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Regulator of calcineurin 1 modulates vascular contractility and stiffness through the upregulation of COX-2-derived prostanoids

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ABSTRACT

Cyclooxygenase-2 (COX-2) derived-prostanoids participate in the altered vascular function and mechanical properties in cardiovascular diseases. We investigated whether regulator of calcineurin 1 (Rcan1) participates in vascular contractility and stiffness through the regulation of COX-2. For this, wild type (*Rcan1*$^{+/+}$) and *Rcan1*-deficient (*Rcan1*$^{-/-}$) mice untreated or treated with the COX-2 inhibitor rofecoxib were used. Vascular function and structure were analysed by myography. COX-2 and phospo-p65 expression were studied by western blotting and immunohistochemistry and TXA_2 production by ELISA. We found that *Rcan1* deficiency increases COX-2 and IL-6 expression and NF-κB activation in arteries and vascular smooth muscle cells (VSMC). Adenoviral-mediated re-expression of Rcan1.4 in *Rcan1*$^{-/-}$ VSMC normalized COX-2 expression. Phenylephrine-induced vasoconstrictor responses were greater in aorta from *Rcan1*$^{-/-}$ compared to *Rcan1*$^{+/+}$ mice. This increased response were diminished by etoricoxib, furegrelate, SQ 29548, cyclosporine A and parthenolide, inhibitors of COX-2, TXA_2 synthase, TP receptors, calcineurin and NF-κB, respectively. Endothelial removal and NOS inhibition increased phenylephrine responses only in *Rcan1*$^{+/+}$ mice. TXA_2 levels were greater in *Rcan1*$^{-/-}$ mice. In small mesenteric arteries, vascular function and structure were similar in both groups of mice; however, vessels from *Rcan1*$^{-/-}$ mice displayed an increase in vascular stiffness that was diminished by rofecoxib. In conclusion, our results suggest that Rcan1 might act as endogenous negative modulator of COX-2 expression and activity by inhibiting calcineurin and NF-kB pathways to maintain normal contractility and vascular stiffness in aorta and small mesenteric arteries, respectively. Our results uncover a new role for Rcan1 in vascular contractility and mechanical properties.

1. Introduction

Prostanoids synthesized by cyclooxygenases (COX), the constitutive COX-1 or the inducible COX-2, are critical modulators of vascular tone in physiological and pathological conditions. COX-2 is induced by inflammatory stimuli and other factors important for cardiovascular diseases such as angiotensin II or endothelin-1 [1–4] and it is increased in pathological conditions [5–7]. In addition, vasoconstrictor substances from COX-2 participate in the greater vasoconstrictor responses, endothelial dysfunction and increased vascular stiffness observed in different pathologies such as hypertension both in animal models [2,8–13] and in humans [14,15].

Calcineurin (Cn) is a calcium/calmodulin dependent serine/threonine protein phosphatase that plays a key role in many cellular processes [16]. The interaction of Cn with the nuclear factor of activated T cell (NFAT) c1-c4 family triggers their rapid translocation to the nucleus where they become transcriptionally active [17]. NFATs were originally identified in lymphoid cells, but they play a

Abbreviations: Cn, calcineurin; COX-2, cyclooxygenase-2; KHS, Krebs Henseleit Solution; NO, nitric oxide; *Rcan1*, Regulator of calcineurin 1; ROS, reactive oxygen species; SMA, small mesenteric arteries; TXA_2, thromboxane A_2; VSMC, vascular smooth muscle cells.

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critical role in the regulation of physiological functions of many different cells, including vascular smooth muscle cells (VSMC) and endothelial cells [18–20]. Cn inhibitors are widely used drugs that ameliorate organ rejection. However, they produce cardiovascular side effects such as hypertension through modulation of NO and prostanoids pathways. Thus, hypertension in cyclosporine-treated cardiac transplant recipients is associated with decreased prostaglandin levels and increased thromboxane A_2 synthesis [21]. Although many of the effects of Cn inhibitors can be explained by inhibition of the Cn/NFAT pathway, it has also been described that cyclosporine A toxicity is at least partly independent of this pathway [22,23]. Both Cn/NFAT and the nuclear transcription factor NF-κB are known modulators of the expression of a number of enzymes involved in the control of vascular tone and structure. Among the best characterized are eNOS [24,25], COX-2 [26–30] and some sources of reactive oxygen species (ROS) such as the NADPH Oxidase [31]. Interestingly, a crosstalk between NFAT and NF-κB in the control of COX-2 expression has been suggested in human epithelial cells [32].

Regulator of calcineurin 1 (Rcan1) was first identified as a negative regulator of Cn activity [33]. However, increasing evidence indicate that Rcan1 can also increase [34,35] or not affect [36,37] Cn activity, suggesting that the effects of Rcan1 on Cn may be context-dependent. In addition, novel evidence point to its role in triggering the stabilization of the inhibitor of NF-κB (Iκ-B) therefore attenuating NF–κB transcriptional activation [38–41]. The fact that Rcan1 has been implicated in the development of several inflammatory diseases, point to the importance of Rcan1 as a potent negative regulator of inflammation [38–40,42,43]. However, at cardiovascular level, the role of Rcan1 is still far from being totally understood. Thus, Rcan1 has been described as inhibitor [35,44] and as inductor [35] of cardiac hypertrophy depending on the stimuli. In addition, we previously reported that *Rcan1*-deficient (*Rcan1*$^{-/-}$) mice were resistant to angiotensin II-induced aneurysms, neointima formation and atherosclerosis [36,37]. However, knowledge about its role in vascular contractility is limited and no evidence on how *Rcan1* participates in vascular structure and mechanical properties has been provided. The aim of the present study was to shed light on the role of Rcan1 and its possible downstream pathways namely Cn, NF-kB and COX-2 in vascular contractility and structure.

2. Methods

2.1. Animal experimental design

All experimental procedures were approved by the Animal Care and Use Committee of Centro Nacional de Investigaciones Cardiovasculares (CNIC), Universidad Autónoma de Madrid and by the Madrid regional authorities (ref. PROEX 345/14), and conformed to European Union guidelines for the care and experimental use of animals. The study was conducted in accordance with the National Institutes of Health (NIH) Guide for the Care and Use of Laboratory Animals (NIH Publication No. 85-23, revised 1996) and the Spanish Policy for Animal Protection RD53/2013, which meets the European Union Directive 2010/63/UE on the protection of animals used for experimental and other scientific purposes. The studies are also in accordance with the ARRIVE guidelines for reporting experiments involving animals [45]. A total of 114 mice were used.

Three-month-old male *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice in C57BL6 genetic background as previously described [46] were used. Animals (approximate weight 25 gr) were breed at the Animal Care Facility of the CNIC. *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ matched controls were produced by crossing the *Rcan1* heterozygous mutant line. Some *Rcan1*$^{-/-}$ mice were treated with the selective COX-2 inhibitor rofecoxib (10 mg·kg^{-1}·day^{-1} i.p., 14 days) or vehicle (0.5% carboxymethylcellulose, 0.025% tween 20, 14 days), as previously described [13]. Animals were housed under controlled conditions at 25 °C in a 12 h light/dark cycle with *ad libitum* access to water and food. Overall mouse health was assessed by daily inspection for signs of discomfort, weight loss, or changes in behaviour, mobility, and feeding or drinking habits. All mice were genotyped by PCR of tail samples using the following primers: *Rcan1*, 5′-GGTGGTCCACGTGTGTGAGA-3′, 5′ −ACGTGAACAAAGGCTGGTCCT-3, and 5′-ATTCGCAGCGCATCGCCTTCTATCGCC-3′. Blood pressure was measured by tail-cuff plethysmography. For this, animals were trained for one week prior to final blood pressure measurements by blinded observers that were unaware of the vascular experiments. Measurements were done always at the same time of the day from 8 to 10 am. 5 individual observations were performed and averaged for each animal. For cell culture procedures and vascular reactivity experiments, mice used were sacrificed by CO_2 inhalation and exsanguination.

2.2. Tissue preparation

Aorta and first-order branches of the mesenteric artery were dissected free of fat and connective tissue and placed in cold Krebs Henseleit Solution (KHS) (115 mM NaCl, 25 mM $NaHCO_3$, 4.7 mM KCl, 1.2 mM $MgSO_4{\cdot}7H_2O$, 2.5 mM $CaCl_2$, 1.2 mM KH_2PO_4, 11.1 mM glucose, and 0.01 mM Na_2EDTA) bubbled with a 95% O_2-5% CO_2 mixture. Analysis of vascular function, structure and mechanics was done on the same day. For immunohistochemistry or immunofluorescence, aortic segments were fixed in 4% paraformaldehyde. Other vascular segments were immediately frozen in liquid nitrogen and kept at −70 °C until further processing for gene expression and western blot studies.

2.3. Plasma samples

Blood samples were collected in tubes containing EDTA (Deltalab), inverted gently from 4 to 6 times and placed in ice. After that, blood sample were centrifuged at 1500 x g for 15 min. Plasma samples were frozen at −70 °C.

2.4. Vascular function

Thoracic aorta and small mesenteric arteries from *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice were dissected and segments, 2 mm in length, were mounted in a small-vessel chamber myograph for measurement of isometric tension according to the method described previously [12]. After a 30-min equilibration period in oxygenated KHS at 37 °C and pH 7.4, segments were stretched to their optimal lumen diameter for active tension development. Segments were washed with KHS and left to equilibrate for 30 min; then, contractility was tested by an initial exposure to a high-K^+ solution (120 mM, KCl). The presence of endothelium was determined by the ability of 10 μM acetylcholine to induce relaxation in arteries precontracted with phenylephrine to achieve a contractile response of approximately 50% of K^+-KHS contraction. Afterwards, concentration-response curves to acetylcholine (1 nM–10 μM) in phenylephrine precontracted arteries or to phenylephrine (1 nM–30 μM) were performed. The role of Cn or NF-κB signalling pathways on phenylephrine-induced contraction was analysed in arteries incubated with the Cn inhibitor cyclosporine A (200 ng/ml) or the NF-κB inhibitor parthenolide (1 μM), which were added when the aorta was removed from the animal. The COX-2 inhibitor etoricoxib (1 μM), the TXA synthase inhibitor furegrelate (1 μM), the TP inhibitor SQ 29548 (1 μM), the NOS inhibitor *N*-nitro-L-arginine methyl ester (L-NAME, 100 μM), or the antioxidant tiron (1 mM) were added 30 min before phenylephrine. Vasoconstrictor responses were expressed as a percentage

of the contraction to KCl and vasodilator responses were expressed as a percentage of the previous contraction to phenylephrine.

In another set of experiments, endothelium removal was performed. For this, a hair was introduced into the vessel lumen and used it to mechanically remove the endothelial layer. Endothelium removal was assessed by the inability of 10 μM acetylcholine to produce vasodilatation.

2.5. Pressure myography

The structural and mechanical properties of small mesenteric arteries were studied with a pressure myograph (Danish Myo Tech, Model P100, J.P. Trading I/S, Aarhus, Denmark). Vessels were placed on two glass microcannula and secured with surgical nylon suture. After any small branches were tied off, vessel length was adjusted so that the vessel walls were parallel without stretch. Intraluminal pressure was then raised to 120 mmHg, and the artery was unbuckled by adjusting the cannula. The segment was then set to a pressure of 45 mmHg and allowed to equilibrate for 60 min at 37 °C in calcium-free KHS ($0Ca^{2+}$; omitting calcium and adding 1 mM EGTA) extravascular and intravascularly perfused, gassed with a mixture of 95% O_2 and 5% CO_2. Intraluminal pressure was then reduced to 3 mmHg. A pressure-diameter curve was obtained by increasing intraluminal pressure in 20 mmHg steps from 3 to 120 mmHg. Finally, the artery was set to 45 mmHg in $0Ca^{2+}$-KHS and then pressure-fixed with 4% paraformaldehyde in 0.2 M phosphate buffer, pH 7.2-7.4 at 37 °C for 60 min and kept in 4% paraformaldehyde at 4 °C for confocal microscopy studies.

Internal and external diameters were continuously measured under passive conditions (D_{i0Ca}, D_{e0Ca}) for 2 min at each intraluminal pressure. The final value used was the mean of the measurements taken during the last 30 s when the measurements reached a steady state. From internal and external diameter measurements in passive conditions, the following structural and mechanical parameters were calculated:

$$\text{Wallthickness(WT)} = (D_{e0Ca} - D_{i0Ca})/2$$

$$\text{Cross-sectionalarea(CSA)} = (\pi/4) \times (D_{e0Ca}{}^2 - D_{i0Ca}{}^2)$$

$$\text{Wall : lumen} = (D_{e0Ca} - D_{i0Ca})/2D_{i0Ca}$$

Incremental distensibility represents the percentage of change in the arterial internal diameter for each mmHg change in intraluminal pressure and was calculated according to the formula:

$$\text{Incrementaldistensibility} = \Delta D_{i0Ca}/(D_{i0Ca} \times \Delta P) \times 100.$$

Circumferential wall strain (ε) = $(D_{i0Ca} - D_{00Ca})/D_{00Ca}$, where D_{00Ca} is the internal diameter at 3 mmHg and D_{i0Ca} is the observed internal diameter for a given intravascular pressure both measured in $0Ca^{2+}$ medium.

Circumferential wall stress (σ) = $(P \times D_{i0Ca})/(2WT)$, where P is the intraluminal pressure ($1\ mmHg = 1.334 \times 10^3\ dyn\ cm^{-2}$) and WT is wall thickness at each intraluminal pressure in $0Ca^{2+}$-KHS.

Arterial stiffness independent of geometry is determined by the Young's elastic modulus (E = stress/strain). The stress-strain relationship is non-linear; therefore, it is more appropriate to obtain a tangential or incremental elastic modulus (E_{inc}) by determining the slope of the stress-strain curve ($E_{inc} = \delta\sigma/\delta\varepsilon$). E_{inc} was obtained by fitting the stress-strain data from each animal to an exponential curve using the equation: $\sigma = \sigma_{orig}e^{\beta\varepsilon}$, where σ_{orig} is the stress at the original diameter (diameter at 3 mmHg). Taking derivatives on the equation presented earlier, we see that $E_{inc} = \beta\sigma$. For a given σ-value, E_{inc} is directly proportional to β. An increase in β implies an increase in E_{inc}, which means an increase in stiffness.

2.6. Determination of aortic structure and elasticity

Aortic rings elasticity was determined using a wire myograph (Danish Myo Tech) as previously described [47] with some modifications. After a 30-min equilibration period in oxygenated $0Ca^{2+}$-KHS to avoid active constriction, passive vascular properties were assessed by exposure to stepwise increases in stretching (200 μm steps, 1 min each) while recording the developed force. This approach calculates vessel elasticity independently of smooth muscle tone but it does not exclude the influence of vascular remodelling.

The vessels were assumed to be cylinders, the length of which did not vary with changes in radius. Since the vessels were found to be flat between the wires, the internal circumference (L) and hence diameter of each vessel segment at each corresponding pressure were calculated using the equation below where *d* is the diameter of the mounting wires and *s* is the separation of the wires from each other's inner surface.

$$L = (\pi + 2)xd + 2xs$$

The circumferential wall force per unit length is the passive wall tension T and was calculated from the following equation which took into account the fact that the forces produced by a ring are equal to twice the force which would have been produced by an equivalent aortic strip, where *F* is the force exerted by the vessel on the tension transducer and *g* is the vessel length:

$$T = F/2xg$$

Effective transmural pressure P was calculated from Laplace's equation where *L* is the internal circumference corresponding to the wall tension:

$$P = 2x\pi xT/L$$

Assuming that the wall cross-sectional area remains practically constant through a range of intraluminal pressures, this parameter was calculated from individual histological hematoxylin-eosin stained transverse sections once the stretching curve was performed. External diameter (De) was then extrapolated from the formula:

$$CSA = \pi/4x(De^2 - Di^2)$$

where Di is the internal diameter calculated from internal circumference at each pressure:

$$Di = L/\pi$$

Wall thickness was then calculated as stated above.

2.7. Organization of internal elastic lamina

The elastin organization within the internal elastic lamina was studied in segments of small mesenteric arteries, using fluorescence confocal microscopy based on the autofluorescent properties of elastin (excitation wavelength 488 nm and emission wavelength 500–560 nm), as previously described [48]. Briefly, the experiments were performed in intact pressure-fixed segments with a Leica TCS SP2 confocal system (Leica Microsystems, Wetzlar, Germany). Serial optical sections from the adventitia to the lumen (z step = 0.5 μm) were captured with a x40 oil objective (Zoom 4), using the 488 nm line of the confocal microscope. A minimum of two stacks of images of different regions were captured in each arterial segment. Quantitative analysis of the internal elastic lamina was performed with Metamorph Image Analysis Software, as previously described [48]. From each stack of serial images, individual projections of the internal elastic lamina were reconstructed,

and total fenestrae number, mean fenestrae area and area occupied by elastin were measured.

2.8. Cell culture

For isolation of VSMC, abdominal and thoracic aortas from *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice were dissected and processed as previously described [36]. Explants were not disturbed during the first days. After outgrowth of adherent cells, cultures were trypsinized and subcultured. All experiments were performed during passages 3–7. Before stimulation, cells were rendered quiescent for 48 h by incubation in serum-free medium. For *Rcan1* re-expression experiments, VSMCs were infected with adenoviruses expressing either GFP-tagged Rcan1.4 (Adeno-Rcan1) or GFP (Adeno-GFP; 1.6×10^8 pfu/ml). Infection efficiency was monitored by flow cytometry (FACS Canto; BD) as previously described [36].

2.9. Immunoblot analysis

Protein extracts from VSMC or aortic segments were obtained using an ice-cold lysis buffer, separated under reducing conditions on SDS-polyacrylamide gels and transferred to nitrocellulose membranes as described [36].

Protein detection was performed with antibodies and dilutions as follows: anti-COX-2 (1:1000 Cayman, cat: 160126), anti-TXA synthase (1:1000 Cayman, cat: 160715), anti-phospo-p65 (1:2000 Cell Signaling, cat:3031), anti-α-tubulin (1:40,000 Sigma-Aldrich, cat:T6074), anti-β-actin (1:100,000 Sigma Aldrich; cat:A3854), anti-eNOS (1:500, BD, cat:610297), anti-p-Akt (1:500, Cell Signalling, cat:9271S), anti-Akt (1:1000, Cell Signaling, cat 9272.), anti-collagen I/III (1:1000; Calbiochem Darmstadt, Germany) and HRP-conjugated secondary antibodies. Immunocomplexes were detected with enhanced chemiluminescence (ECL) detection reagent (Millipore) and subjected to autoradiography (Hyperfilm ECL; Amersham). Signals on the immunoblot were quantified using ImageJ (v 1.45s). β-actin or α-tubulin were used as loading control.

2.10. RT and real-time PCR analysis

Total RNA was DNase-treated and reverse-transcribed as described [13]. Real-time quantitative RT-PCR was performed using TaqMan Gene Expression assays specific for mouse *Cox–2* (Mm00478374_m1), *Il–6* (Mm00446190_m1) *and Adr1* (Mm01328600_m1) or Syber probes: *Col1a2* (Fw: GTCCTAGTCGATGGCTGCTC, Rv: AGCACCACCAATGTCCAGAG), *Fn1* (Fw: ATGAGAAGCCTGGATCCCCT, Rv: GGAAGGGTAACCAGTTGGGG), *Cd3* (Fw: TGGCTACTACGTCTGCTACA, Rv: TATGGCTACTGCTGTCAGGT), *Il2* (Fw: AGATGAACTTGGACCTCTGCG, Rv: AAAGTCCACCACAGTTGCTG). Calculations were made from measurements of 2 replicates of each sample. The amount of target mRNA in samples was estimated by the 2CT relative quantification method using *Hprt1* (Mm00446968_m1) or *β2 microglobulin* (Fw: ACCCTGGTCTTTCTGGTGCTT, Rv: TAGCAGTTCAGTATGTTCGGCTT) for normalization.

2.11. Measurement of TXB_2 and PGE_2

The levels of the metabolite of TXA_2, TXB_2, were determined in the incubation medium using an enzyme immunoassay commercial kit following the manufacturer's instructions (Cayman Chemical). For this, aortic segments from each mice were equilibrated in 300 μl of oxygenated KHS (37 °C) for 30 min. Thereafter, to mimic the situation of the arteries mounted in the myograph, a phenylephrine concentration–response curve was performed keeping the same doses and times. At the end, the medium and the arteries were collected and frozen (−70 °C) for TXB_2 and protein determination, respectively. TXB_2 values were measured and normalized per mg protein.

Plasma PGE_2 levels were determined using an enzyme immunoassay commercial kit following the manufacturer's instructions (Arbor Assays).

2.12. Measurement of vascular NOx levels

Vascular NOx (nitrite plus nitrate) levels were determined in the incubation medium using a colorimetric commercial kit (Arbor Assays) following the manufacturer's instructions. For this, aortic segments from each mice were equilibrated in 100 μl of oxygenated KHS (37 °C) for 30 min. Thereafter, to mimic the situation of the arteries mounted in the myograph, arteries were stimulated with phenylephrine and then 10 μM acetylcholine was added for 5 min. At the end, the medium and the arteries were collected and frozen (−70 °C) for NOx and protein determination, respectively. NOx values were measured and normalized per mg protein.

2.13. Immunohistochemistry

Paraffin-embedded sections were stained using standard histology procedures. Immunostaining was carried out in 3 μm thick tissue sections that were deparaffinised and antigen retrieved using the PT Link system (Dako Diagnósticos S.A, Barcelona, Spain) with Sodium Citrate Buffer (10 mmol/L) adjusted to pH = 6. Endogenous peroxidase was blocked and aorta sections were incubated with the NF-kB p-p65 subunit antibody (1:500) or COX-2 antibody (1:100 Santa Cruz Biotechnology, Inc, sc-1747) overnight at 4 °C. After washing, slides were treated with the corresponding anti-IgG biotinylated-conjugated secondary antibody (Amersham Bioscience) followed by the avidin-biotin-peroxidase complex, and 3,3′-diaminobenzidine as chromogen (Dako). Sections were counterstained with Carazzi's hematoxylin and mounted with DPX. The specificity was checked by omission of primary antibodies and use of non-immune sera. Images were obtained with the Nikon Eclipse E400 microscope, and analysed by Imagepro-plus (Media Cybernetics Inc. Rockville, MD, USA). All samples were evaluated in a blinded fashion. For each mouse, the mean score value was obtained by evaluating 4 different high-power fields (x40) per section.

2.14. Immunofluorescence

Paraffin-embedded sections were treated as described above. Aorta sections were blocked with FBS 5% and incubated with the anti-COX-1 (1:100, Abcam, Cambrigde, UK, cat:ab109025), anti-COX-2 (1:50), anti-Calcineurin A (1:250, Pharmigen, cat:556350) or anti-Rcan1 (1:500, Sigma-Aldrich, cat: D6694) antibodies overnight at 4 °C. After washing, slides were treated with the corresponding fluorescent secondary antibody (Amersham Bioscience). Sections were counterstained with DAPI (Sigma) and mounted with Prolong (Thermofisher). The specificity was checked by omission of primary antibodies and use of non-immune sera. Images were obtained with a Leica TCS SP2 confocal system (Leica Microsystems, Wetzlar, Germany). Serial optical sections (z step = 1 μm) were captured with a x20 or x40 oil objective and analysed by Imagepro-plus (Media Cybernetics Inc. Rockville, MD, USA). All samples were evaluated in a blinded fashion.

2.15. Statistical analysis

All data are expressed as means ± s.e.m. For studies, we performed a minimum of 5 independent experiments. For statistical analysis, we used normalized data to reduce variability of baseline between independent experiments in protein and gene expression

studies. Data were normalized as fold increase over *Rcan1*$^{+/+}$ mean. Group size for parametric testing was at least n = 5, regardless of any statistical power analysis. Student's *t*-test, Mann-Whitney or one- or two-way ANOVA and Bonferroni's post-hoc tests were calculated using GraphPad Prism 5 software (GraphPad Software Inc., San Diego, CA). Statistical significance was assigned at $p < 0.05$.

2.16. Materials

The compounds used were supplied as follow: acetylcholine, phenylephrine, L-NAME, tiron, parthenolide, and furegrelate were from Sigma-Aldrich Co. (St Louis, MO, USA); SQ 29548 (ICN Iberica); etoricoxib was a gift from Dr Godessart (Amirall Prodesfarma); Rofecoxib was from LKT Laboratories (St. Paul, MN, USA) and cyclosporin A was from Novartis.

3. Results

No changes in systolic blood pressure were found between both genotypes (*Rcan1*$^{+/+}$: 98.9 ± 3, n = 5; *Rcan1*$^{-/-}$: 93.4 ± 3.6, n = 5).

3.1. Rcan1 modulates vascular contractility

Contractile responses induced by KCl and endothelium-dependent relaxation induced by acetylcholine were similar in aorta and small mesenteric arteries from *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice (Figs. 1A and S1A and Table 1). However, phenylephrine-induced contraction was greater in aortic segments from *Rcan1*$^{-/-}$ mice (Fig. 1B, Table 1). This difference was not observed in small mesenteric arteries (Fig. S1B). mRNA levels of α1A-adrenoceptors were smaller in aorta from *Rcan1*$^{-/-}$ mice (Fig. 1C).

Endothelium removal increased contractile responses to phenylephrine in aorta from *Rcan1*$^{+/+}$ but it did not modify phenylephrine responses in *Rcan1*$^{-/-}$ mice (Fig. 1D) indicating that endothelial modulation of vasoconstrictor responses is lost in aorta from *Rcan1*$^{-/-}$ mice. Similarly, the NOS inhibitor L-NAME enhanced phenylephrine-induced contraction only in aorta from *Rcan1*$^{+/+}$ mice (Fig. 1E) suggesting decreased NO availability in *Rcan1*$^{-/-}$ mice. However, aortic eNOS expression, Akt activation (Figs. 1F and S2) and aorta NOx levels (Fig. S1C) were similar in *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice pointing to an unaltered NO production in arteries from *Rcan1*$^{-/-}$ mice. Interestingly, the antioxidant tiron inhibited the phenylephrine response only in aorta from *Rcan1*$^{-/-}$ mice (Fig. 1E) suggesting that oxidative stress might be implicated in the decreased NO availability observed in *Rcan1*$^{-/-}$ mice.

3.2. Rcan1 modulates vascular contractility through COX-2 dependent mechanisms

COX-2-derived vasoconstrictor prostanoids are also involved in the reduced NO effects and in the increased vasoconstrictor responses observed in pathological conditions [5,8,9,12]. COX-2 expression was observed in the three layers of the vascular wall in aorta from both genotypes (Figs. 2A and S3A). Both *Cox-2* mRNA and protein levels were greater in aorta (Figs. 2B, 2C, S2 and S3A) and VSMC (Figs. 2E, 2F and S2) obtained from *Rcan1*$^{-/-}$ than in *Rcan1*$^{+/+}$ mice. Moreover, COX-2 protein was increased in small mesenteric arteries from *Rcan1*$^{-/-}$ (Figs. 2D and S2). Notably, adenoviral-mediated re-expression of Rcan1-4 in VSMC from *Rcan1*$^{-/-}$ mice decreased COX-2 expression (Figs. 2G and S2). COX-1 was also expressed in the three layers of the vascular wall and its mRNA levels were not affected by *Rcan1* deletion (Fig. S3B and C).

We then investigated the functional implication of the increased COX-2 expression observed in vessels from *Rcan1*$^{-/-}$ mice. The selective COX-2 inhibitor etoricoxib did not modify phenylephrine responses in aorta from *Rcan1*$^{+/+}$ mice but inhibited this response in *Rcan1*$^{-/-}$ mice (Fig. 3A), suggesting that COX-2-derived contractile prostanoids are responsible for the increased phenylephrine responses in *Rcan1*$^{-/-}$ mice. Similarly, the TXA synthase inhibitor furegrelate (Fig. 3A) and the TP antagonist SQ 29548 (Fig. 3B) decreased the phenylephrine-induced contraction only in aorta from *Rcan1*$^{-/-}$ mice, indicating that TXA_2 is a major contractile prostanoid involved in the hypercontractility to phenylephrine observed after *Rcan1* deletion. Accordingly, aortic segments from *Rcan1*$^{-/-}$ mice released greater levels of TXB_2 than *Rcan1*$^{+/+}$ mice (Fig. 3C) without modification of TXA synthase protein levels (Figs. 3D and S2). However, plasma levels of PGE_2 were similar in both genotypes (Fig. S3D).

3.3. Calcineurin and NF-κB pathways mediate the increased participation of COX-2-derived prostanoids on vasoconstrictor responses in Rcan1$^{-/-}$ mice

Both NF-kB and Cn/NFAT can modulate COX-2 expression [26–29,32] and previous reports have indicated that Rcan1 may regulate NF-kB via a Cn-dependent or independent mechanism [38,40]. Then we questioned the role of Cn and NF-κB in the altered vascular function observed in *Rcan1*$^{-/-}$ mice. Cn, Rcan1 and COX-2 colocalized in the vascular wall (Fig. S4). We previously described that basal Cn enzymatic activity is unaltered in aortas from *Rcan1*$^{-/-}$ mice [36]. Interestingly, the gene expression of the Cn target IL-2 that regulates T cell activation [49], was increased in aorta from *Rcan1*$^{-/-}$ mice in parallel with an augmented gene expression of the T cell marker CD3 (Fig. 4A). On the other hand, phosphorylated p65 protein levels were greater both in VSMC and aorta from *Rcan1*$^{-/-}$ compared to *Rcan1*$^{+/+}$ mice (Figs. 4B, 4C and S2), suggesting higher NF-κB activity in *Rcan1*-deficient mice. In agreement, the mRNA levels of *Il-6*, an NF-κB target, were increased in aorta and VSMC from *Rcan1*$^{-/-}$ compared to *Rcan1*$^{+/+}$ mice (Fig. 4A).

We then analysed the participation of Cn and NF-κB in the vascular contractile responses to phenylephrine. Neither the Cn inhibitor Cyclosporine A (CsA) (Fig. 4D) or the NF-kB inhibitor parthenolide (Fig. 4E) modified phenylephrine contraction in *Rcan1*$^{+/+}$ mice. However, a significant reduction of phenylephrine responses was observed for both inhibitors in *Rcan1*$^{-/-}$ mice (Fig. 4D and E), indicating the participation of both signalling pathways in the increased contractile response observed in *Rcan1*$^{-/-}$ mice. Moreover, incubation of the arteries with CsA or parthenolide plus etoricoxib did not modify the effect of each drug alone on phenylephrine responses (Fig. 4D and E), suggesting that Rcan1 modulates COX-2 activation through Cn and NF-kB pathways.

3.4. Rcan1 modulates vascular mechanical properties through COX-2 dependent mechanisms

We have recently demonstrated the key role of COX-2 in vascular stiffness [13]. Then, we analysed whether Rcan1 contributes to the structure and mechanics of the vascular wall. Vessel and lumen diameter, wall:lumen ratio, wall thickness and cross-sectional area were similar in small mesenteric arteries from *Rcan1*$^{+/+}$ and *Rcan1*$^{-/-}$ mice (Fig. 5A) although a tendency towards a smaller diameter was observed in arteries from *Rcan1*$^{-/-}$ mice. Media thickness and media CSA were smaller in aorta from *Rcan1*$^{-/-}$ compared to *Rcan1*$^{+/+}$ mice (Fig. 5B).

Small mesenteric arteries from *Rcan1*$^{-/-}$ mice showed a significant increase of vascular stiffness as shown by the leftward shift in the stress-strain curve and the increase in the elastic modulus β (Fig. 6A). This effect might be due to a more compact elastin within the internal elastic lamina as shown by a tendency towards

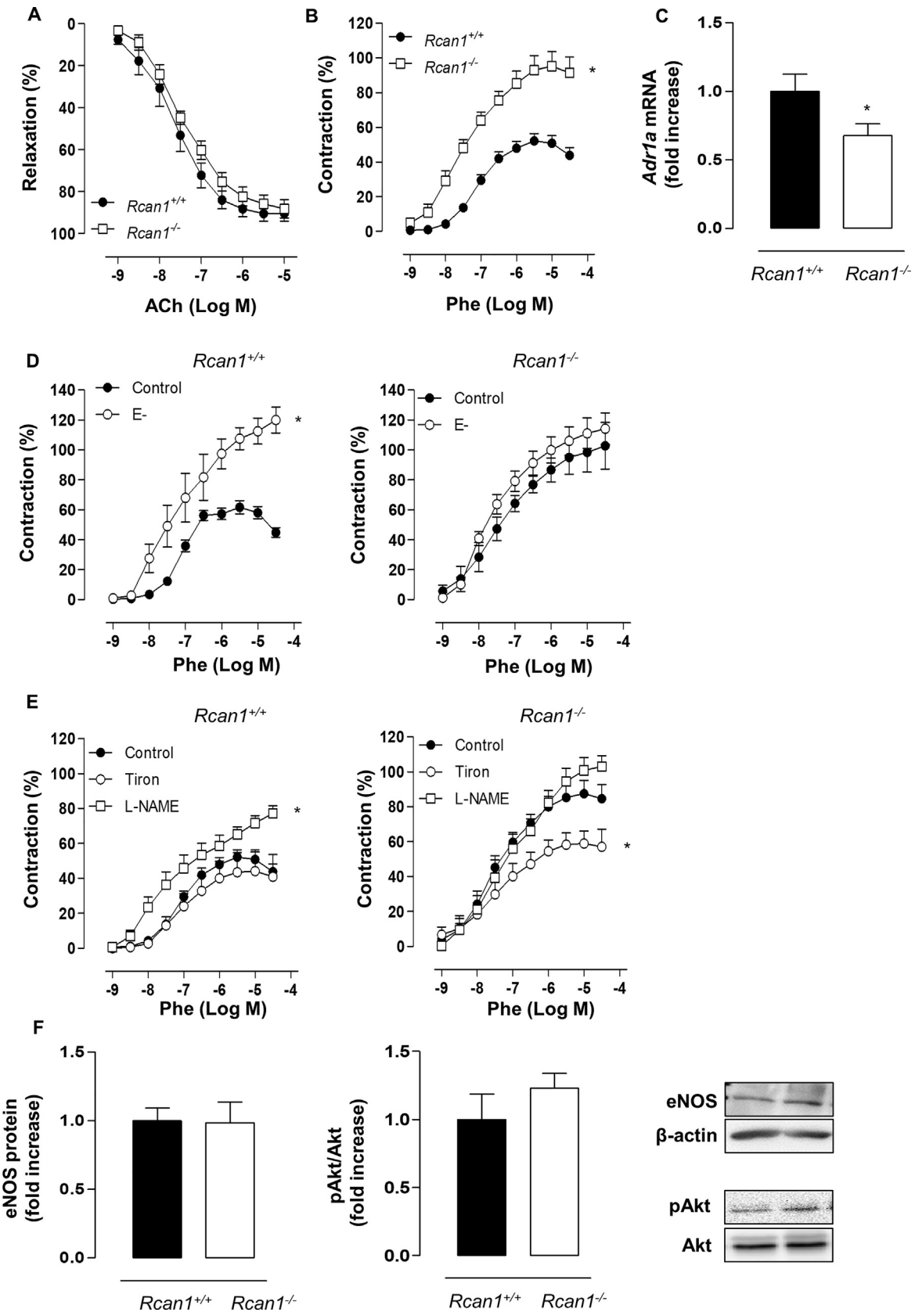

Fig. 1. Rcan1 modulates aortic vascular contraction. Vascular responses to acetylcholine (ACh, A), and phenylephrine (Phe, B) in aortic segments from *Rcan1*$^{+/+}$ (n = 16) and *Rcan1*$^{-/-}$ (n = 14) mice. (C) mRNA levels of $\alpha 1_A$-adrenoceptors (Adr1a) in aorta from *Rcan1*$^{+/+}$ (n = 12) and *Rcan1*$^{-/-}$ (n = 12) mice. (D) Concentration–response curves to phenylephrine-induced contraction in intact and endothelium-denuded (E-) aortic segments from *Rcan1*$^{+/+}$ (n = 6) and *Rcan1*$^{-/-}$ (n = 6) mice. (E) Effect of L-NAME (100 μM) or tiron (1 mM) on vasoconstrictor responses to phenylephrine (n = 5-6). (F) eNOS and phospho-Akt (pAkt) and representative immunoblots in homogenates of aorta from *Rcan1*$^{+/+}$ (n = 6) and *Rcan1*$^{-/-}$ mice (n = 8). β-actin and total Akt was used as loading control. Data are expressed as mean ± s.e.m. *p < 0.05 *vs* control or *Rcan1*$^{+/+}$ mice.

Table 1
Maximal response (Emax) and pD_2 (-logEC50 values) of KCl and phenylephrine (Phe) in aorta and small mesenteric arteries (SMA) from *Rcan1*$^{+/+}$ and *Rcan1*$^{-/-}$ mice.

		Rcan1$^{+/+}$		*Rcan1*$^{-/-}$	
		Emax (mN/mm)	pD_2	Emax (mN/mm)	pD_2
Aorta	KCl	2.3 ± 0.2 (16)		1.9 ± 0.2 (15)	
	Phe	1.3 ± 0.2 (16)	7.10 ± 0.05	2.1 ± 0.2 (15)*	8 ± 0.28*
SMA	KCl	1.9 ± 0.1 (6)		1.8 ± 0.2 (6)	
	Phe	2.2 ± 0.2 (6)	6.15 ± 0.17	2 ± 0.1 (6)	6.4 ± 0.2

Data are expressed as mean ± s.e.m of the number of animals indicated in parenthesis.
* $p < 0.05$ vs *Rcan1*$^{+/+}$.

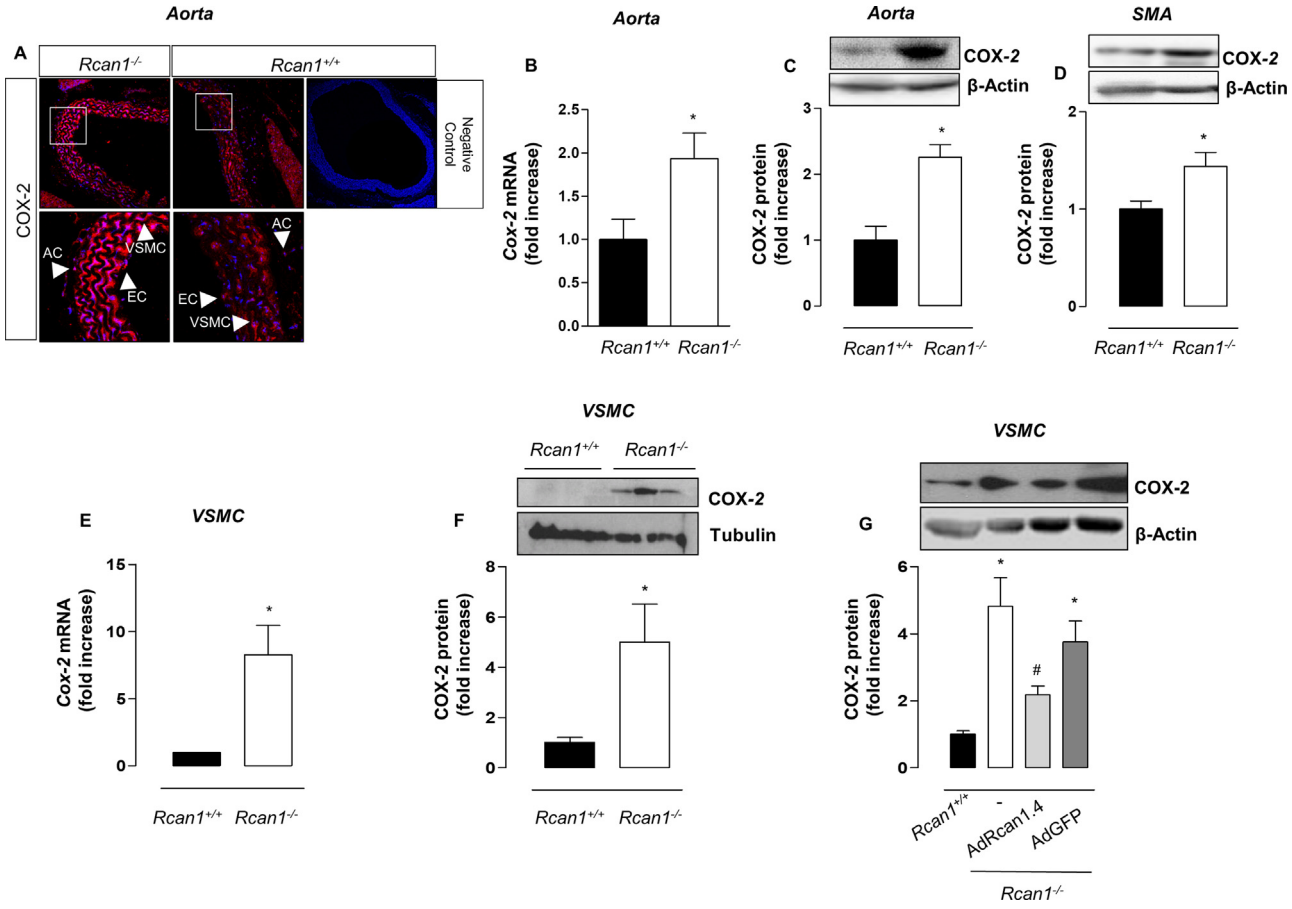

Fig. 2. Rcan1 modulates vascular COX-2 expression. (A) Representative COX-2 immunofluorescence of aortic sections from *Rcan1*$^{+/+}$ (n = 5) and *Rcan1*$^{-/-}$ (n = 5) mice; negative control is also shown. Image size: 375 × 375 μm. Arrows point to endothelial cells (EC), vascular smooth muscle cells (VSMC) and adventitial cells (AC). qPCR analysis of *Cox-2* mRNA levels in aorta or VSMC (B,E) derived from *Rcan1*$^{+/+}$ (n = 8) and *Rcan1*$^{-/-}$ (n = 9) mice. COX-2 immunoblot and quantification in extracts from aorta (C), small mesenteric arteries (SMA) (D) or VSMC (F) derived from *Rcan1*$^{+/+}$ (n = 5) and *Rcan1*$^{-/-}$ (n = 6) mice. β-actin or tubulin were used as loading control in aortic and mesenteric homogenates or VSMC, respectively. (G) COX-2 immunoblot and quantification in extracts from VSMC derived from *Rcan1*$^{+/+}$ (n = 9) and *Rcan1*$^{-/-}$ (n = 9) mice infected with adenovirus encoding GFP-Rcan1.4 (AdRcan1.4) or empty GFP-adenovirus (AdGFP). β-actin was used as a loading control. Gene and protein data are expressed as fold increase of the control group mean value. Data are expressed as mean ± s.e.m. *$p < 0.05$ *vs Rcan1*$^{+/+}$; #$p < 0.05$ *vs Rcan1*$^{-/-}$.

a smaller fenestra area and number that resulted in an increased area occupied by elastin without changes in collagen I/III expression (Fig. 6B and C). Vessel elasticity in aorta was similar in aorta from *Rcan1*$^{-/-}$ and *Rcan1*$^{+/+}$ mice as shown by the similar tension-extension distance and 1/slope (Fig. 6D). However, mRNA level of collagen 1a2 but not fibronectin was increased in arteries from *Rcan1*$^{-/-}$ compared to *Rcan1*$^{+/+}$ mice (Fig. 6D).

Rofecoxib treatment did not affect vascular structure neither in small mesenteric arteries or in aorta (Fig. 5A and B) although again, a tendency towards increased vessel size was observed in mesenteric arteries. However, rofecoxib clearly diminished vascular stiffness both in mesenteric arteries and in aorta (Fig. 6A and D). Intriguingly, rofecoxib treatment did not affect elastin structure in mesenteric arteries (Fig. 6B) but it decreased collagen mRNA levels without affecting fibronectin in aorta (Fig. 6E).

4. Discussion

This study demonstrates that aorta and small mesenteric arteries from *Rcan1*-deficient mice exhibit increased COX-2 expression. In conductance arteries, this is associated with increased production of contractile prostanoids such as TXA_2, which contribute to the augmented vasoconstrictor responses. Moreover, COX-2 derived prostanoids are responsible at least in part, for the increased vascular stiffness observed particularly in small mesenteric arteries from these mice.

Earlier studies demonstrated that during inflammation or vascular damage Rcan1 has a key role in modulating angiogenesis, migration and proliferation [50,51]. In addition, we have previously described that Rcan1 plays a critical role in vascular remodelling associated with pathological states such as aneurysms,

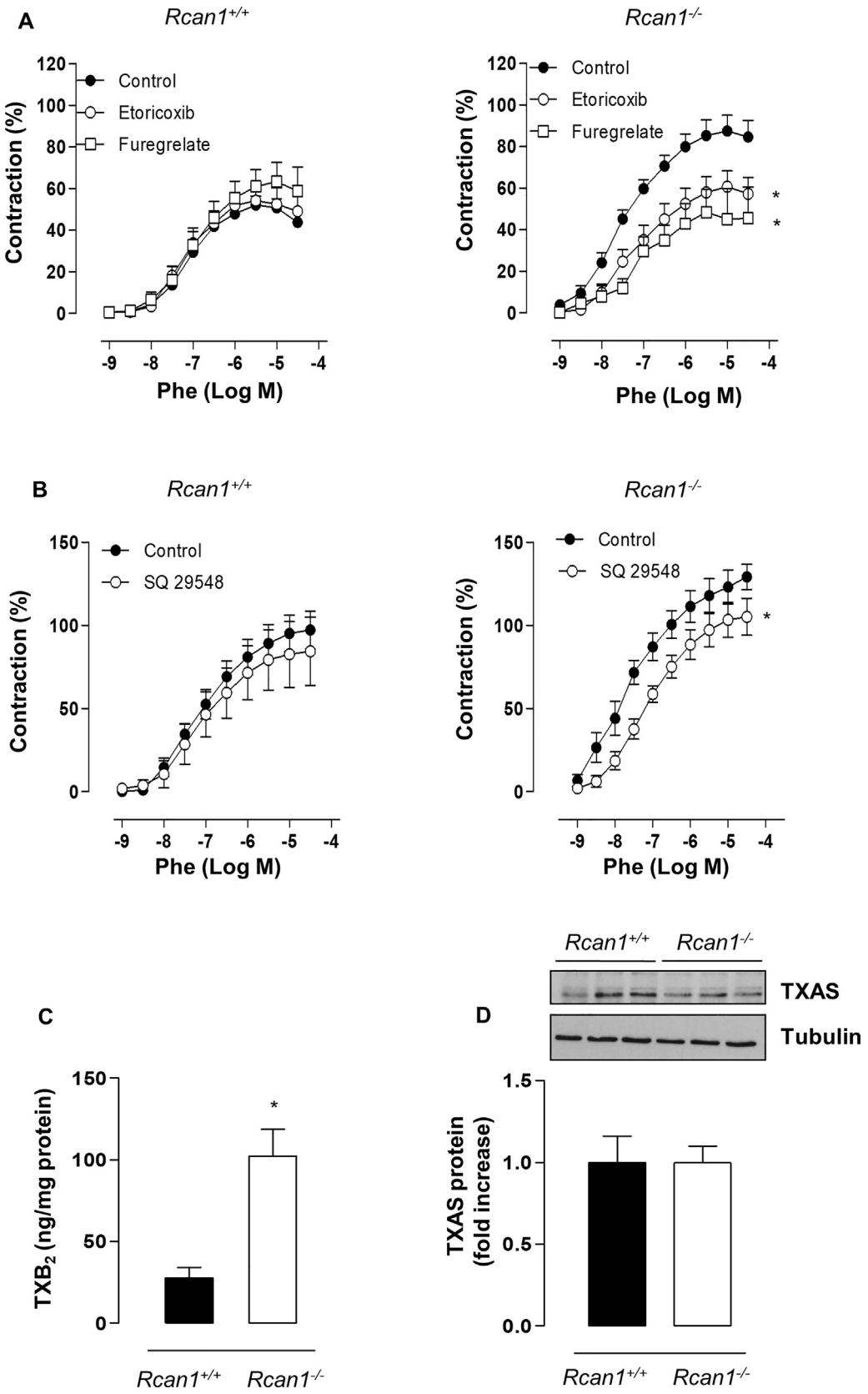

Fig. 3. Rcan1 modulates COX-2 participation on vascular contractility. Effect of etoricoxib (1 μM), furegrelate (100 μM) and SQ 29548 (1 μM) on the concentration–response curve to phenylephrine in intact aortic segments from *Rcan1*$^{+/+}$ (n = 8–16) and *Rcan1*$^{-/-}$ (n = 8–14) mice (A,B). TXB_2 production (C) and TXA synthase (TXAS) protein expression (D) in aorta from *Rcan1*$^{+/+}$ (n = 6) and *Rcan1*$^{-/-}$ (n = 6) mice. β-actin was used as a loading control. Data are expressed as mean ± s.e.m. *p < 0.05 *vs* control or *Rcan1*$^{+/+}$ mice.

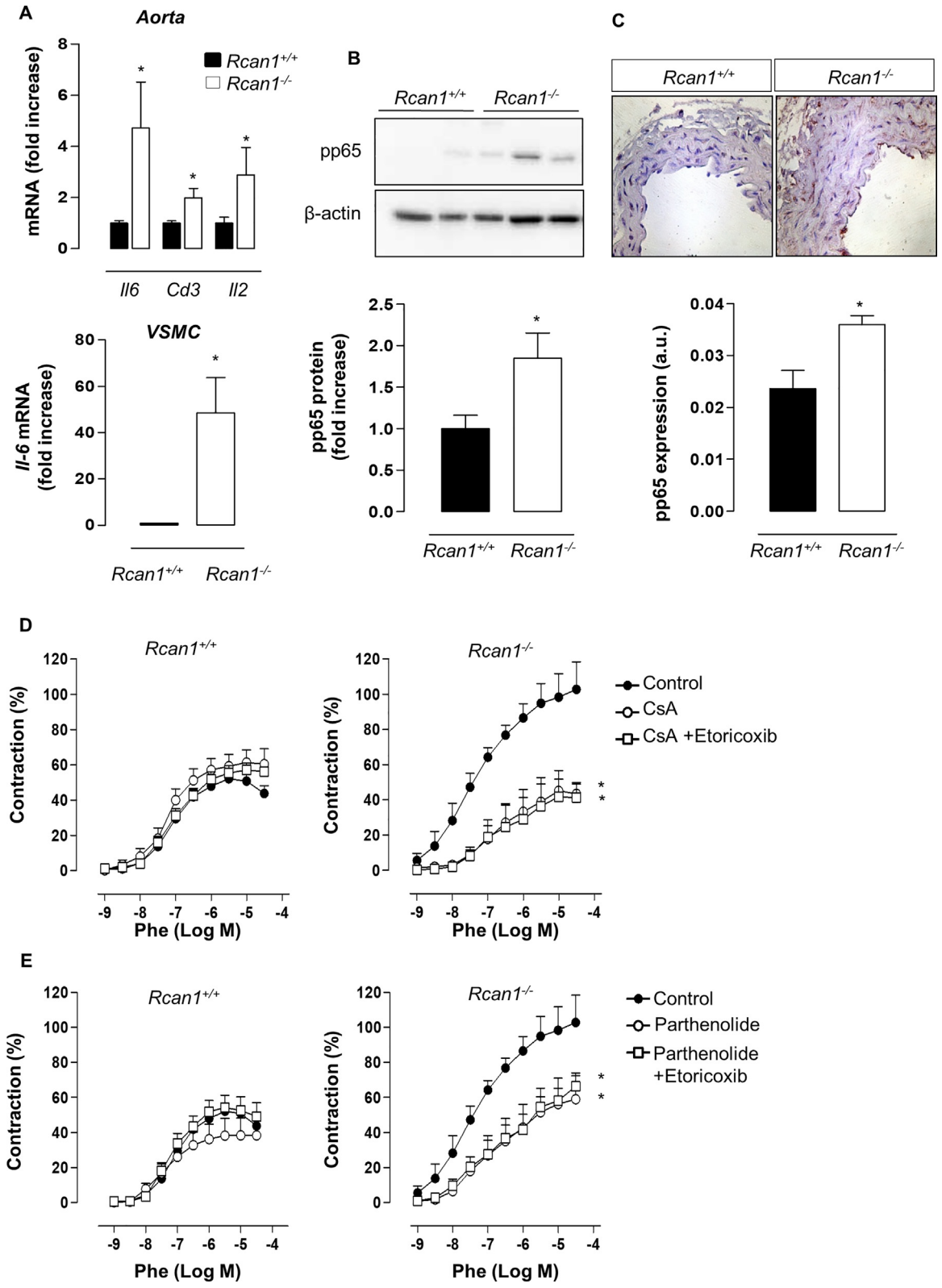

Fig. 4. Rcan1 modulates NF-kB activation. (A) mRNA expression of *Il-6, Cd3 and Il2* in aorta (upper panel) and of *Il6* in VSMC (bottom panel) from *Rcan1*$^{+/+}$ (n = 5–7) and *Rcan1*$^{-/-}$ (n = 5–8). (B) Representative phospho-p65 (pp65) immunoblot and quantification in VSMC from *Rcan1*$^{+/+}$ (n = 6) and *Rcan1*$^{-/-}$ mice (n = 8). β-actin was used as a loading control. (C) Representative pp65 immunostaining and quantification of paraffin-embedded aortic sections from *Rcan1*$^{+/+}$ (n = 5) and *Rcan1*$^{-/-}$ (n = 5) mice. Gene and protein data are expressed as fold increase of the control group mean value. (D and E) Effect of cyclosporine A (CsA, 200 ng/ml) or parthenolide (1 μM) in the absence or in the presence of etoricoxib (1 μM) on the concentration–response curve to phenylephrine in aortic segments from *Rcan1*$^{+/+}$ (n = 6-8) and *Rcan1*$^{-/-}$ (n = 6-8) mice. Data are expressed as mean ± s.e.m. *p < 0.05 *vs* control or *Rcan1*$^{+/+}$ mice.

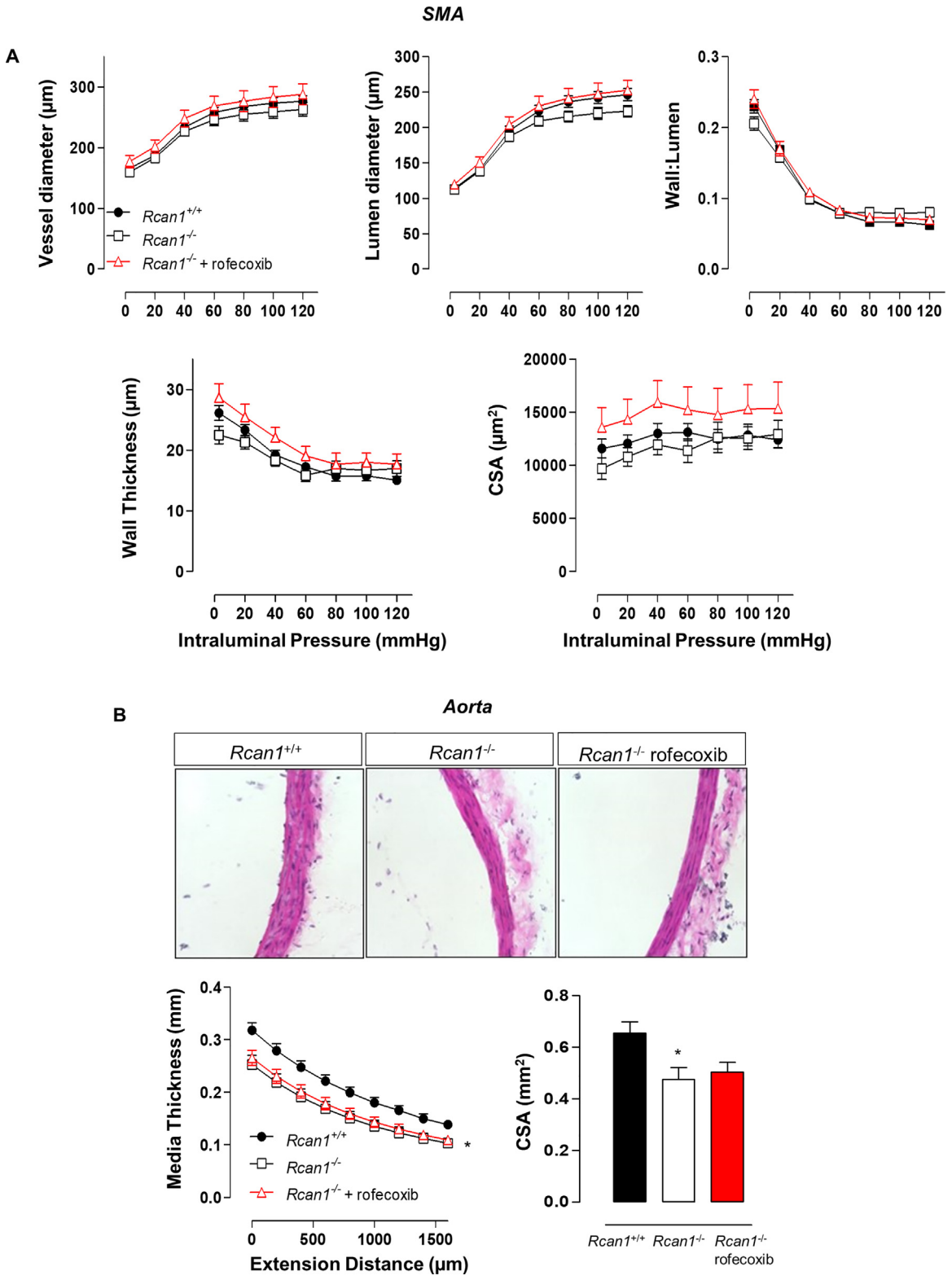

Fig. 5. Rcan1 modulates vascular structural properties. (A) Vessel and lumen diameter, wall:lumen ratio, wall thickness and cross sectional area (CSA) of small mesenteric arteries (SMA) from *Rcan1*$^{+/+}$ (n = 10), *Rcan1*$^{-/-}$ (n = 10), and *Rcan1*$^{-/-}$ mice treated with rofecoxib (n = 6). (B) Media thickness and CSA of aorta from *Rcan1*$^{+/+}$ (n = 7), *Rcan1*$^{-/-}$ (n = 9) and *Rcan1*$^{-/-}$ mice treated with rofecoxib (n = 6). Representative images of hematoxylin-eosin stained aortic sections from all groups of mice are also shown. x60. Results are expressed as mean ± s.e.m. *p < 0.05 *vs Rcan1*$^{+/+}$ mice.

neointima formation and atherosclerosis [36,37]. However, the contribution of Rcan1 to vascular tone and mechanical properties is poorly explored. In fact, to our knowledge, only other study performed in pressurized mesenteric arteries has described decreased vascular contractile responses to phenylephrine [52]. We did not observe differences in blood pressure or contractile responses in the wire-myograph mounted- small mesenteric arteries, however, *Rcan1* deletion increased vasoconstrictor responses to

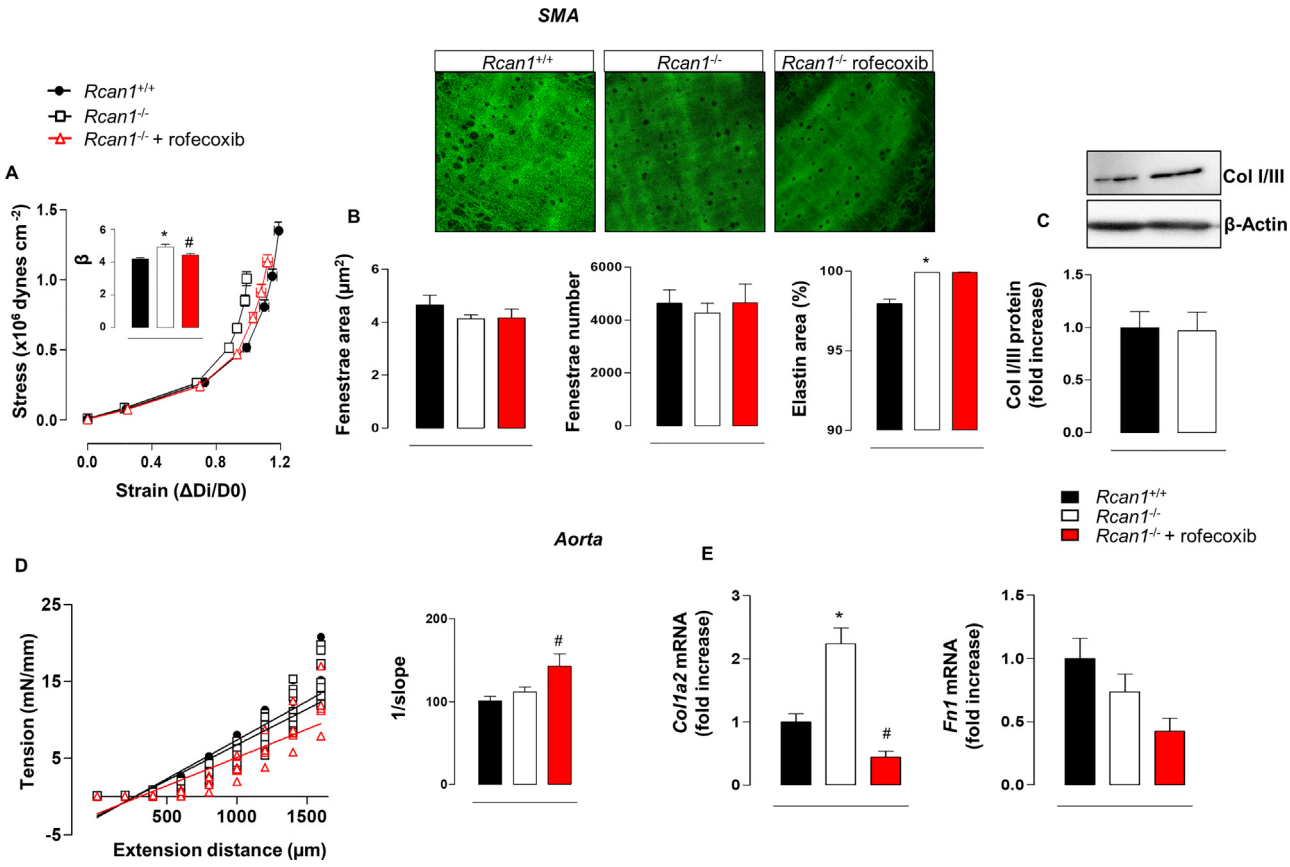

Fig. 6. Rcan1 modulates vascular mechanical properties. (A) Mechanical parameters (stress-strain curve and β values) of small mesenteric arteries (SMA) from *Rcan1*$^{+/+}$ (n = 10), *Rcan1*$^{-/-}$ (n = 10), and *Rcan1*$^{-/-}$ mice treated with rofecoxib (n = 6). (B) Representative images and quantification of internal elastic lamina structure of SMA (n = 6–9). Image size: 93.75 × 93.75 μm (x40, zoom 4). (C) Representative collagen I/III (Col I/III) immunoblot and quantification in extracts from SMA from *Rcan1*$^{+/+}$ (n = 6) and *Rcan1*$^{-/-}$ mice (n = 7). β-actin was used as a loading control. (D) Tension-extension distance relationship and 1/slope of this relationship in aorta from *Rcan1*$^{+/+}$ (n = 10), *Rcan1*$^{-/-}$ (n = 12), and *Rcan1*$^{-/-}$ mice treated with rofecoxib (n = 6). (E) mRNA level of collagen 1a2 (*Col1a2*) and fibronectin (*Fn1*) in aortic homogenates from *Rcan1*$^{+/+}$ (n = 11), *Rcan1*$^{-/-}$ (n = 9), and *Rcan1*$^{-/-}$ mice treated with rofecoxib (n = 6). Gene data are expressed as fold increase of the control group mean value. Results are expressed as mean ± s.e.m. $^{*}p < 0.05$ *vs* *Rcan1*$^{+/+}$ mice; $^{\#}p < 0.05$ *vs* *Rcan1*$^{-/-}$.

phenylephrine in aorta despite having a decrease in mRNA levels of α1A-adrenoceptors, suggesting vascular bed specific differences. The mechanisms responsible for this increased contractile response are also independent of altered contractile capacity of the SMC since KCl responses were similar in both groups. However, we found that endothelial removal or NOS inhibition increased vasoconstrictor responses to phenylephrine in wild type mice but not in *Rcan1*$^{-/-}$ mice, suggesting that the protective role of the endothelium is lost after *Rcan1* deletion. In fact, vasoconstrictor responses were similar in endothelium-denuded arteries from *Rcan1*$^{+/+}$ and *Rcan1*$^{-/-}$ mice (not shown). Interestingly, acetylcholine-induced relaxation, eNOS expression, Akt activation and vascular NOx levels were similar in arteries from both genotypes, arguing against an altered NO production in arteries from *Rcan1*$^{-/-}$ mice. Mechanisms responsible for the loss of protective endothelium might include increased oxidative stress, since the antioxidant tiron inhibited phenylephrine responses only in vessels from *Rcan1*$^{-/-}$ mice. However, other mechanisms might be also involved.

Prostanoids are lipid mediators involved in the control of vascular tone, and increased COX-2 expression/activity participates of features of vascular damage including hypercontractility, associated to pathologies like hypertension [8–10,12]. Several reports have demonstrated a relationship between COX-2 and Rcan1. In human glioblastoma, endothelial cells and mouse primary astrocytes, Rcan1 deficiency induced an increase in COX-2 expression [38,51,53], observations now extended to VSMC. Thus, aorta, small mesenteric arteries and VSMC from *Rcan1*-deficient mice exhibited increased COX-2 expression and endothelial cells show a strong positive staining for the enzyme. Moreover, re-expression experiments of Rcan1 in VSMC (present study) and overexpression of Rcan1 in human vein endothelial cells [54] induced a significant decrease in COX-2 expression, suggesting that Rcan1 is an important modulator of COX-2 expression in different cell types of the vascular wall. Importantly, the increased COX-2 expression induced by *Rcan1* deletion was responsible for the increased phenylephrine contraction observed in aorta from *Rcan1*$^{-/-}$ mice, as shown by the inhibitory effect of the COX-2 inhibitor etoricoxib on vascular contraction, only observed in *Rcan1*$^{-/-}$ mice. The COX-2-derived prostanoid responsible for the increased vascular contraction was probably TXA_2 because both TXAS inhibition with furegrelate and TP blockade with SQ 29548 inhibited phenylephrine responses only in *Rcan1*$^{-/-}$ mice. Moreover, increased vascular TXA_2 production was observed in these mice. Of note, we cannot definitively affirm whether TXA_2 is produced only in the endothelium. Rather we believe that TXA_2 released from the three layers of the vascular wall likely counteract the vasodilator effect of basally released endothelium-derived NO in arteries from *Rcan1*$^{-/-}$ mice. Although little evidence exist that in healthy conditions COX-2 produces TXA_2, in pathological conditions associated with increased inflammation, increased COX-2 and TXA_2 production have been described [7,55]. Specifically, COX-2-derived TXA_2 seems to be involved in vascular hypercontractility in spontaneously hypertensive rats [5,56]. Finally, we could also hypothesize that excessive COX-2 might also contribute to an increased oxida-

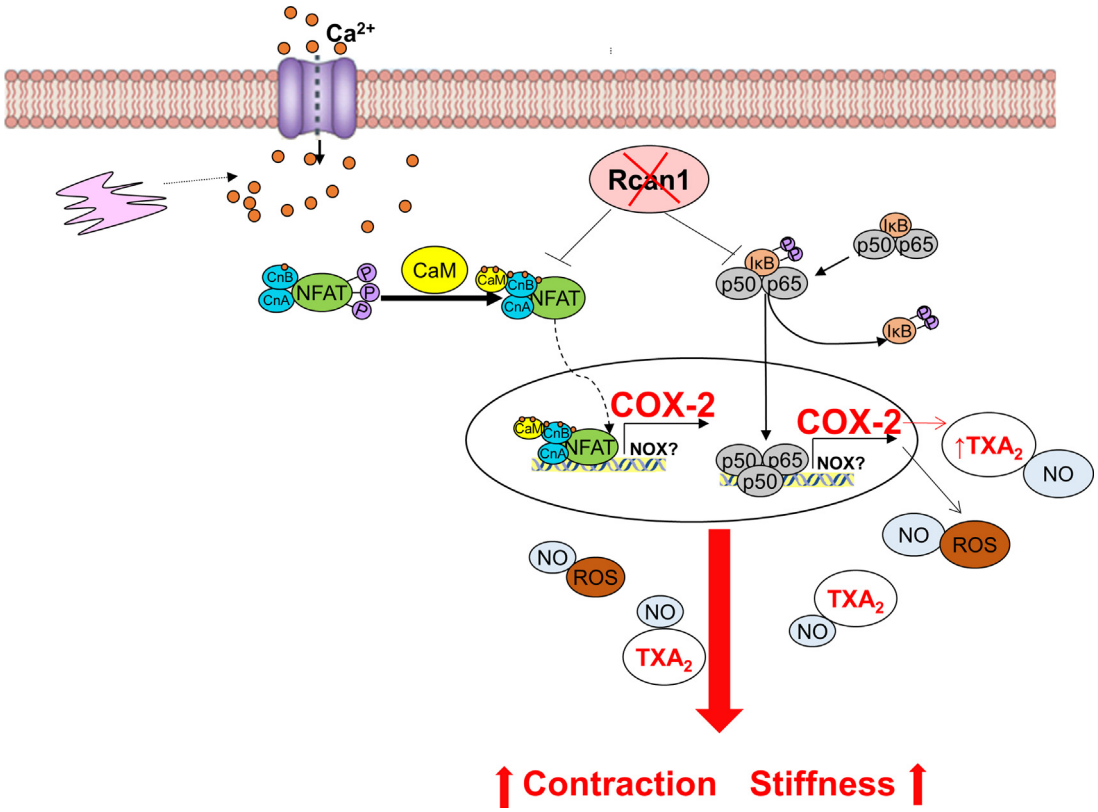

Fig. 7. Rcan1 modulation of vascular tone and stiffness. Vascular tone and stiffness are modulated, among others, by nitric oxide (NO), reactive oxygen species (ROS) likely derived from different sources including NADPH Oxidase (NOX) or COX-2 and prostanoids derived from COX-2. At vascular level, COX-2 expression is regulated by different signalling pathways such as Cn/NFAT and NF-kB. Cn/NFAT signalling pathway is activated by increase in intracellular calcium (Ca^{2+}). Phosphorylation of IkB is required for the canonical activation of NF-kB, IkB-phosphorylated is released from the NF-kB complex, and the p50/p65 subunit translocate to the nucleus. In physiological conditions, Rcan1 acts as negative modulator of Cn/NFAT and NF-kB and prevent the activation of both signalling pathways. Rcan1 deficiency increases Cn-NFAT and NF-kB activities and, therefore, COX-2 expression and TXA_2 and ROS production. In these conditions, an increase in ROS and a decrease in NO bioavailability are produced. All these factors facilitate an increase in vasoconstriction and in the vascular stiffness.

tive stress milieu in arteries from *Rcan1*$^{-/-}$ mice [57,12]. All together, these results demonstrate that at vascular level Rcan1 is an endogenous negative modulator of COX-2 expression and activity and this maintains normal vessel contractility in conductance arteries.

Several evidence demonstrate the critical role of COX-2-derived prostanoids in vascular remodelling and vessel stiffness in diverse pathological conditions [12,13,58,59]. Specifically, we have shown that increased COX-2 expression is associated with increased vascular stiffness and that genetic or pharmacological COX-2 blockade restore normal vascular mechanical properties in hypertension [13]. Our data demonstrate that *Rcan1* deletion differently affect vascular structure in aorta and small mesenteric arteries. Thus, wall thickness remained unaltered in mesenteric arteries but diminished in aorta from *Rcan1*$^{-/-}$ mice. This aortic hypotrophy might have mechanical consequences [60] and help to explain why vascular stiffness (determined by the relationship between passive tension and diameter) was similar in arteries from both genotypes despite of the possible presence of more collagen deposition as shown by mRNA data. Interestingly, small mesenteric arteries from *Rcan1*$^{-/-}$ mice exhibit increased vascular stiffness which is likely associated to altered elastin structure within the internal elastic lamina [48] without apparent changes in collagen. Of note, rofecoxib treatment did not modify vessel structure in aorta or in mesenteric arteries but it significantly improved vessel elasticity in both vessel types, suggesting that Rcan1 is an endogenous modulator of normal vessel mechanical properties through the COX-2 pathway. The underlying mechanisms warrant further investigation since rofecoxib normalized collagen mRNA expression but not elastin alterations in aorta and resistance arteries, respectively.

It is well known that different transcription factors such as CREB, NFAT or NF-κB are involved in the regulation of COX-2 expression in different cell types such as astrocytes, VSMC, epithelial and endothelial cells [26,28,29,32]. Moreover, a relationship between NFAT and NF-κB in the control of COX-2 has been described [32]. Earlier studies identified Rcan1 as a negative regulator of Cn activity [44] and most evidence support these findings in inflammatory situations. However, other evidence suggests that Rcan1 can also act as a facilitator for Cn activity [34,35] and we recently demonstrated that at vascular level, Rcan1 deletion does not affect Cn activity neither in basal situation nor in response to angiotensin II [36]. Here we demonstrate that the Cn pathway is involved in the increased participation of COX-2-derived prostanoids in phenylephrine responses observed in aorta from *Rcan1*$^{-/-}$ mice, since the Cn inhibitor CsA, inhibited phenylphrine contractile response and no further effect was observed after CsA and etoricoxib coincubation. On the other hand, Rcan1 regulates NF-κB activity through mechanisms involving the stabilization of the IκB-NF-κB complex [38,43]. Thus, *Rcan*1 deficiency has been associated to the increase in NF-κB signalling pathway in lung, lymphoma and in mast cells [39–41]. Consistent with this, we found higher levels of phosphorylated-p65 protein and *Il-6* in *Rcan1*$^{-/-}$ aortas and VSMCs. In addition, parthenolide inhibited phenylephrine-contractile responses only in *Rcan1*$^{-/-}$ aortas and no further effect was observed by parthenolide and etoricoxib coincubation, suggesting that Rcan1 is a negative modulator of NF-κB at vascular level.

In summary, we describe a basal proinflammatory vascular profile of Rcan1 deficient mice with an activated calcineurin and NF-κB pathways, increased cytokine levels and COX-2 expression and activity that lead to greater vascular contractile responses in aorta. In addition, Rcan1 deficiency increases COX-2 expression in small mesenteric arteries that is responsible for increased vascular stiffness. Our results uncover a new role for Rcan1 in vascular contractility and mechanical properties. (Fig. 7).

Author contributions statement

ABG-R, VE, LSDdelC, MG-A and NM-B contributed to performance the experimental work; ABG-R and VE to writing the original draft; AMB, MS, MRC and JMR to writing, review & editing.

Conflict of interest

Authors declare no competing financial interests.

Declarations of transparency and scientific rigour

This Declaration acknowledges that this paper adheres to the principles for transparent reporting and scientific rigour of preclinical research recommended by funding agencies, publishers and other organizations engaged with supporting research.

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Appendix A. Supplementary data

Supplementary data associated with this article can be found, in the online version, at https://doi.org/10.1016/j.phrs.2018.01.001.

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