidase activity) in this untoasted meal that degraded CA and its derivatives during the pre-incubation and incubation steps. This constituted a major drawback that greatly limited the value of U-SFM as a substrate for in situ enzymatic production of CA.

To overcome this issue, the enzymatic treatment was performed on its dry methanolic extract. Here, all CAEsters were hydrolysed into free CA by AnFaeB after only 10 min, leading to a 95% hydrolysis yield (Tab. 6). This result was slightly lower than the 100% hydrolysis yield obtained in T-SFM. However, the net higher initial TCAD content in U-SFM led to a higher free CA production compared to T-SFM (68.1 ± 1.7 and 59.8 ± 2.1 μmol/g DDM, data not shown).

The maximum hydrolysis yield of around 70 and 78% was obtained after 240 min incubation for T-RSM and U-RSM respectively (Tab. 7) and almost all sinapine, mono- and di-sinapoyl glucose isomers were hydrolysed at the end of incubation (Supplementary Material Fig. S2A). Furthermore, regarding the initial TSAD content present in U-RSM, a loss of 16% was observed after the 30 min pre-incubation step (before adding the enzyme in the medium, Tab. 7). Again, this loss was related to the possible endogenous PPO activity present in U-RSM. In addition, TSAD content was unchanged during T-RSM hydrolysis, assuming that possible endogenous enzymes were deactivated during toasting.

As previously, the enzymatic hydrolysis was performed on the dry methanolic extract of U-RSM and, surprisingly, the hydrolysis yield was only 55% (Tab. 7, Supplementary Material Fig. S2B). Moreover, it was observed the appearance of a new phenolic compound in the reaction medium (Supplementary Material Fig. S2B, compound 8) and according to the mass and NMR (1H, 13C) spectra, it was identified as 1,2-di-O-sinapoyl-β-D-glucose (Supplementary Material Figs. S3 and S4). This new phenolic compound might result from transesterification of sinapine (or mono-sinapoyl glucose isomers) with the glucose molecules present in the extract. Regarding this matter, most feruloyl esterases displays a classical Ser-His-Asp catalytic triad when it performs transesterification with ferulic acid esters via the formation and subsequent breakdown of a covalent acyl-enzyme intermediate (Prates et al., 2001; Dilokpimol et al., 2016; Kelle et al., 2016) and it is reasonable to presume that it will be the same for sinapine (or mono-sinapoyl glucose isomers) in the presence of AnFaeA in the buffered extract of U-RSM. A possible catalytic mechanism of action is proposed in Supplementary Material, Figure S5. On the other hand, regarding the TSAD content, it remained higher and constant during hydrolysis kinetics (around 59.2 μmol SAE/DDM, data not shown). Thus, this might indirectly confirm the presence of phenolic-degrading enzymes (i.e. PPO for instance) in U-RSM that were not extracted with methanol.

### 4.2 Release of free sinapic acid from RSM samples with AnFaeA

The treatments were performed under optimal pH (5.5), temperature (55 °C) and enzyme concentration (39 nkat/g) for the complete hydrolysis of all the SAEsters present in rapeseed samples. Moreover, the total phenolic content of RSM samples was expressed as total sinapic acid derivatives (TSAD) content because 98% of their phenolics were shown to be sinapic acid esters (Khattab et al., 2010; Siger et al., 2013).

The maximum hydrolysis yield of around 70 and 78% was obtained after 240 min incubation for T-RSM and U-RSM respectively (Tab. 7) and almost all sinapine, mono- and di-sinapoyl glucose isomers were hydrolysed at the end of incubation (Supplementary Material Fig. S2A). However, AnFaeA only partially managed to hydrolyse kaempferol-based sinapic acid derivatives, as well as di-sinapoyl gentiobiosides and some tri-sinapoyl gentiobiosides (Supplementary Material Fig. S2A). Furthermore, regarding the initial TSAD content present in U-RSM, a loss of 16% was observed after the 30 min pre-incubation step (before adding the enzyme in the medium, Tab. 7). Again, this loss was related to the possible endogenous PPO activity present in U-RSM. In addition, TSAD content was unchanged during T-RSM hydrolysis, assuming that possible endogenous enzymes were deactivated during toasting.

Table 6. Maximal amount and yield of caffeic acid (CA) after enzymatic treatment of T-SFM and U-SFM dry methanolic extract with ChlE and AnFaeB (from Laguna et al., 2019).

| Sample | Enzyme | Initial TCAD* in the meal (μmol CAE/g DDM) | TCAD at t=0 of enzyme incubation¹ (μmol CAE/g DDM) | Maximum CA released after incubation with enzyme (μmol/g DDM) | Incubation time (min) | CA yield (%) based on TCAD at t=0 of enzyme incubation¹ |
|--------|--------|------------------------------------------|---------------------------------------------|------------------------------------------------|----------------------|---------------------------------------------------|
| T-SFM  | ChlE   | 57.4 ± 1.5                                | 59.9 ± 1.7                                  | 54.0 ± 1.1                                      | 240                  | 90                                                 |
|        | AnFaeB |                                           |                                             | 59.8 ± 2.1                                      | 180                  | 100                                                |
| U-SFM  | ChlE   | 71.7 ± 0.5                                | 66.5 ± 2.0                                  | 55.4 ± 0.3                                      | 240                  | 77                                                 |
|        | AnFaeB |                                           |                                             | 68.1 ± 1.7                                      | 10                   | 95                                                 |

Note: *: TCAD = total caffeic acid derivatives; CAE = caffeic acid equivalent. DDM = defatted dry matter. Values are mean ± SD (n = 2). ¹: The substrate was pre-incubated for 30 min in MOPS buffer at 50 °C before adding enzyme (t=0 of enzyme incubation).
In view of these results, we demonstrate the ability of various fungal carboxylic ester hydrolases to hydrolyse the different phenolic esters present in RSM and SFM samples. However, from all the substrates tested for enzymatic hydrolysis, U-SFM methanolic extract and U-RSM showed the best compromise between initial TSAD or TCAD content, hydrolysis yields and released CA or SA amounts. Finally, in order to develop a sustainable and efficient green process, it would be beneficial to use the ethanol/water mixtures previously studied in Section 2.2 to recover the phenolic compounds from the meals, instead of methanol.

5 Valorization of enriched protein/phenolic fractions produced after dry fractionation processes of U-RSM and U-SFM

Here, the potential valorization routes of the previously hydrolysed phenolic acid and some interesting fractions produced after the separation steps were evaluated.

5.1 Synthesis of novel hydroxyalkyl esters and bis-aryl esters from sinapic acid and caffeic acid and evaluation of their antioxidant activity in and oil-in-water emulsion

It is well known that the esterification of phenolic acids with aliphatic alcohols of adequate chain-length improved their antioxidant activity in heterophase lipid systems (oil-in-water emulsions, liposomes, etc.) and even in cultured cells (Laguerre et al., 2010; Bayrasy et al., 2013; Sørensen et al., 2014). This phenomenon, related to the "cut-off effect", was attributed to a better location and concentration of the lipophilic phenolic esters at the interfacial phase where lipid oxidation occurs. Thus, we were interested in the chemical synthesis of novel hydroxyalkyl esters (R–O–(CH₂)n–OH) and bis-aryl esters (R–O–(CH₂)ₙ–O–R), from sinapic acid or caffeic acids and aliphatic α,ω-diols (OH–(CH₂)ₙ–OH) of increasing chain length from 2 to 12 carbon atoms (Fig. 7), and the evaluation of their antioxidant activities (Laguna et al., 2020). Here, we studied the impact of the presence of the hydroxyl group or the second phenolic unit at the end of the alkyl chain of hydroxyalkyl esters or bis-aryl esters respectively on their antioxidant activities in an oil-in-water emulsion model. Furthermore, SA and CA were selected because, as previously observed in Section 3, they can be easily recovered from U-RSM and U-SFM after enzymatic treatments.

To determine the antioxidant activity of the different molecules in an oil-in-water emulsion, we decided to perform the conjugated autoxidizable triene (CAT) assay as described by (Laguerre et al., 2008). Regarding sinapic and caffeic hydroxyalkyl esters, their antioxidant activities were in the same range or lowered compared to their corresponding initial phenolic acid (data not shown). These results implied that there was a negative influence of the polar hydroxyl group at the end of the alkyl chain of the hydroxyalkyl esters in their

| Sample          | Initial TSAD\(^{*}\) in the meal (μmol SAE/g DDM) | TSAD at t=0 of enzyme incubation\(\(\downarrow\) | Maximum SA released after 240 min incubation with enzyme (μmol/g DDM) | SA yield (%) based on TCAD at t=0 of enzyme incubation\(\(\uparrow\) |
|-----------------|-----------------------------------------------|-----------------------------------------------|---------------------------------------------------------------|---------------------------------------------------------------|
| T-RSM           | 46.0 ± 0.3                                   | 44.8 ± 0.2                                    | 31.6 ± 0.1                                                    | 70                                                           |
| U-RSM           | 64.2 ± 1.2                                   | 53.1 ± 1.0                                    | 41.3 ± 0.3                                                    | 78                                                           |
| U-RSM extract   | 64.2 ± 1.2                                   | 59.2 ± 2.3                                    | 32.3 ± 0.4                                                    | 55                                                           |

Note: \(\uparrow\): TSAD = total sinapic acid derivatives; SAE = sinapic acid equivalent. DDM = defatted dry matter. Values are mean ± SD (n = 2). \(\downarrow\): The substrate was pre-incubated for 30 min in MOPS buffer at 55 °C before adding enzyme (t=0 of enzyme incubation).

Fig. 7. Hydroxyalkyl esters of sinapic acid and caffeic acid (A) and bis-aryl esters of sinapic acid and caffeic acid (B).
antioxidant activities in the model emulsion system. Indeed, these molecules containing two hydrophilic groups and separated by an apolar skeleton are different than classical phenolipids containing one hydrophilic head and a hydrophobic tail. Thus, it is strongly suspected that the decrease in the antioxidant activity observed in this study was related to the negative effect of the hydroxyl group at the end of the alkyl chain. Moreover, this –OH group could possibly affect the orientation at the interface and/or the distribution of hydroxyalkyl esters in the different phases of the emulsion.

On the other hand, an improvement of the antioxidant activity was observed for the two bis-aryl esters series until an optimum was reached with the bis-aryl esters containing a hydroxyalkyl esters in the different phases of the emulsion. Finally, numerous perspectives of self-tanning, these fractions would limit the amounts of formaldehyde implemented during tanning or they could be directly incorporated into the diet of ruminants.

Here, two incubation conditions were study: (i) one at high temperature (50 °C) with a meal fraction:water mixture of 1:2 (w/w) during 48 h and the other one at (ii) low temperature (4 °C) with a meal fraction:water mixture of (1:10) (w/w) at pH 9.0 for 48 h. Moreover, microbial proteolysis on meal fractions were quantified during 24 h in rumen batch fermentations. In the case of the U-RSM-PFe-0.1, the net production of ammonia (NH3) and isovalerate (IC5) produced by dry fractionation processes

### 5.2 Self-tanning of untoasted rapeseed and sunflower meals fraction produced by dry fractionation processes

Phenolic compounds present in rapeseed meal and sunflower meal samples have been studied for their ability to interact with proteins that might lead to the reduction of their simulated degradation by rumen microorganisms (Wischser et al., 2013; Bongartz et al., 2018). Thus, we aim to quantify the impact of treatments – hot or cold and at basic pH – on the rumen degradability of proteins in rapeseed and sunflower positive fraction of the electrodes (PFe of U-RSM-0.1 and U-SFM-0.5) previously obtained after electrostatic-sorting in Section 2.1.2 (Broudiscou et al., 2020). Indeed, these fractions, enriched with proteins and simple phenolics, are suitable raw materials for studying the protein tanning ability of endogenous phenolic compounds. Hence, using this potential property of self-tanning, these fractions would limit the amounts of formaldehyde implemented during tanning or they could be directly incorporated into the diet of ruminants.

### Table 8. Net productions of ammonia (mg/tube) and isovalerate (mmoles/tube) from U-RSM-0.1 and U-SFM-0.5 fraction and culture medium final pH (from Broudiscou et al., 2020).

| Source | Probability Pr > F | Treatment Means per treatment (N= 3) | NH3 | IC5 | pH |
|--------|-------------------|-------------------------------------|-----|-----|----|
| CO     | 0.011             | 0.60                                | 0.82| 6.08 (0.93) | 0.15 (0.070) | −14.8 (0.064) | 6.16 (0.51) |
| HO     | 0.019 (0.20)      | −5.7 (0.53)                         | 6.08 (0.76) | 0.88 (0.20) | −12.6 (0.11) | 6.12 (0.20) |
| CT     | 0.339             | −13.4                               | 6.09| 0.59 | −3.3 | 6.21 |

Note: IC5: isovalerate; NH3: ammonia; RSD: residual standard deviation; comparison of cold and hot pretreatments (CO and HO) with control (CT): the first-order errors are in brackets.

In view of these results, one could assume that it would be theoretically more efficient to use twice as much phenolic acid than bis-aryl esters. However, to validate this assumption, additional experiments are still required to assess the effect of surfactant type and concentration in emulsions and to determine physical properties of bis-aryl ester and their partitioning in the emulsion. Finally, numerous perspectives of application of these bis-aryl esters can also be foreseen, as for instance in the field of materials, as additives or precursors of polymers for the production of packaging with antioxidant properties.
Regarding U-SFM-PFe-0.5, both pretreatments did not significantly modify the production of ammonia and iso-valerate (Tab. 8). However, the trend was similar to that observed with U-RSM-PFe-0.1 and the cold pretreatment was associated with the lower ammonia production. In conclusion, only the cold and alkaline pretreatment allowed to decrease the microbial proteolysis of the fractions from RSM and SFM. Nevertheless, the proportion was too small to provide a nutritional advantage in feeding ruminants.

6 Conclusions and perspectives

Nowadays, rapeseed and sunflower meals are mainly used as animal feed because of their high concentration in protein but they can also be used as a potential source of bioactive natural phenolic compounds. Consequently, any type of process allowing the recovery of phenolic compounds without degradation of other constituents (such as proteins), and the production of meals with improved nutritional value would be beneficial for the oilseed industry. Regarding this matter, in order to obtain proteins with high quality from rapeseed and sunflower meals, we demonstrated that it is necessary to use mild conditions during the desolventization steps. Furthermore, it also allows to conserve higher quantities of phenolic compounds. Moreover, dry fractionation has shown its interest as a pre-purification process for rapeseed and sunflower meals. Regarding this matter, self-tannation by cold treatment of the rapeseed and sunflower meal fractions, obtained by electrostatic sorting and enriched with proteins/phenolic compounds, made it possible to reduce the microbial proteolytic activity in vitro and therefore led to a tanning effect. Yet, despite these interesting results, dry fractionation could not separate phenolic compounds from proteins, the only available solution remaining solvent extraction. Regarding this point, we demonstrated that it could be possible to use mixtures based on water and ethanol in substitution for methanol, and as effective as the latter in extracting phenolic compounds. In this way, it would be interesting to consider the use of ethanol to replace hexane in the extraction of the residual oil after pressing the seeds. Moreover, the co-extraction of oil and phenolic compounds from meals with ethanol and the suitable amount of water could lead to the simultaneous recovery of the oil, the hydro-alcoholic phase containing the phenolic compounds and the delipidated and dephenolized meals enriched with proteins. These resulting meals could be then used to obtain fractions enriched only with protein by performing dry fractionation processes. On the other hand, once extracted the phenolic fraction, we proved that it could serve as a source of free bioactive phenolic acids by using different fungal carboxylic ester hydrolases. Later on, sinapic acid and caffeic acid were esterified to produced novel bis-aryl esters with strong antioxidant activities. In view of these results, the PHENOEO project has demonstrated that rapeseed and sunflower meals could be integrated in a green process of biorefinery for the recovery of their bioactive phenolic compounds. However, there are still many parameters that need to be explored before concluding on the efficiency of the different processes presented here, such as the impact on the techno-functional properties and the amino acid profile of the different fractions after dry fractionation processes or enzymatic hydrolysis and their feasibility on a larger scale, in particular in terms of energy costs and expenses, aspects that were not addressed in this work. Finally, regarding the production of bis-aryl esters, chemical synthesis is not likely the most suitable from an environmental point of view. Accordingly, in order to develop a sustainable and efficient process, it would be interesting to test the enzymes that we have used for the hydrolysis of phenolic esters in Section 4 and in particular AnFaeA which has shown an acyl transfer activity.

Supplementary Material

Figure S1. Chromatograms obtained after enzymatic hydrolysis of U-SFM with 30 nkat AnFaeB/g DDM (A) and 10 nkat ChlE/g DDM (B) at t=0 min (red) and t=240 min (black) at 326 nm. 3-O-cafeoylquinic acid (3-CQA) (1), 4-O-cafeoylquinic acid (4-CQA) (2), 5-O-cafeoylquinic acid (5-CQA) (3), caffeic acid (4), 5-O-coumaroylquinic acid (5-CoQA) (5), 5-O-feruloylquinic acid (5-FQA) (6), 3,5-Di-O-cafeoylquinic acid (5,3-diCQA) (7) and 4,5-Di-O-cafeoylquinic acid (3,5-diCQA) (8). From Laguna (2019).

Figure S2. Chromatograms obtained after enzymatic hydrolysis of U-RSM (A) and U-RSM dry methanolic extract (B) with 39 nkat AnFaeA/g DDM at t=0 min (red) and t=240 min (black) at 323 nm. Sinapoyl-kaempferol derivatives (1), sinapine (SNP) (2), trans-sinapic acid (SA) (3), cis-sinapic acid (SA) (4), di-sinapoyl derivatives (5), Tri-sinapoyl derivatives (6), sinapoyl glucose (7) and 1,2-di-O-sinapoyl-β-D-glucose (8). From Laguna (2019).

Figure S3. Chemical structure and NMR \(^1\)H and \(^{13}\)C data of 1,2-di-O-sinapoyl-β-D-glucose. From Laguna et al. (2019).

Figure S4. Chemical structure and LC-MS data of 1,2-di-O-sinapoyl-β-D-glucose. From Laguna et al. (2019).

Figure S5. Possible mechanism for the formation of 1,2-di-O-sinapoyl-β-D-glucose by transesterification of sinapine with 1-O-sinapoyl-β-D-glucose catalyzed by AnFaeA. Note: Mechanism adapted from Dilokpimol et al. (2016).

The Supplementary Material is available at http://www.ocl-journal.org/10.1051/ocl/2020056/olm.

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References

Amarowicz R, Nazck M, Shahidi F. 2000. Antioxidant activity of crude tannins of canola and rapeseed hulls. J Am Oil Chem Soc 77: 957–961.

Aouf C, Lecomte J, Villeneuve P, Dubreucq E, Fulcrand H. 2012. Chemo-enzymatic functionalization of gallic and vanillic acids: synthesis of bio-based epoxy resins prepolymers. Green Chem 14: 2328.

Aouf C, Zago E, Lecomte J, et al. 2016. Polyaromatic dimers, method for preparing same and use of same. Patent WO2016/097657.

Auger B, Marnet N, Gautier V, et al. 2010. A detailed survey of seed coat flavonoids in developing seeds of Brassica napus L. J Agric Food Chem 58: 6246–6256.

Barat A, Rouau X. 2014. New dry technology of environmentally friendly biomass refinery: glucose yield and energy efficiency. Biotechnol Biofuels 7: 138.

Barry TN. 1976. The effectiveness of formaldehyde treatment in protecting dietary protein from ruminal microbial degradation. Proc Nutr Soc 35: 221–229.

Baumert A, Milkowski C, Schmidt J, Nimtz M, Wray V, Strack D. 2005. Formation of a complex pattern of sinapate esters in Brassica napus seeds, catalyzed by enzymes of a serine carboxypeptidase-like acyltransferase family? Phytochemistry 66: 1334–1345.

Bayrasy C, Chabi B, Laguerre M, et al. 2013. Boosting antioxidants by lipophilization: a strategy to increase cell uptake and target mitochondria. Pharm Res 30: 1979–1989.

Bongartz V, Böttger C, Wilhelmy N, Schulze-Kayers N, Südekum K-H, Schieber A. 2018. Protection of protein from ruminal degradation by alkali-induced oxidation of chlorogenic acid in sunflower meal. J Anim Physiol Anim Nutr 102: e209–e215.

Borredon M-E, Berger M, Dauguet S, et al. 2011. Débouchés actuels et futurs du tournesol produit en France–Critères de qualité. Innov Agron 14: 19–38.

Broudisou L-P, Laguna O, Solé-Jamault V, Dauguet S. 2020. Methods assessment of self-tanning of rapeseed and sunflower meal fractions enriched in proteins and phenolic compounds using in vitro measurement of protein rumen degradability. OCL 27: 1.

Cai R, Amftield SD. 2001. A rapid high-performance liquid chromatographic method for the determination of sinapine and sinapic acid in canola seed and meal. J Am Oil Chem Soc 78: 903–910.

Carré P, Citeau M, Robin G, Estorges M. 2016. Hull content and chemical composition of whole seeds, hulls and germ in cultivars of rapeseed (Brassica napus). OCL 23: A302.

Dilokpimol A, Mäkelä MR, Aguilar-Pontes MV, Benoit-Gelber I, Hildén KS, de Vries RP. 2016. Diversity of fungal feruloyl esterases: updated phylogenetic classification, properties, and industrial applications. Biotechnol Biofuels 9: 231.

Dimitrios B. 2006. Sources of natural phenolic antioxidants. Trends Food Sci Technol 17: 505–512.

Diot-Néant F, Migeot L, Hollande L, Reano FA, Domeneck S, Allais F. 2017. Biocatalytic synthesis and polymerization via ROMP of new biobased phenolic monomers: a greener process toward sustainable antioxidant polymers. Front Chem 5: 126.

Fang J, Reichelt M, Hidalgo W, Agnolet S, Schneider B. 2012. Tissue-specific distribution of secondary metabolites in rapeseed (Brassica napus L.). PLoS ONE 7(10): e84006.

Faulds CB. 2010. What can feruloyl esterases do for us? Phytochem Rev 9: 121–132.

Ferreira J, Balsavería S, Sainz M, et al. 2018. Reducing intensity-dependent scratches in optical fibres. J Photochem Photobiol A Chem 362: 127–136.

Fine F, Lucas J-L, Chardigny J-M, Redlingshöfer B, Renard M. 2015. Food losses and waste in the French oilcrops sector. OCL 22: A302.

González MJ, Medina I, Maldonado OS, Lucas R, Morales JC. 2015. Antioxidant activity of alkyl gallates and glycosyl alkyl gallates in fish oil in water emulsions: Relevance of their surface active properties and of the type of emulsifier. Food Chem 183: 190–196.

González-Pérez S, Vereijken JM. 2007. Sunflower proteins: overview of their physicochemical, structural and functional properties. J Sci Food Agric 87: 2173–2181.

Gopalan N, Rodríguez-Duran LV, Saucedo-Castaneda G, Nampoothiri KM. 2015. Review on technological and scientific aspects of feruloyl esterases: A versatile enzyme for biofinetion of biomass. Bioresour Technol 193: 534–544.

Grajeda-Iglesias C, Salas E, Barouh N, Baréa B, Panya A, Figueroa-Espinoza MC. 2016. Antioxidant activity of protocatechuates evaluated by DPPH, ORAC, and CAT methods. Food Chem 194: 749–757.

Gullón B, Eiges G, Moreira MT, Herrera R, Labidi J, Gullón P. 2018. Yeber mate waste: A sustainable resource of antioxidant compounds. Ind Crops Prod 113: 398–405.

Guo W, Hou Y, Ren S, Tian S, Wu W. 2013. Formation of deep eutectic solvents by phenols and choline chloride and their physical properties. J Chem Eng Data 58: 866–872.

Hernández-Jabalera A, Cortés-Giraldo I, Dávila-Ortíz G, et al. 2015. Influence of peptides–phenolics interaction on the antioxidant profile of protein hydrolysates from Brassica napus. Food Chem 178: 346–357.

Holland L, Domeneck S, Allais F. 2018. Chemo-enzymatic synthesis of renewable sterically-hindered phenolic antioxidants with tunable polarity from lignocellulosal and vegetal oil components. Int J Mol Sci 19: 3358.

Horbury MD, Turner MAP, Peters JS, et al. 2020. Exploring the photochemistry of an ethyl sinapate dimer: an attempt toward a better ultraviolet filter. Front Chem 8: 633.

Ivanova P, Chalova V, Uzunova G, Koleva L, Manolov I. 2016. Biochemical characterization of industrially produced rapeseed meal as a protein source in food industry. Agric Agric Sci Proc 10: 55–62.

Jensen SK, Liu Y-G, Eggum BO. 1995. The effect of heat treatment on glucosinolates and nutritional value of rapeseed meal in rats. Anim Feed Sci Technol 53: 17–28.

Kalaydzhiev H, Ivanova P, Stoyanova M, et al. 2020. Valorization of rapeseed meal: influence ofethanol antimentries removal on protein extractability, amino acid composition and fractional profile. Waste Biomass Valoriz 11: 2709–2719.

Karamač M, Kosińska A, Staffa I, Hernández T, Dueñas M. 2012. Antioxidant activity of phenolic compounds identified in sunflower seeds. Eur Food Res Technol 235: 221–230.

Kelle S, Nieter A, Kringins U, Zelena K, Linke D, Berger RG. 2016. Heterologous production of a feruloyl esterase from Pleurotus sapidus synthesizing feruloyl-saccharide esters: FAE production from P. sapidus synthesizing feruloyl-saccharide esters. Bio-technol Appl Biochem 63: 852–862.

Kennedy JA, Jones GP. 2001. Analysis of proanthocyanidin cleavage products following acid-catalysis in the presence of excess phloroglucinol. J Agric Food Chem 49: 1740–1746.

Khattab R, Eskin M, Aliani M, Thiyam U. 2010. Determination of sinapic acid derivatives in canola extracts using high-performance liquid chromatography. J Am Oil Chem Soc 87: 147–155.

Kikuzaki H, Hisamoto M, Hirose K, AkHyama K, Taniguchi H. 2002. Antioxidant properties of ferulic acid and its related compounds. J Agric Food Chem 50: 2161–2168.
Kreps F, Vrbiková L, Schmidt Š. 2014. Industrial rapeseed and sunflower meal as source of antioxidants. Int J Eng Res Appl 4: 45–54.

Kumar N, Pruthi V. 2014. Potential applications of ferulic acid from natural sources. Biotechnol Rep 4: 86–93.

Laguerre M, López-Giraldo LJ, Lecomte J, et al. 2008. Conjugated autoxidizable triene (CAT) assay: a novel spectrophotometric method for determination of antioxidant capacity using triacetyl-glycerol as ultraviolet probe. Anal Biochem 380: 282–290.

Laguerre M, López-Giraldo LJ, Lecomte J, et al. 2010. Relationship between hydrophobicity and antioxidant activity of “phenolipids” in emulsion: a parabolic effect of the chain length of rosmarynate esters. J Agric Food Chem 58: 2869–2876.

Laguna O. 2019. Valorisation des composés phénoliques des tourteaux de colza et tournesol: du fractionnement des matières premières vers la synthèse de molécules multifonctionnelles. In: Sciences agricoles. Université Montpellier. Français. NNT: 2019MONTG007. tel-02142245f.

Laguna O, Barakat A, Alhamada H, et al. 2018. Production of proteins and phenolic compounds enriched fractions from rapeseed and sunflower meals by dry fractionation processes. Ind Crops Prod 118: 160–172.

Laguna O, Odinot E, Bisotto A, et al. 2019. Release of phenolic acids from sunflower and rapeseed meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Laguna O, Durand E, Baréa B, et al. 2020. Synthesis and evaluation of antioxidant activities of novel hydroxyalkyl esters and bis-aryl esters based on sinapic and caffeic acids. J Agric Food Chem 68: 9308–9318.

Lin F-H, Lin J-Y, Gupta RD, et al. 2011. Microwave-assisted extraction of phenolic compounds from wine lees and spray-dried wine. Food Chem 125: 826–832.

Milkowksi C, Strack D. 2010. Sinapate esters in brassicaceous plants: biochemistry, molecular biology, evolution and metabolic engineering. Planta 232: 19–35.

Millet M, Poupard P, Guilois-Dubois S, Zanchi D, Guyot S. 2019. Self-aggregation of oxidized procyanidins contributes to the formation of heat-reversible haze in apple-based liqueur wine. Food Chem 276: 797–805.

Mosenthin R, Messerschmidt U, Sauer N, Carré P, Quinac A, Schöne F. 2016. Effect of the desolventizing/toasting process on chemical composition and protein quality of rapeseed meal. J Anim Sci Biotechnol 7: 36.

Naczk M, Shahidi F. 2004. Extraction and analysis of phenolics in food. J Chromatogr A 1054: 95–111.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Naczk M, Shahidi F. 2004. Extraction and analysis of phenolics in food. J Chromatogr A 1054: 95–111.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.

Newkirk R, Classen H. 2002. The effects of toasting canola meal on physical and chemical changes of rapeseed and sunflower meals using different carboxylic esters hydrolases from Aspergillus niger. Ind Crops Prod 139: 111579.
Zago E, Durand E, Barouh N, Lecomte J, Villeneuve P, Aouf C. 2015a. Synthesis of lipophilic antioxidants by a lipase-b-catalyzed addition of peracids to the double bond of 4-vinyl-2-methoxyphenol. J Agric Food Chem 63: 9069–9075.

Zago E, Lecomte J, Barouh N, et al. 2015b. Influence of rapeseed meal treatments on its total phenolic content and composition in sinapine, sinapic acid and canolol. Ind Crops Prod 76: 1061–1070.

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