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Unique Characteristics of Radiation-Induced Apoptosis in the Postnatally Developing Small Intestine and Colon of Mice

T. Miyoshi-Imamura,[a,b] S. Kakinuma,[b] M. Kaminishi,[b] M. Okamoto,[b] T. Takabatake,[b] Y. Nishimura,[b] T. Imaoka,[b] M. Nishimura,[b] K. Murakami-Murofushi[a] and Y. Shimada[b,1]

[a] *Genetic Counseling Program, Graduate School of Humanities and Sciences, Ochanomizu University, 2-1-1 Otsuka, Bunkyo-ku, Tokyo, 112-8610, Japan; and* [b] *Experimental Radiology for Children's Health Research Group, National Institute of Radiological Sciences, 4-9-1 Anagawa, Inage-ku, Chiba, 263-8555, Japan*

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We examined the response of the developing mouse intestine to X radiation using neonates (1 day postpartum), infants (2 weeks postpartum) and adults (7 weeks postpartum). Irradiated adult small intestinal crypts displayed two waves of apoptosis. The first wave peaked at 3 h and was followed by a broad wave with a peak persisting from 24 to 48 h. p53 was expressed during the first wave but not the second wave. For the infant small intestine, the intensity of the first wave was approximately half that of the adult wave, and for the colon the intensity was even smaller. In neonates, apoptosis was delayed, peaking at 6 h for small intestinal crypts and at 24 h for colonic crypts. Although no apoptosis occurred at 3 h postirradiation in neonates, p53 was present in both the small intestine and colon, owing at least in part to the inability of p53 to increase the level of Noxa, a p53-dependent pro-apoptosis protein, suggesting a discontinuity in the p53-Noxa-caspase pathway in neonates. By contrast, the induction of p21, a pro-survival protein, was greater in neonatal cells than in adult cells. Thus it appears that the developing and adult intestine mount distinct apoptotic responses to radiation.

INTRODUCTION

Fetuses and young children should not be considered simply as small adults but rather as a unique cohort when assessing the health risks of exposure to environmental carcinogens such as ionizing radiation. Members of this cohort appear to be especially vulnerable to radiation because their organs grow more rapidly and are less differentiated than those of adults. Radiation damage to the tissues has been shown to depend on the degree of cell proliferation and the extent of differentiation. A century ago, Bergonie and Tribondeau stated that "tissues appear to be more radiosensitive if their cells have a greater proliferative capacity, divide more rapidly, and are less-well differentiated" (*1*). The cells of the developing cerebral cortex and the developing kidney are highly susceptible to radiation-induced apoptosis—a sensitivity that is lost after differentiation (*2, 3*). Irradiated hematopoietic and mammary stem cells of weanling mice have been shown to be more severely damaged than those of adult mice (*4, 5*). Exposure of infant mice to 1.5 Gy of radiation depresses their levels of hematopoietic stem cells for a long time thereafter (*6*).

Apoptosis is a form of programmed cell death, i.e., a genetically controlled self-destruction process, occurring during developmental tissue morphogenesis and adult tissue homeostasis (*7*). Apoptosis can be induced by exposure to exogenous DNA-damaging agents including ionizing radiation. Intestinal tract organs of wild-type and genetically engineered adult mice have been used extensively as *in vivo* systems to assess the effects of radiation-induced apoptosis (*8*). The properties that have been monitored include dose response, temporal patterns of apoptosis, spatial distribution of susceptible cells in crypts, differential susceptibilities of small intestinal and colonic epithelial cells and of regions within the colon, and p53, p21 and Bcl-2 activities (*9–24*). However, similar studies using the intestines of neonates and infants have not been undertaken. Therefore, in this study we characterized the features of radiation-induced apoptosis in the postnatally developing small intestine and colon of C57BL/6J mice and compared these features to those of identically treated adult mice. We found that the postnatally developing intestine is more resistant to radiation-induced apoptosis than with the adult intestine, which could be ascribed in part to an apparent inability to completely carry out the post-p53-mediated pathway to apoptosis.

[1] Address for correspondence: Experimental Radiology for Children's Health Research Group, National Institute of Radiological Sciences, 4-9-1 Anagawa, Inage-ku, Chiba, 263-8555, Japan: e-mail: y_shimad@nirs.go.jp.

MATERIALS AND METHODS

Mice

Female C57BL/6J mice were purchased from Charles River Laboratories (Kanagawa, Japan). All mice were exposed to a 12-h dark-light cycle, a temperature of 23 ± 2°C, and 50 ± 10% humidity. They were fed a standard laboratory diet (MB-1; Funabashi Farm Co., Ltd., Chiba, Japan) and given water *ad libitum*. The experimental protocol was reviewed and approved by our institution's animal use committee.

Irradiation of Mice

Irradiation was performed using a Pantak X-ray generator (Pantak Ltd., East Haven, CT). One-day-old (neonate), 2-week-old (infant), and 7-week-old (adult) mice were whole-body irradiated with 2 Gy at a dose rate of 0.7 Gy/min (200 kVp, 20 mA, with a filter composed of 0.5-mm-thick copper and aluminum plates). Subsequently, mice were killed humanely at 0 (unirradiated), 3, 6, 12, 24, 48 and 72 h after irradiation.

Pathology

Unirradiated and irradiated mice were killed after ether anesthesia. Then their small intestines and colons were removed, rinsed in ice-cold phosphate-buffered saline, and fixed quickly in 10% neutral-buffered formalin for about 12 h. Each organ was divided into proximal, middle and distal sections. All samples were embedded in paraffin, sectioned transversely (3–4 μm thick), and stained with hematoxylin and eosin.

Immunohistochemistry

Immunostaining of paraffin-embedded samples followed standard procedures. To retrieve antigens using the primary antibodies [rabbit polyclonal anti-active caspase-3 (1:750, AF835; R&D Systems, Abingdon, UK); rabbit polyclonal anti-p53 (1:500, NCL-p53-CM5; Novocastral Laboratories Ltd., Newcastle, UK); rabbit polyclonal anti-Noxa (1:100, LS-B184/6830; LifeSpan Biosciences, Seattle, WA); rabbit polyclonal anti-p21 (1:500, sc397; Santa Cruz Biotechnology Inc., Santa Cruz, CA); and rat polyclonal anti-Ki-67 (1:100, M7249; DAKO Carpinteria, CA)], the tissue sections in 10 m*M* sodium citrate, pH 6.0, were heated at 120°C for 20 min. After the primary antibodies were washed away, sections were incubated with a peroxidase-conjugated secondary antibody [Histofine® Simple Stain MAX PO(R) or Histofine® Simple Stain MAX PO(Rat); Nichirei Biosciences, Tokyo, Japan]. Peroxidase activity was visualized by first staining with 3,3′-diaminobenzidine (Simple Stain DAB Solution, Nichirei Biosciences, Tokyo, Japan) and then counterstaining with hematoxylin.

Scoring the Numbers and Types of Small Intestinal and Colonic Crypts

The crypt number was defined as the total number of crypts per circumference and was determined by counting the number from crypts found in two to three transverse sections of three mice. For the purpose of identifying the small intestinal crypts, the presence of Paneth cells defined the crypts of mature mice, and clear epithelial invaginations into the mucosa defined the crypts of neonatal and infant mice. We counted a crypt undergoing cleavage, i.e., "crypt fission" or "branching", as two crypts.

Morphologically, apoptosis was defined as the presence of an apoptotic body. This definition correlated well with one that relied on immunohistochemical staining of active caspase 3 (*17*). To quantify the extent of apoptosis, we used two different scoring systems: scoring the number of crypts (as a percentage) containing one or more active caspase 3-positive cells per circumference, and scoring the mean number of apoptotic cells per crypt. However, the extent of apoptosis in the small intestine of 1-day-old mice was scored as the mean number of active caspase 3-positive cells per circumference in the transverse sections, because crypts had not yet formed. The same scoring systems were used to quantify the number of p53-positive crypts (as a percentage) and the mean number of p53-positive cells per crypt. All scoring was performed without knowing whether the mice had been irradiated.

Statistical Analysis

Data are expressed as means ± SEM. Each experiment used three mice. The Student's t test ($P < 0.05$) was used to determine whether experimental values differed significantly between two groups.

RESULTS

Normal Development of Crypts in the Small Intestine and Colon

We first examined the development of intestinal crypts, focusing on morphology and the number of crypts. Plots of the number of crypts present as a function of time and examples of crypt cells expressing Ki-67, reflecting active phases of the cell cycle, are shown in Fig. 1. Supplementary Fig. S1 shows micrographs that document the developmental changes occurring in the anatomical structures of normal small intestinal and colonic crypts between 1 day and 7 weeks postpartum.

1. Small intestine

In the small intestine at 1 day postpartum, invaginating clusters of epithelial cells could be found, but crypts had not yet formed. Morphologically apparent crypts were identified in the proximal region between 4 days and 1 week postpartum and were found next in the middle region and finally in the distal region (Fig. 1A). Then the size and number of crypts increased rapidly (with occasional crypt fission) until 4 weeks postpartum when morphologically mature crypts appeared (Fig. 1A and Supplementary Fig. S1). The number of crypts was nearly constant thereafter (Fig. 1A). Goblet cells were observed 1 day postpartum in the distal region, and Paneth cells, located at the bottom of crypts, developed 1 to 2 weeks postpartum (Supplementary Fig. S1).

At 1 day postpartum, Ki-67 expression was observed in the nuclei of clustered epithelial cells, presumably indicating crypt formation. At 2 weeks postpartum, Ki-67 staining was uniform within a crypt, i.e., was independent of cell position. By 7 weeks postpartum, cells in the proliferative zone (above cell position 4) were most heavily stained (Fig. 1C), as has been reported previously (*18, 19*).

2. Colon

Crypt-like structures were observed at 1 day postpartum (Fig. 1B and Supplementary Fig. S1). Crypt

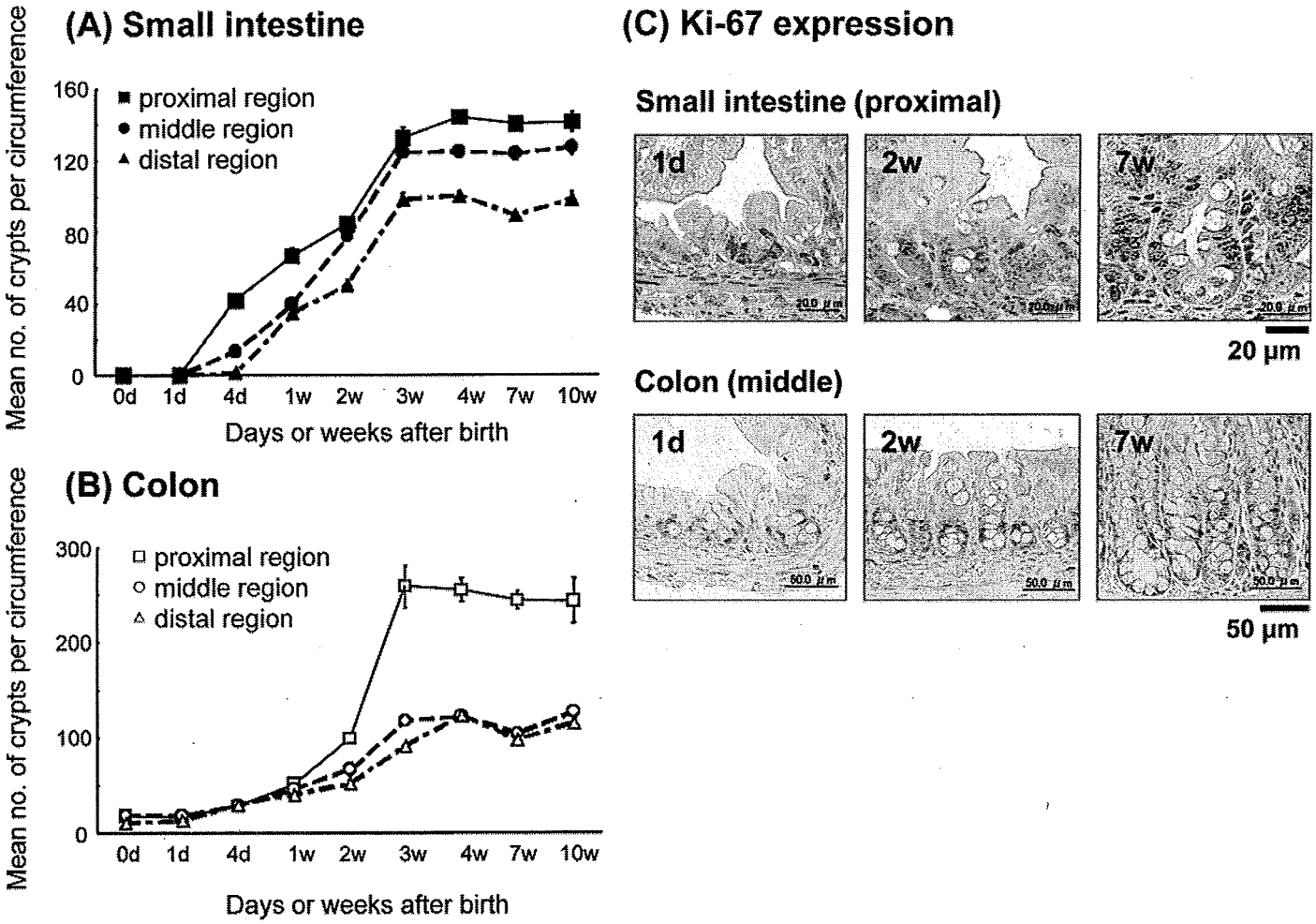

FIG. 1. Developmental changes in the mean number of crypts per circumference for the proximal, middle and distal regions of the small intestine (panel A) and the colon (panel B). Data are reported as means ± SEM. Each experiment used three mice. Panel C: Photomicrographs of Ki-67-stained sections of the small intestine and colon of 1-day-old (1d), 2-week-old (2w) and 7-week-old (7w) mice.

number and size increased with age until 3 to 4 weeks postpartum and were accompanied by frequent crypt fission. Morphologically mature crypts appeared 4 weeks postpartum. The proximal region contained approximately twice as many crypts as did the middle and distal regions (Fig. 1B). Goblet cells developed during the fetal stage (data not shown), which was much earlier than in the small intestine (Supplementary Fig. S1). Ki-67-positive cells were located in the basal one-half to two-thirds of the crypt regardless of age, which was distinctly different from what was seen in the small intestine (Fig. 1C).

Radiation-Induced Apoptosis in Small Intestinal and Colonic Crypts

We next analyzed radiation-induced apoptosis in mice exposed to 2 Gy at 1 day, 2 weeks and 7 weeks postpartum as representatives of neonates with immature or undifferentiated crypts, infants with active proliferative crypts, and adults with a steady number of crypts maintained, respectively (Fig. 1). The dose of 2 Gy was selected based on evidence that the apoptosis in response to ionizing radiation saturates at 1 Gy (*25*). Apoptotic cells were defined as those containing apoptotic bodies and strongly staining for active caspase 3 (*17*). The age and region dependences of the apoptotic response in these mice are shown in Figs. 2, 3 and 4.

1. Small intestine

For mice irradiated at 7 weeks postpartum, apoptosis of crypt cells occurred in two waves (Figs. 2A and 3A). The first wave peaked 3 h after irradiation, as reported previously (*17*), followed by a rapid decrease. The percentage of apoptotic crypts in the first wave was greater than 75% in all regions of the small intestine (Figs. 2A and 4A), and the average number of apoptotic cells was two per crypt (Fig. 3A). The second wave arose thereafter, and its peak level persisted until 48 h. From 10% to 20% of the crypts exhibited apoptosis (Fig. 2A). There were no clear differences among the temporal patterns of the proximal, middle and distal regions (Figs. 2A and 3A). The second apoptotic wave might

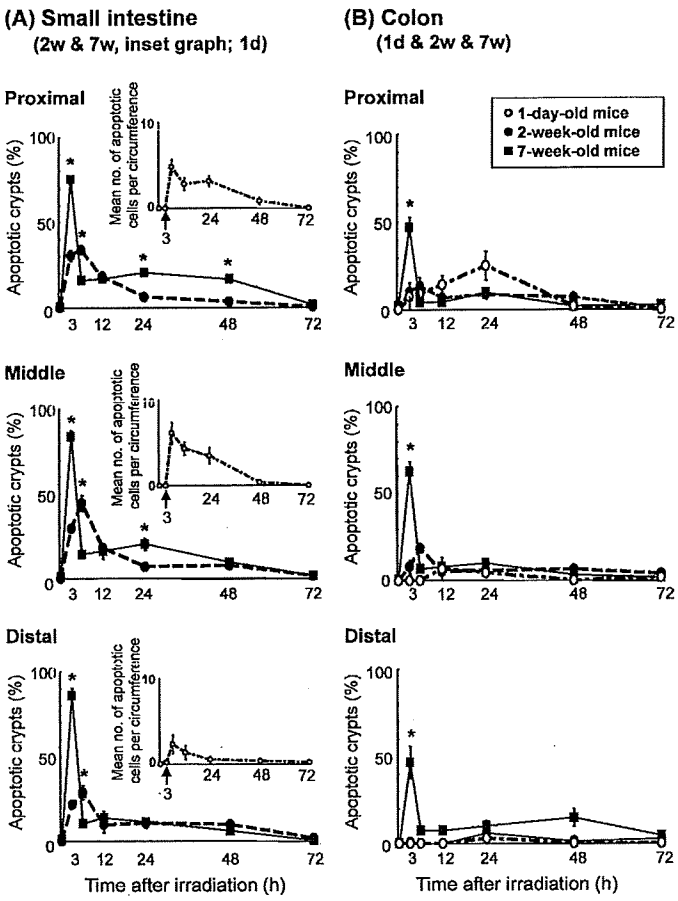

FIG. 2. Percentage of apoptotic crypts as a function of time for the proximal, middle and distal regions of the small intestine (panel A) and the colon (panel B) of mice irradiated at 1 day (1d), 2 weeks (2w) and 7 weeks (7w) postpartum. The inset is a plot of the mean number of apoptotic cells per circumference for 1-day-old irradiated mice. All data are reported as means ± SEM. Each experiment used three mice. For the data points for 2- and 7-week-old mice labeled with an asterisk (*), $P < 0.05$.

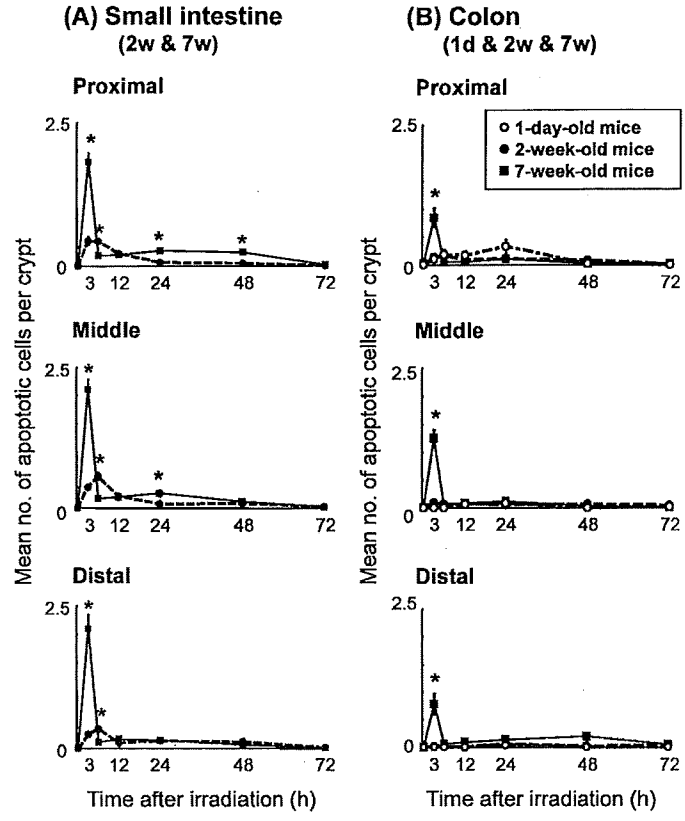

FIG. 3. Mean number of apoptotic cells per crypt as a function of time for the proximal, middle and distal regions of the small intestine of mice irradiated 2 weeks (2w) and 7 weeks (7w) postpartum (panel A) and the colon of mice irradiated 1 day (1d), 2 weeks (2w), and 7 weeks (7w) postpartum (panel B). All data are reported as means ± SEM. Each experiment used three mice. For the data points for 2- and 7-week-old mice labeled with an asterisk (*), $P < 0.05$.

have been the result of a delayed mitotic crisis involving cells that escaped apoptosis during the first wave but then reached the G_2/M checkpoint (*12*).

Compared with the results for 7-week-old mice, the percentage of apoptotic small intestinal crypts in 2-week-old mice at 3 h postirradiation was significantly lower (approximately 20% to 30%; $P < 0.05$; Figs. 2A and 4A). The peak of the first wave occurred 6 h postirradiation, and the average numbers of apoptotic cells per crypt were 0.4, 0.6 and 0.3 for the proximal, middle and distal regions, respectively (Fig. 3A). Only the crypts of the distal region were involved in the second wave of apoptosis (Fig. 2A).

Because no small intestinal crypts were found before the first postpartum day, for the tissues of 1-day-old mice, we used the mean number of apoptotic cells per circumference as a measure of apoptosis. We did not observe any apoptotic cells 3 h after irradiation, instead finding that the maximum apoptosis index occurred at 6 h and persisted until 24 h (Figs. 2A inset and 4A). The mean numbers of apoptotic cells per circumference at 6 h postirradiation were 4.8, 6.2 and 2.3 for the proximal, middle and distal regions, respectively (Fig. 2A insets). At 1 day postpartum, there were approximately 300 to 400 epithelial cells per circumference, which means that approximately 1% of the epithelial cells were very radiosensitive.

2. Colon

For 7-week-old mice, the apoptotic response peaked sharply 3 h postirradiation and was followed by a smaller broad response between 24 and 48 h (Figs. 2B, 3B and 4B). Fewer apoptotic cells were found in colonic crypts than in small intestinal crypts (Fig. 3B), as reported previously (*20*). The percentage of apoptotic crypts for the first wave ranged from 47% to 63%, and the average was slightly more than 0.8 apoptotic cell per crypt (Fig. 2B). Conversely, the apoptosis index of colonic crypts of mice irradiated 2 weeks postpartum was much lower, and the maximum values observed for crypts in the proximal and middle regions occurred at 6 h (Fig. 2B). No apoptotic cells were found in the distal region until 12 h postirradiation (Fig. 2B). Colonic cells

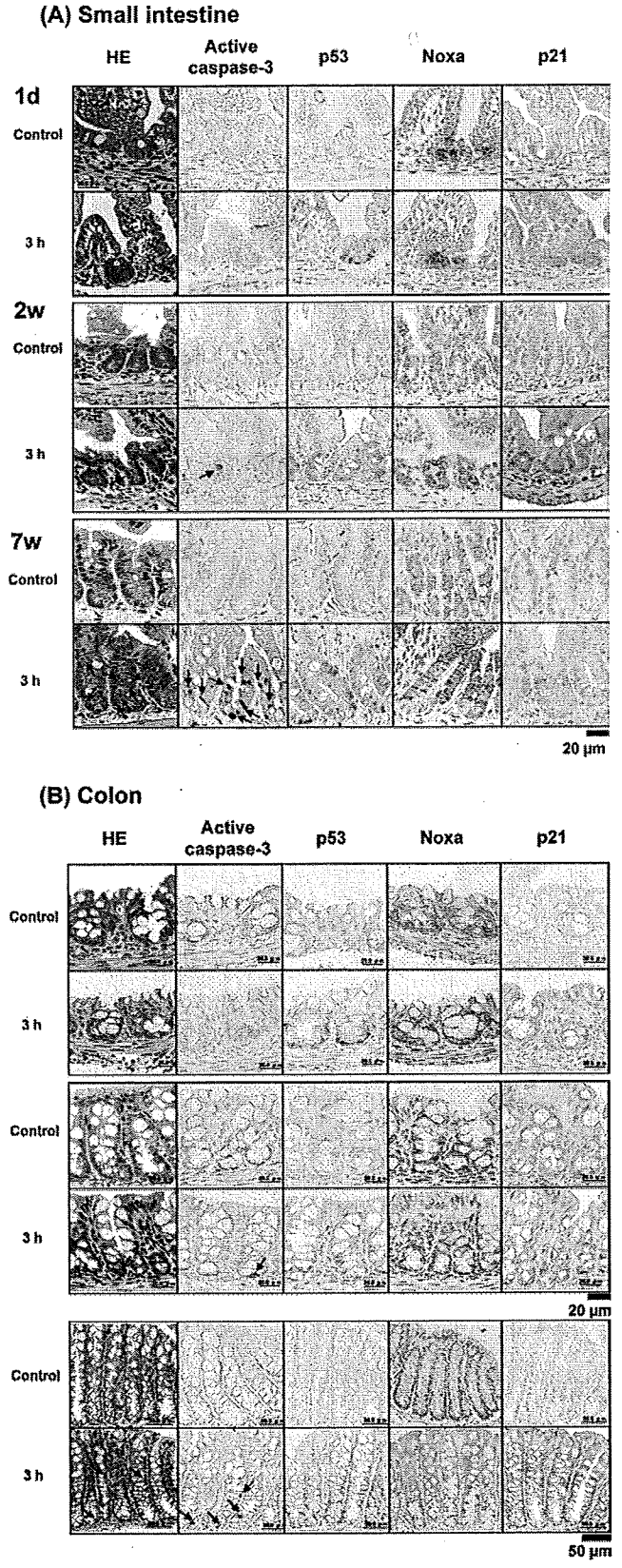

FIG. 4. Photomicrographs of small intestinal (panel A) and colonic (panel B) tissue sections for unirradiated mice and for 1-day-old (1d), 2-week-old (2w) and 7-week-old (7w) mice 3 h after irradiation. From left to right, sections were stained with hematoxylin and eosin or were immunohistologically stained for active caspase 3, p53, Noxa and p21. The arrows point to apoptotic bodies or active caspase 3-positive apoptotic cells.

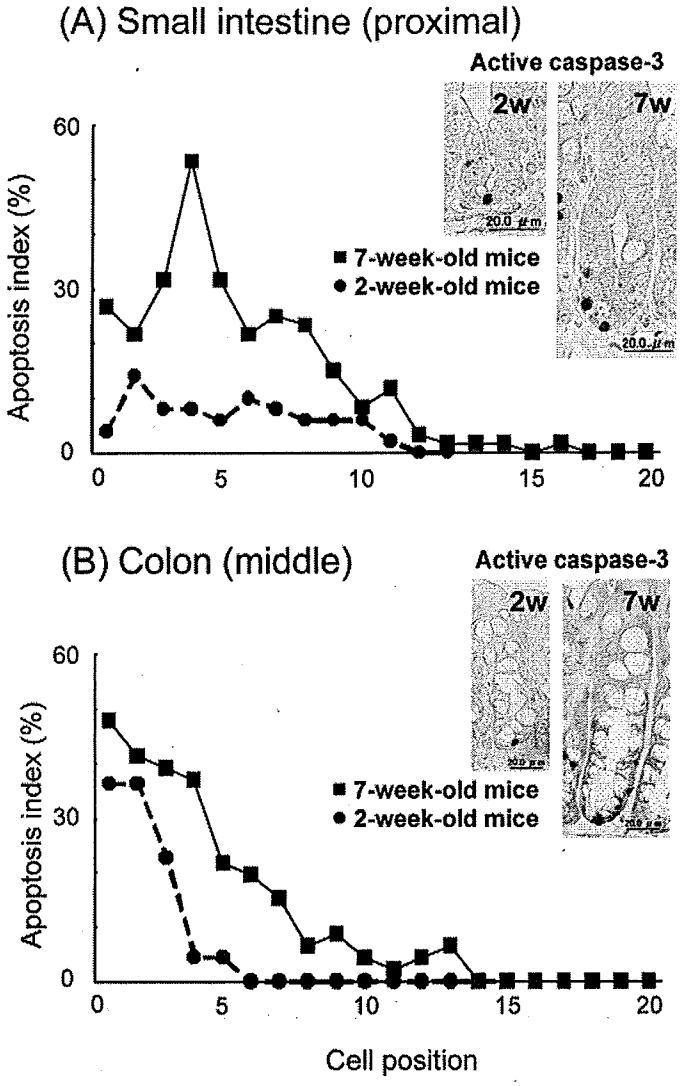

FIG. 5. Age-dependent distribution of apoptotic cells in small intestinal (panel A) and colonic (panel B) crypts. The 2-week-old and 7-week-old mice were irradiated 3 h before being killed. The apoptosis index was calculated using active caspase 3 expression.

irradiated at 1 day postpartum had a unique response to radiation—apoptosis occurred 12 to 24 h postirradiation (Fig. 2B). Moreover, there were regional differences in the mean numbers of apoptotic cells present. The maximum apoptosis index was greater for the proximal colon than for the middle region, and few if any apoptotic crypts were seen in the distal region during the 72-h postirradiation period (Figs. 2B and 3B).

Distribution of Apoptotic Cells in the Small Intestine and Colon

The distribution of apoptotic cells along small intestinal and colonic crypt lengths was determined for 22–50 apoptotic half-crypts of irradiated mice at 2 and 7 weeks of age. The distribution at 3 h postirradiation is shown in Fig. 5. Small intestinal crypts in 7-week-old irradiated mice showed a characteristic peak in the

apoptosis index around cell position 4, a putative stem cell site (*17, 20, 21*). In this study, the small intestinal crypts of 2-week-old mice had smaller apoptosis indices than did those of the adult intestine. Additionally, no specific cell position was associated with a greater radiosensitivity than was any other; instead, similar values for the apoptosis index were broadly distributed as the base of the crypt was approached. Conversely, the frequency of apoptosis in colonic crypts of 7-week-old mice was greatest at cell positions 1–4, the putative stem cell zone (*17, 20*), and declined as the cell position number increased. The colons of 2-week-old mice also had apoptotic cells at the bottom of crypts. These results suggest that normal differentiation of small intestinal cells leads to susceptibility to radiation-induced apoptosis uniquely at cell position 4 during development of the crypt functional architecture, whereas colon cells remain unchanged with regard to cell position-associated radiosensitivity, as indicated by the lack of a difference between 2- and 7-week-old mice.

p53, Noxa and p21 Expression

We examined the age dependence of expression of p53, Noxa and p21, each of which is a crucial determinant of apoptosis (Figs. 4 and 6). Expression of p53 was observed 3 h postirradiation in the cells of the basal half of most intestinal crypts of 7-week-old mice (Fig. 4A and B). Eighty-one percent of the small intestinal crypts were p53 positive, and 100% of the colonic crypts were p53 positive (Fig. 6). Expression of p53 was also high in small intestinal and colonic crypts of irradiated 2-week-old mice, with frequencies of 86% and 95%, respectively. Notably, the apoptosis indices of 2-week-old mice were much lower than those of 7-week-old mice for both the small intestinal and colonic crypts (Fig. 6). Significant p53 expression also occurred in the intestinal crypts of irradiated 1-day-old mice (Fig. 6). Small intestinal cells expressing p53 were clustered in the intervillus region (Fig. 4A); those of the colon were restricted to cells in newly forming crypts (Fig. 4B). It is possible that a link between p53 expression and apoptosis does not exist in the intestine of 1-day- and 2-week-old mice. The levels of p53 expression in the intestinal crypts of 1-day-, 2-week- and 7-week-old mice were negligible at 24 h postirradiation (during the second wave of apoptosis), suggesting that apoptosis was independent of p53 during this time (Fig. 6).

To further characterize the apoptotic response, we immunohistochemically examined the expression of the p53-dependent pro-death factor, Noxa, and pro-survival factor, p21, in the intestinal epithelia of 1-day-old, 2-week-old and 7-week-old irradiated mice. In the small intestine of irradiated 7-week-old mice, Noxa expression increased markedly in cells at positions greater than 4 compared to unirradiated cells, and the expression was

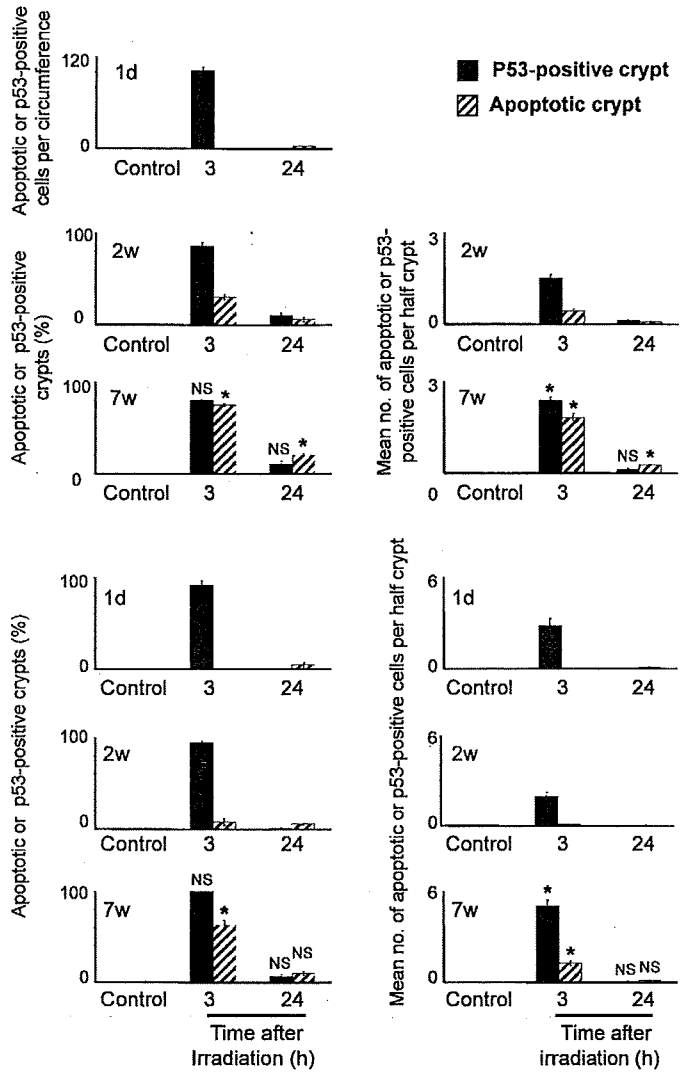

FIG. 6. Percentages of apoptotic crypts and p53-positive crypts (left column) and the mean numbers of apoptotic and p53-positive cells per half crypt (right column) present in tissue sections of the proximal small intestine (panel A) and the colon (panel B) of irradiated mice. For the data for small intestine tissue presented in the right column, mice were 2 weeks old (2w) or 7 weeks old (7w). For 1-day-old (1d) mice, the data for small intestine tissue in the right column are presented as the mean number of apoptotic or p53-positive cells per circumference. Times were 3 h and 24 h after irradiation. The control group was not irradiated. Data are reported as means ± SEM. Each experiment used three mice. For the data points for 7-week-old mice labeled with an asterisk (*), $P < 0.05$ compared to 2-week-old mice.

clearly visible (see dots in Fig. 4A). In irradiated 2-week-old mice, this expression was moderately induced compared to that in 7-week-old mice (Fig. 4A). Unexpectedly, the intervillus region of the small intestine of 1-day-old neonates expressed a substantial amount of Noxa, but the expression level was not influenced by radiation (Fig. 4A). In the colon, Noxa expression was less than that in the small intestine at all ages examined, which may correlate with the relatively low frequency of apoptosis compared to the small intestine (Fig. 4B). The

level of Noxa expression after irradiation in the colonic epithelial cells of 7-week-old mice was increased only slightly. By contrast, in 1-day-old and 2-week-old mice, Noxa expression remained almost unchanged after irradiation (Fig. 4B).

The basal p21 expression levels in the nuclei of small intestinal crypts were the same for 7-week-old mice regardless of whether they had been irradiated. The level of p21 expression increased slightly in cells of 2-week-old irradiated mice. At 1 day postpartum, strong p21 expression was observed both in the nuclei of clustered cells and the cytoplasm of villus and intervillus epithelial cells (Fig. 4A). For the colonic cells of all mice examined, p21 expression was always greater when the mice had been irradiated, as reported previously (*22*). The expression of p21 occurred in the nuclei of cells within the proliferative zone in adult crypts of 7-week-old mice, whereas it occurred throughout crypts of 1-day- and 2-week-old mice (Fig. 4B).

DISCUSSION

For this study, we documented the developmental changes that occur in the small intestine and colon of mice in response to ionizing radiation, an inducer of apoptosis. Unexpectedly, the rapidly growing intestinal epithelial cells of neonatal and infant mice were more radioresistant than were those of adults. Three notable differences were found for how adult and immature crypt cells responded to radiation, as follows. Immature epithelial cells had a delayed first wave of apoptosis in comparison with those of adults. Cells that were extraordinarily susceptible to radiation at mature crypt position 4 were not absorbed in the immature crypt. Especially in neonates, regional differences in radiosensitivities along the intestinal tract were also found—the cells of the small intestinal and colonic distal regions were more radioresistant than were those of the proximal regions.

It has been believed that actively proliferating, immature, undifferentiated epithelial cells are much more sensitive to radiation-induced apoptosis than are differentiated cells (*2–5, 26*). However, as we report here, the intestinal epithelial cells of neonatal and infant mice were more resistant to radiation-induced apoptosis than were those of adults. It has been reported that a dynamic balance between pro-survival and pro-death proteins may determine whether cells undergo apoptosis (*27–29*). In this study, p53 accumulated in irradiated cells regardless of the age of the mouse, whereas there were marked differences in the expression levels of Noxa (a p53-induced pro-apoptosis protein) and p21 (a pro-survival protein) between neonatal, infant and adult intestines. Noxa expression in irradiated adult small intestine increased markedly over the basal level, whereas this was not the case in irradiated neonatal mice. By contrast, p21 expression increased significantly in the cytoplasm and nucleus of neonatal cells in comparison with adult cells. It has been reported that nuclear p21 is necessary for cell cycle arrest (*30, 31*), whereas cytoplasmic p21 inhibits an initiator caspase (*32, 33*). Therefore, after radiation-induced DNA damage, p53 may only marginally induce the expression of pro-apoptosis proteins in neonatal intestinal cells, whereas the cell cycle of neonatal intestinal cells may be arrested efficiently by p53, allowing the cell time to repair its DNA and prevent apoptosis. The balance between cell cycle arrest and apoptosis in response to DNA damage probably changes with development.

Another possible mechanism underlying the radioresistance of neonatal and infant epithelial cells is related to the increased expression of cellular Bcl-2. Bcl-2, which is survival factor, is expressed throughout the intestinal epithelium at embryonic day 14.5 (*34*). At embryonic day 18.5, Bcl-2 expression is restricted to the base of villi; in adult cells, it has been detected in only a small fraction of crypt cells (*11*). Thus the different radiosensitivities of the adult and infant small intestine and colon may be partly explained by their distinctly different Bcl-2 expression profiles (*11, 13, 14*). It was recently found that Wnt/beta-catenin mediates the radioresistance of mouse mammary progenitor cells (*35*). The Wnt signaling pathway is intimately involved in the regulation of intestinal development and maintenance (*36–40*). During the late fetal period, Wnt signaling occurs in newly formed villi and by 3 days postpartum in intervillus cells. After weaning, Wnt activity is confined to the cells of the crypt base (*41*). Therefore, developmental changes in Bcl-2 activity and Wnt signaling may also account in part for radioresistance. Additionally, radioresistance may also be associated with the degree to which cells can repair DNA damage, the rate and mode of stem cell division, and the type and abundance of intestinal microflora present (*42–45*).

Potten has hypothesized that the observed greater resistance to apoptosis by colonic crypts (compared with small intestinal crypts) may account for the greater incidence of colonic carcinoma (*24, 46*). We found here that neonatal and infant intestinal epithelial cells were more resistant to radiation-induced apoptosis than were those of adults. Temporally, $Apc^{Min/+}$ mice, a murine model of human familial adenomatous polyposis, have been shown to be most sensitive to intestinal tumor induction when irradiated at 10 to 12 days of age (*47, 48*). This age-related tumor susceptibility could be explained by a failure of cells with sustained DNA damage to undergo apoptosis and is a possible mechanism carcinogenesis that is consistent with Potten's proposal (*24, 46*).

In conclusion, we demonstrated that, in the developing intestine of C57BL/6J mice, the extent of radiation-induced apoptosis is dependent on age, tissue type and

organ region. Neonatal and infant intestinal epithelial cells were more resistant to radiation-induced apoptosis than were those of adults. When the molecular mechanisms underlying age-related radiosensitivity are better characterized, it may be possible to predict more accurately when children exposed to radiation will develop cancer and use preventive measures to decrease their risk.

SUPPLEMENTARY INFORMATION

Supplementary Fig. S1. Photomicrographs of transverse sections of small intestinal (A) and colonic (B) tissue taken from unirradiated female mice between the ages of 1 day (1d) and 7 weeks postpartum (7w). The arrows labeled A point to clusters of epithelial cells. Arrows labeled B point to crypts undergoing fission. http://dx.doi.org/10.1667/RR1905.1.S1

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Complicated biallelic inactivation of *Pten* in radiation-induced mouse thymic lymphomas

Yu Yamaguchi[a,b,1,2], Takashi Takabatake[b,1], Shizuko Kakinuma[b], Yoshiko Amasaki[b], Mayumi Nishimura[b], Tatsuhiko Imaoka[b], Kazumi Yamauchi[b], Yi Shang[b], Tomoko Miyoshi-Imamura[b,c], Hiroyuki Nogawa[a], Yoshiro Kobayashi[d], Yoshiya Shimada[b,*]

[a] *Department of Biology, Graduate School of Science, Chiba University, Yayoicho, Inage-ku, Chiba 263-8522, Japan*

[b] *Experimental Radiobiology for Children's Health Research Group, Research Center for Radiation Protection, National Institute of Radiological Sciences, 4-9-1, Anagawa, Inage-ku, Chiba 263-8555, Japan*

[c] *Genetic Counseling Program, Graduate School of Humanities and Sciences, Ochanomizu University, 2-1-1 Otsuka, Bunkyou-ku, Tokyo 112-8610, Japan*

[d] *Department of Biomolecular Science, Faculty of Science, Toho University, Miyama 2-2-1, Funabashi, Chiba 274-8510, Japan*

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ABSTRACT

Inactivation of the phosphatase and tensin homolog gene (*Pten*) occurs via multiple tissue-dependent mechanisms including epigenetic silencing, point mutations, insertions, and deletions. Although frequent loss of heterozygosity around the *Pten* locus and plausible involvement of epigenetic silencing have been reported in radiation-induced thymic lymphomas, the proportion of lymphomas with inactivated *Pten* and the spectrum of causal aberrations have not been extensively characterized. Here, we assessed the mode of *Pten* inactivation by comprehensive analysis of the expression and alteration of *Pten* in 23 radiation-induced thymic lymphomas developed in B6C3F1 mice. We found no evidence for methylation-associated silencing of *Pten*; rather, complex structural abnormalities comprised of missense and nonsense mutations, 1- and 3-bp insertions, and focal deletions were identified in 8 of 23 lymphomas (35%). Sequencing of deletion breakpoints suggested that aberrant V(D)J recombination and microhomology-mediated rearrangement were responsible for the focal deletions. Seven of the 8 lymphomas had biallelic alterations, and 4 of them did not express Pten protein. These *Pten* aberrations coincided with downstream Akt phosphorylation. In conclusion, we demonstrate that *Pten* inactivation is frequently biallelic and is caused by a variety of structural abnormalities (rather than by epigenetic silencing) and is involved in radiation-induced lymphomagenesis.

1. Introduction

The phosphatase and tensin homolog (Pten) is an important lipid phosphatase that antagonizes the phosphatidylinositol-3-kinase (PI3K)/Akt signaling pathway [1,2]. The PI3K/Akt signaling pathway is aberrantly activated in a variety of tumors, often resulting from defects in the *PTEN* gene [3,4]. Once activated, Akt promotes fundamental cellular processes such as cell survival, growth, proliferation, angiogenesis, and cellular metabolism. Pten also plays a crucial role as guardian of genome integrity by maintaining chromosomal stability through physical interaction with centromeres and by controlling DNA repair, both of which are independent of Akt activation [5].

PTEN is mutated in a variety of human carcinomas [6,7], and *PTEN* is the second most frequently mutated gene in human cancers after *TP53* [8,9]. Germline mutations of *PTEN* in humans are responsible for Cowden disease, which is characterized by a high risk for thyroid and breast cancers [10]. In addition to genetic alterations resulting in missense, nonsense or frameshift mutations, epigenetic silencing of *PTEN* has been reported in the pathogenesis of gastric and breast cancers [11,12]. Furthermore, overexpression of *PTEN*-targeting microRNAs correlates with decreased expression of PTEN protein in hepatocellular [13] and ovarian cancers [14]. These reports indicate that there are multiple mechanisms responsible for *PTEN* inactivation.

Interestingly, there are significant differences in the location of mutations in *PTEN* with respect to cancer type. For example, a high proportion of glioblastomas have missense mutations in exon 6, which encodes part of the phosphatase domain of PTEN, whereas few mutations have been found in exons 7 and 8, which encode

* Corresponding author. Tel.: +81 43 206 3200; fax: +81 43 206 4138.
E-mail address: y_shimad@nirsgo.jp (Y. Shimada).

[1] These authors contributed equally to this study.

[2] Present address: Pharmaceutical Research Laboratories, Sanwa Kagaku, Kenkyusho Co., Ltd., 363 Shiosaki, Hokusei-cho, Inabe-shi, Mie 511-0406, Japan.

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Table 1
Summary of expression and aberrations of Pten in 23 thymic lymphomas, in parallel with the activation of a downstream factor of Akt.

Tumor ID	LOH[a] status	Structural alteretations[b]	Transcriptional changes[b]	Predicted changes[c]	Pten protein	Akt protein[d]
TL5	–	Duplication of exon6 and exon7	Additional faint long product	Ins of 103 amino acids	A faint larger band	Activated
		509 Ins TGT	nd	170S → M and C	Low	
TL14	C3H	Del of exon4 and exon5	Additional faint short product	Stop at codon264	Absent	Activated
		9621ns A	nd	Stop at codon333	Absent	
TL8	B6	Homozygous deletion	Absent	–	Absent	Activated
TL11	C3H	Homozygous deletion	Aberant splicing	Stop at codon54	Absent	Activated
TL20	B6	Homozygous deletion	Lack of sequence for exon 1	–	Absent	Activated
TL15	–	862 G → G/T	nd	Stop codon	Low	Activated
TL19	C3H	158 T → T/C[e]	nd	53 V → A	Low	nd
TL21	C3H	364 A → T	nd	122 I → F	Low	Activated
TL12	C3H	nd	nd	–	Low	Activated
TL3	–	nd	nd	–	Low	nd
TL6	–	nd	nd	–	Low	nd

[a] Lost allele is shown. (–), retention of heterozygosity.
[b] Ins, insertion; Del, deletion; nd, not detected.
[c] S, serine; M, methionine; C, cysteine; V, valine; A, alanine; I, isoleucine; F, phenylalanine.
[d] "Activated" means that the phosphorylation of Akt protein at Ser473 was detected; nd, not detected.
[e] Sequencing analysis indicates the presence of both mutated and non-mutated sequence, the latter of which may be due to contaminated normal cells.

the C2 domain. In contrast, endometrial carcinomas rarely contain mutations in exon 6; rather, frameshift mutations in exons 7 and 8 are common [6,7]. Deletion of *PTEN* has been identified in 77% of prostate cancer cases, with 25% containing homozygous deletions [15]. Because previous studies have examined only single or limited categories of causal alterations, the overall contribution of each causal *PTEN* alteration remains unclear for many tumor types.

Radiation is a clear etiology for leukemia and lymphoma. Radiation-induced murine thymic lymphomas have been used as a suitable model of human T-cell acute lymphoblastic leukemia (ALL), many of which exhibit *Notch1* and *Ikaros* mutation, *p15* and *p16* alteration, and aberrant activation of Jak-Stat signaling [16–20]. Loss of heterozygosity (LOH) within a broad genomic region of chromosome 19, including the mouse *Pten* locus, has been demonstrated in many thymic lymphomas [21,22]. Although it has been suggested that *Pten* undergoes epigenetic silencing by DNA methylation in radiation-induced murine thymic lymphomas [22], direct evidence has not yet been reported.

To identify the mode of *Pten* inactivation in hematopoietic malignancies, we systematically analyzed the status of *Pten* alleles and Pten expression at the RNA and protein levels in 23 radiation-induced thymic lymphomas developed in B6C3F1 mice; downstream activation of Akt was also analyzed. These analyses revealed that biallelic structural abnormality of *Pten*, but not epigenetic silencing, plays a significant role in radiation-induced lymphomagenesis.

2. Materials and methods

2.1. Tumor induction

The induction of thymic lymphomas was carried out as described [23], with minor modifications. In brief, female B6C3F1 mice were exposed weekly to 2.0 Gy whole-body X-ray radiation for four consecutive weeks starting at four weeks of age. Mice were observed daily until moribund and were then sacrificed under ether anesthesia. All experiments with mice were conducted according to the legal regulations in Japan and were in compliance with the guidelines for the care of laboratory animals of the National Institute of Radiological Sciences.

2.2. LOH analysis

For LOH analysis, genomic DNA was amplified by PCR using the following polymorphic markers: *D19Mit59*, *D19Mit46*, *D19Mit19*, *D19Mit53* and *D19Mit34* (see Supplementary Table 1). To determine LOH at the *Pten* locus, the microsatellite sequence was searched using the UCSC Genome Bioinformatics database (http://genome.ucsc.edu/). A repetitive region within intron 2, which contained fragment length polymorphism between C57BL/6 and C3H/HeJ mice, was identified. PCR products amplified from genomic DNA of C57BL/6 and C3H/HeJ mice that contained this region were sequenced, and the polymorphism was confirmed. The primer sequences and the conditions for each PCR reaction are described in Supplementary Table 1. PCR products were resolved using a capillary electrophoresis system HAD-GT12 Genetic Analyzer (eGene Inc., Irvine, CA, USA) or by 3% NuSieve agarose (3:1) gel electrophoresis (FMC, Rockland, MA, USA).

2.3. RT-PCR analysis

Total RNA was extracted from tumor tissues using the acid guanidinium thiocyanate–phenol-chloroform method [24], and the cDNA was reverse transcribed using 10 μg total RNA, Moloney murine leukemia virus reverse transcriptase (Toyobo Co., Ltd., Osaka, Japan), and random hexamers (Takara Bio) according to the manufacturer's recommendations. The primer sequences and the conditions for each PCR reaction are described in Supplementary Table 1. PCR products were resolved by 2% agarose gel electrophoresis and analyzed using a Luminescent image analyzer LAS-3000 (Fujifilm, Tokyo, Japan). PCR products were directly sequenced using a Big Dye Terminator v3.1 (Applied Biosystems, Foster City, CA, USA) and an ABI PRISM 3100 Genetic Analyzer (Applied Biosystems) or sequenced after TA cloning using a TOPO TA cloning kit (Invitrogen Co., Carlsbad, CA, USA).

2.4. Bisulfite sequencing analysis

Genomic DNA (1.0 μg) was subjected to bisulfite modification using a CpGenome DNA modification kit, No. S7820 (Chemicon, Temecula, CA, USA), according to the manufacturer's instructions. Bisulfite-modified DNA (40 ng/μl) was then subjected to PCR amplification using primers specific for methylated CpG cytosines as described in Supplementary Table 1. PCR products were sequenced after TA cloning.

2.5. Western blot analysis

Thymic lymphoma cells and normal thymocytes were dissolved in cell lysis buffer (Cell Signaling Technology Inc., Danvers, MA, USA) containing phenylmethanesulfonyl fluoride. Proteins were denatured by heating at 100 °C for 5 min in sample buffer containing SDS, and then lysates (20 μg) were separated by 10% SDS-PAGE and transferred to a PVDF membrane (Millipore Co., Billerica, MA, USA). Anti-Pten, anti-Akt, anti-phospho-Akt (Ser473) and anti-beta-actin (Santa Cruz Biotechnology Inc., Santa Cruz, CA, USA) were used as primary antibodies. Horseradish peroxidase-conjugated anti-rabbit (Cell Signaling Technology) or anti-goat (Santa Cruz Biotechnology) IgG was used as secondary antibody. Signals were developed using ECL plus Western Blotting Detection Reagents (GE Healthcare, Little Chalfont, Buckinghamshire, UK) and analyzed using the LAS-3000 luminescent image analyzer (Fujifilm).

2.6. Array-CGH analysis

We designed and used an Agilent 8 × 15k-formatted mouse custom array-CGH microarray (#020410; Agilent Technologies, Santa Clara, CA, USA), which consisted of about 15,000 oligonucleotide probes, including 1499 for the genomic region covering the *Pten* locus on chromosome 19 (about 430 kbp). Fluorescence labeling of DNA, microarray hybridization and post-hybridization washing were carried out according to the manufacturer's protocol (version 5) for genomic DNA analysis using oligonucleotide array-CGH. Scanning was performed using an Agilent microarray scanner (G2565BA). Signal intensities were measured and evaluated using Agilent

Feature Extraction software v9-5-35 and CGH analytics software v3-5-14, respectively. The microarray data reported in this article have been deposited in the Gene Expression Ominibus (GEO) database, www.ncbi.nlm.nih.gov/geo (accession no. GSE17751).

2.7. Quantitative real-time RT-PCR

Quantitative real-time RT-PCR analysis of miR-19a and miR-21 was performed using a TaqMan MicroRNA assay kit (Applied Biosystems) according to manufacturer's recommendations. Quantitative PCR amplification of cDNAs was performed using a Mx3000P real-time PCR system (Stratagene, La Jolla, CA, USA) and TaqMan Universal PCR Master Mix (Applied Biosystems). Data were normalized to the levels of the small nucleolar RNAs 202 and 234. Each reaction was performed in triplicate. Data were analyzed with MxPro software, version 4.10 (Stratagene).

3. Results

3.1. LOH analysis

Mouse *Pten* encodes a protein product predicted to have 403 amino acid residues and is located at 24.5 cM on chromosome 19. We analyzed LOH using five independent microsatellite simple-sequence-length polymorphism makers on chromosome 19. LOH at the *Pten* locus was also examined using a microsatellite marker within intron 2, which distinguished the polymorphism between C57BL/6 and C3H/HeJ mice. LOH around the *Pten* locus was identified in seven lymphomas (TL8, 11, 12, 14, 19, 20 and 21) (Fig. 1). The LOH frequency (30%; 7 of 23 lymphomas) was roughly consistent with previous studies examining radiation-induced thymic lymphomas in various F1 hybrid mouse strains [22,23,25].

3.2. Expression of Pten mRNA and protein

We examined the expression of *Pten* transcripts by reverse transcriptase (RT)-PCR analysis using three sets of primers (Fig. 2). Altered expression of *Pten* mRNA was observed in 5 of 23 lymphomas (TL5, 8, 11, 14 and 20). TL5 had an additional but faint PCR product that was larger than the predicted product when amplified using primers Ex2F and Ex7R. For TL8, RT-PCR products generated using any primer combination were faint or undetectable. LOH at the *Pten* locus in TL8 indicated that one *Pten* allele remained. Thus the absence of *Pten* transcripts suggested the transcriptional silencing at the *Pten* promoter region in the remaining allele. Using the Ex1F-7R primer pair, TL11 generated one RT-PCR product of the predicted length and three longer RT-PCR products; TL14 generated a faint, short RT-PCR product in addition to a product of the predicted length. TL20 generated a faint product when primers Ex1F and Ex7R were used, but a substantial amount of product was generated using either of the remaining two sets of primers, suggesting that a 5′ portion of exon 1 was missing.

3.3. Sequencing of bisulfite-modified DNA

Because our data for TL8 implicated transcriptional silencing of *Pten* (Figs. 1 and 2), we analyzed the DNA methylation pat-

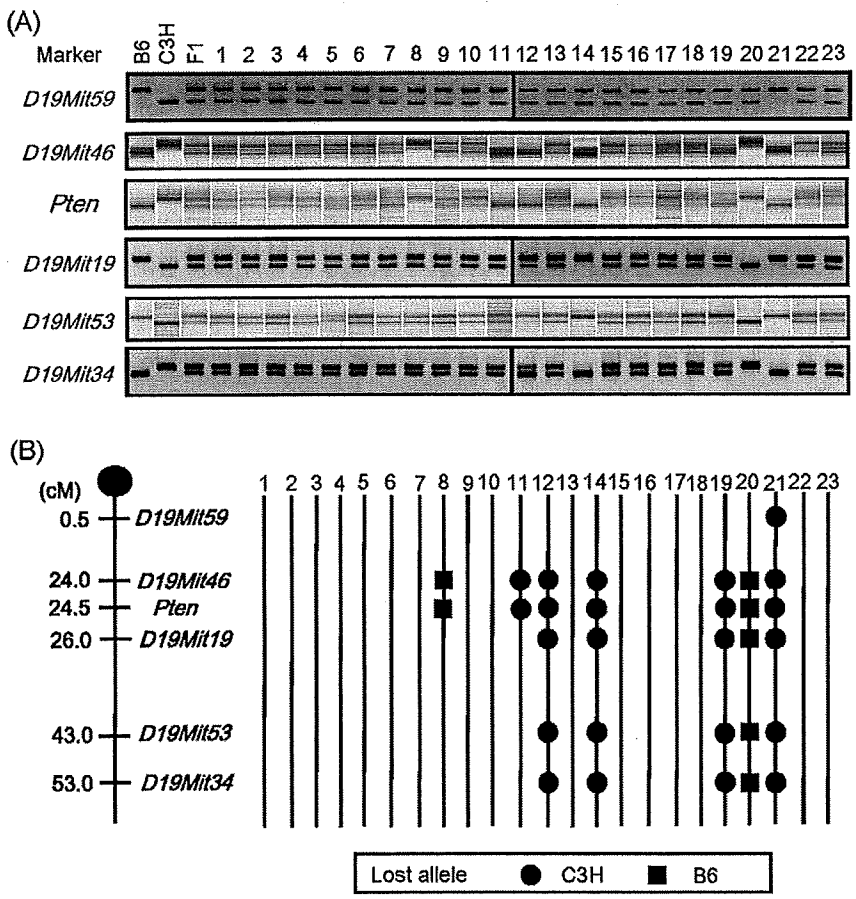

Fig. 1. LOH analysis of chromosome 19 in 23 radiation-induced thymic lymphomas. (A) The first three lanes represent control DNA samples from the maternal C57BL/6 strain (B6), the paternal C3H/HeJ strain (C3H), and the B6C3F1 hybrid (F1), respectively. Numbers above the remaining lanes reflect the tumor identification numbers. PCR amplification of genomic DNA was performed using the indicated polymorphic marker primer pair followed by electrophoretic analysis of amplification products. (B) Schematic diagram of LOH on chromosome 19 in each lymphoma. Lymphoma identification numbers are indicated at top. Polymorphic markers are shown to the right of the chromosome schematic, and marker positions indicating distances (cM) from the centromere are shown at left. The *Pten* marker is located between exons 2 and 3. Absence of a circle or square indicates the retention of heterozygosity.

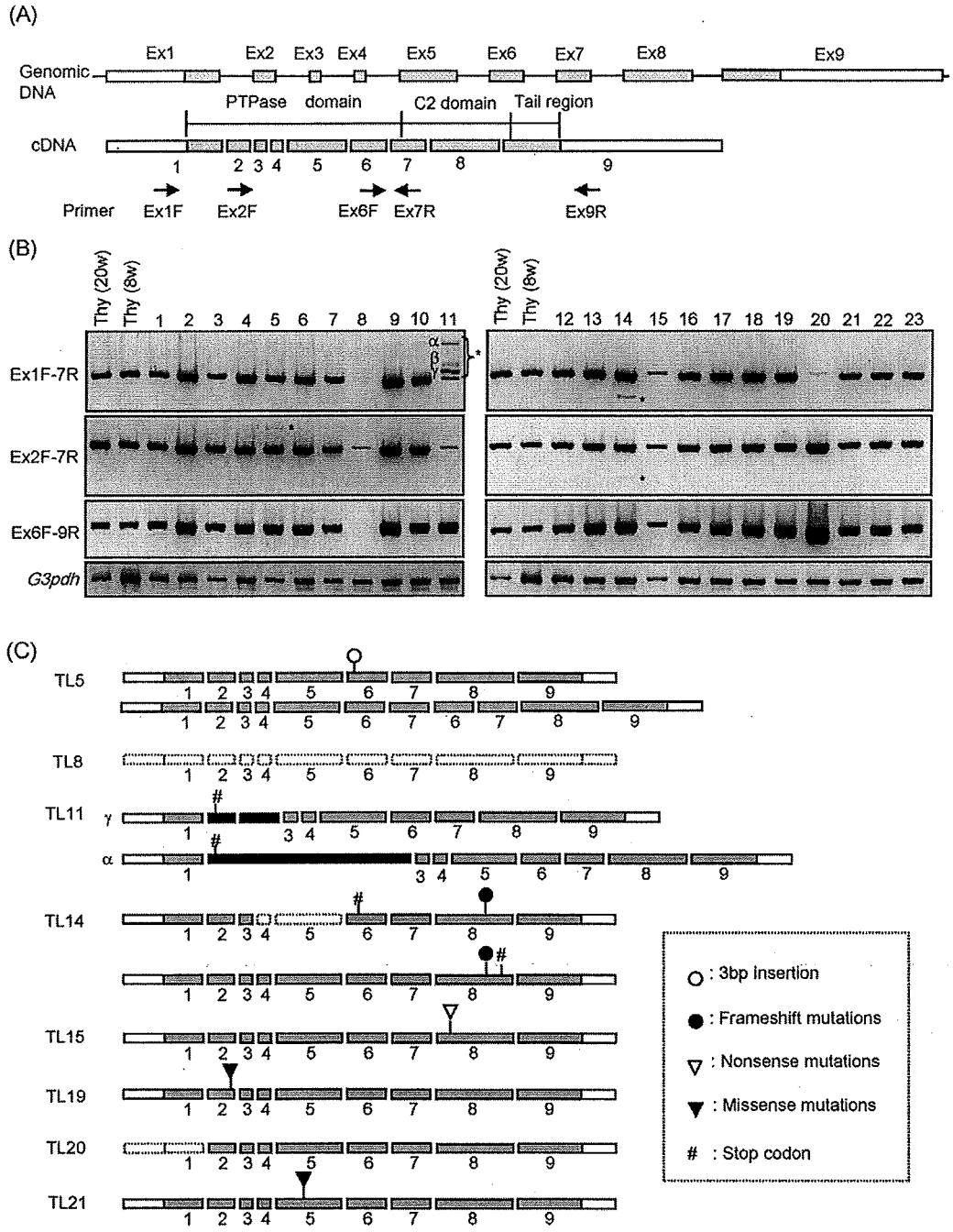

Fig. 2. Alteration of *Pten* in radiation-induced thymic lymphomas. (A) Schematic representation of *Pten*. Shading indicates the *Pten* coding regions. RT-PCR amplification primers and their annealing locations are indicated by arrows. Ex, exon; PTPase, phosphotyrosine protein phosphatase. (B) RT-PCR analysis of *Pten* using the primer pairs indicated at left and genomic DNA isolated from thymocytes (Thy) of 20- or 8-week-old mice (control lanes 1 and 2, respectively), or from the lymphoma indicated at top, was performed and reaction products were subjected to 2% agarose gel electrophoresis. *G3pdh* was used as a control for RT-PCR amplification and as a loading control. Asterisks indicate RT-PCR products longer or shorter than the expected size. (C) Schematic representation of aberrant *Pten* transcripts. Black bars in TL11 indicate inserted intronic sequences. #, Positions of newly generated in-frame stop codons that possibly cause immature translation.

terns in the 5′ (upstream) region of *Pten* (Supplementary Fig. 1A, indicated as P1) in all lymphomas; this region corresponds to the promoter of human *PTEN* that was shown to be aberrantly methylated in T-cell ALLs [26]. However, no aberrant methylation was detected (Supplementary Fig. 1B). Although the CpG sites at positions -39 and -40 relative to the transcriptional start site were both methylated in most of the lymphomas, these CpG sites were also frequently methylated in normal thymocytes. For TL8, we analyzed an additional *Pten* region (Supplementary Fig. 1A, indicated as P2) in which hypermethylation has been suggested to be associated with a lack of *PTEN* expression in non-small cell lung cancer [27]. However, we again did not detect aberrant methylation (Supplementary Fig. 1C). Thus, the absence of *Pten* transcripts likely resulted from a mechanism other than silencing by DNA methylation.

3.4. Sequence analysis of *Pten* transcripts

Next, we determined the sequence of the RT-PCR products from all lymphomas including those products that were longer or shorter than predicted in TL5, 11 and 14 (Fig. 2C). In TL5, the longer

PCR product generated using the Ex2F-7R primer pair contained duplicated exons 6 and 7. The product of predicted length had an insertion of 3 bp, which encoded an amino acid change from Ser170 to Met and Cys. Because the normal *Pten* sequence was not observed, TL5 may contain biallelic *Pten* mutations. TL11 had one RT-PCR product of predicted length and three longer products when amplified using the Ex1F-7R primer pair. The most predominant longer fragment generated from TL11 (Fig. 2B, indicated as γ) contained two large nucleotide insertions of 88 and 122 bp within intron 1 that generated a stop codon, together with a deletion of exon 2. The least predominant longer fragment generated from TL11 (Fig. 2B, indicated as α) contained a 1078-bp insertion in intron 1 and a deletion of exon 2, generating a stop codon. Sequencing of the TL11 PCR product β could not be achieved. The faint product of predicted length had no mutations but may have been derived from contamination of the tumor sample with healthy cells. In TL14, the predominant product of predicted length generated using the Ex6F-9R primer pair had a frameshift mutation due to a 1-bp insertion (962insA) in the poly(A)6 stretch in exon 8, creating a downstream stop codon. The faint/short PCR product of TL14, generated using either Ex1F-7R or Ex2F-7R primer pairs, had both the frameshift mutation and loss of exons 4 and 5, the latter of which generated a stop codon at residue 264. TL15 had an allele with a *Pten* nonsense mutation owing to a substitution (base 862G to T) in addition to a wild-type allele. TL19 had a *Pten* missense mutation owing to a substitution (base 158T to C), resulting in V53A. TL19 generated both mutated and wild-type PCR products, coincident with incomplete allelic LOH in TL19 (Fig. 1A). TL20 expressed only a single *Pten* transcript that lacked exon 1. TL21 had a missense mutation (base 364A to T) in *Pten*, resulting in I122F.

Overall, *Pten* transcripts in radiation-induced lymphomas had a variety of genetic lesions including missense mutations (TL19 and 21), nonsense mutations (TL15), 1- and 3-base insertions (TL14 and 5, respectively), partial intron insertions (TL11), exon duplication (TL5), exon deletion (TL11, 14, 20), and null expression (TL8). Although these mutations are very complicated, they were in good agreement with previous reports demonstrating that missense mutations occur in the Pten phosphatase domain whereas nonsense and frameshift mutations, resulting in protein truncation, occur in the C2 domain of Pten. In total, seven of eight lymphomas carrying a *Pten* mutation (88%) contained biallelic alterations.

3.5. Array-based comparative genomic hybridization (array-CGH) analysis of genomic DNA from TL8, 11 and 20

In order to know the reason for the absence or aberrant transcription of *Pten* in TL8, 11 and 20 (Fig. 2), we analyzed genomic structures around the *Pten* locus in TL8, 11 and 20 using array-CGH, which was designed for intensive analysis of the *Pten* locus. As shown in Fig. 3, the array-CGH profiles suggested partial homozygous deletions (<30 kbp) in the *Pten* locus in these lymphomas, which were positioned within the regions of hemizygous deletion in TL8 and 11. A region of homozygous deletion (~24 kbp) in TL8 occurred in the 5′ (upstream) region of *Pten*, encompassing both the putative promoter region and the transcription initiation site; this may account for the *Pten* silencing observed in TL8 (Fig. 2). Two RT-PCR products from TL11 had a deletion of exon 2 together with one or two insertion(s) of a partial sequence of intron 1 (Supplementary Fig. 2). Array-CGH indicated that TL11 had a homozygous deletion that extended 1 kbp downstream of exon 1 into the 5′ flanking region of exon 2, suggesting that the abnormal transcripts were generated by aberrant splice-site selection likely resulting from the absence of correct splice sites at the intron 1/exon 2 boundary. Array-CGH analysis of TL20 also revealed homozygous deletion of genomic regions (~4 kbp), including both the putative promoter region and the *Pten* transcription initiation site. RT-PCR analysis

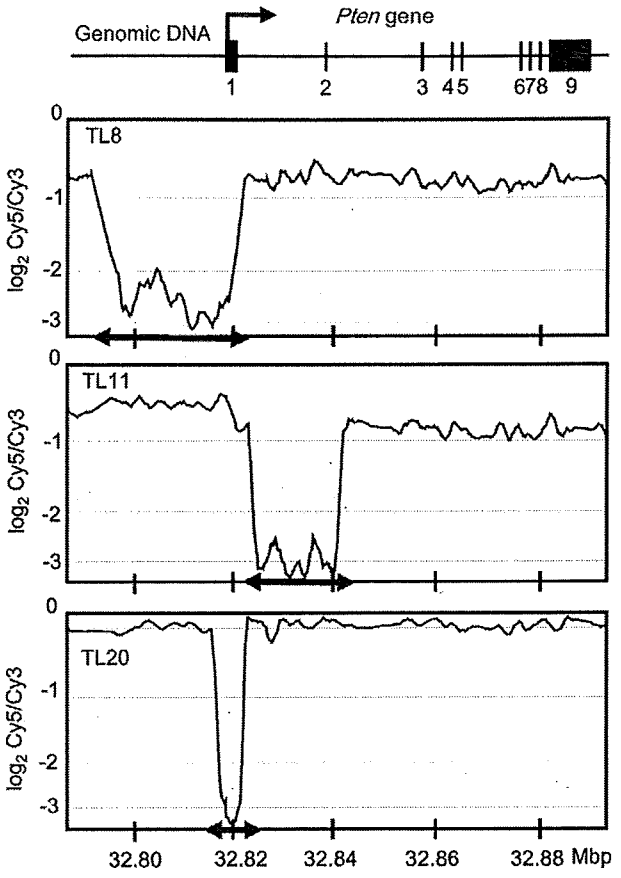

Fig. 3. Array-CGH analysis reveals homozygous focal deletions in three radiation-induced thymic lymphomas (TL8, 11 and 20). Schematic of *Pten* (top), in which shaded bars represent exons aligned to the genomic positions on chromosome 19 indicated along the *x*-axis in the array-CGH profiles below the schematic. Moving averages of the normalized $\log_2$ Cy5/Cy3 ratio, calculated based on 10 data points, are plotted in the array-CGH profiles. Arrows on the *x*-axis correspond to genomic regions amplified by PCR and sequenced to identify breakpoints.

indicated the presence of a *Pten* transcript, however, suggesting that the ectopic transcription was initiated at a cryptic promoter in TL20.

To explore the mechanism responsible for these deletions, genomic regions containing the breakpoints (indicated by the *x*-axis arrows in Fig. 3) were amplified by PCR and sequenced (Fig. 4), which identified nucleotide insertions at the breakpoint junctions in TL8 and 11. In TL11, a pair of recombination signal sequence-like sequences, composed of heptamer- and nonamer-like motifs separated by non-conserved spacers of 12 or 23 bp, were located between but immediately adjacent to the breakpoints, suggesting that illegitimate V(D)J recombination gave rise to the deletion. In contrast, a 0.8-kb templated nucleotide sequence was inserted in the TL20 deletion region. Overlaps of 1 or 2 nucleotides at both breakpoints of the two junctions were identified, suggesting that microhomology-mediated rearrangements might have led to the deletion.

3.6. Loss of Pten and downstream activation of Akt

Pten protein expression varied in the lymphomas we examined (Fig. 5). To determine whether decreased expression correlated with loss of Pten function, we analyzed the degree of Ser473 phosphorylation in Akt (Fig. 5). Pten was not expressed in lymphomas TL8, 11, 14 and 20, consistent with the aberrant stop codon and genomic deletions in the *Pten* promoter region. Pten expression level correlated inversely with phosphorylation of Akt. Lymphomas

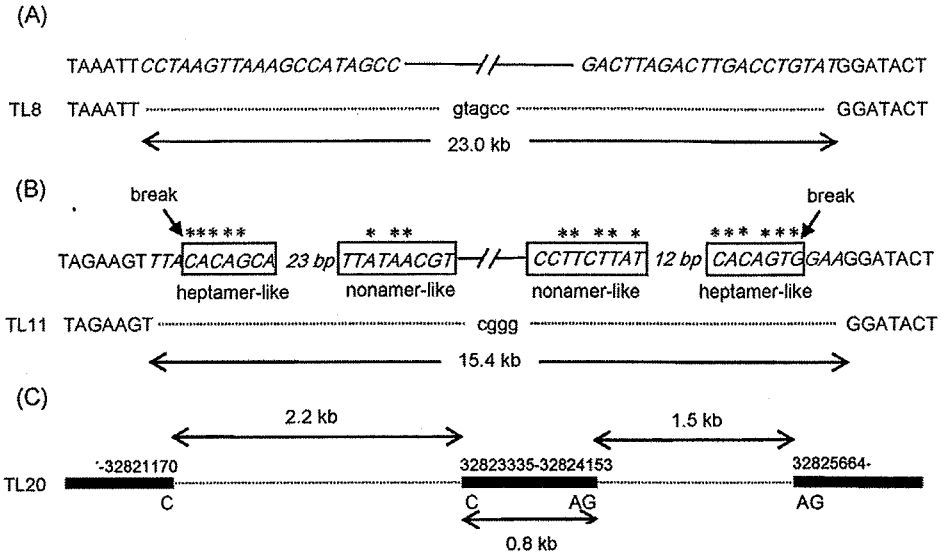

Fig. 4. Sequence analysis of *Pten* focal deletion breakpoints. Schematic diagram representing sequence analysis of focal deletion breakpoints in *Pten* in TL8 (A), TL11 (B) and TL20 (C) lymphomas. Dotted lines indicate deleted genomic regions, and the deleted wild-type sequence is shown in italics in (A and B). Boxes indicate heptamer-like or nonamer-like motifs, and nucleotides marked with asterisks are identical to the canonical heptamer or nonamer sequences. Arrows indicate the sites of Rag-mediated DNA breakage. Lowercase letters indicate nucleotide insertions. Black bars indicate the positions (mm8 assembly) of the retained DNA ends. The uppercase letters below the line in (C) indicate the sequences used to locate microhomology at both ends of each deletion.

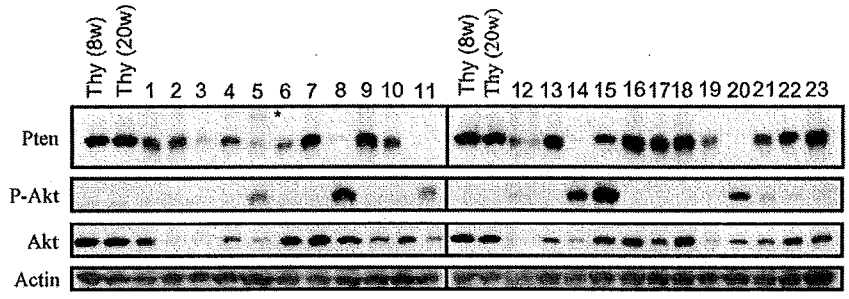

Fig. 5. Analysis of Pten expression and Akt phosphorylation in radiation-induced thymic lymphomas. Western blot analysis of lysates from 23 radiation-induced T-cell lymphomas was performed using antibodies to the proteins indicated at left. Lanes 1 and 2, control thymocytes (Thy) of 8- and 20-week-old mice, respectively. Actin was analyzed as loading control. Asterisk indicates polypeptide with larger mass than expected.

TL5, 19 and 21 expressed mutant Pten containing amino acid substitutions in the phosphatase domain. In TL5 and 21, Akt phosphorylation was weakly observed, suggesting dysfunction of Pten. In TL19, Akt phosphorylation was not observed. The lack of phosphorylation of Akt might have resulted from decreased levels of Akt protein. In TL3 and 12, in which the Pten expression level was low, Akt phosphorylation was not observed owing to negligible expression of Akt protein. In contrast, Akt was highly phosphorylated in TL15, which expressed wild-type Pten. It is possible that other factors that regulate the phosphorylation of Akt, such as PI3K and Ship, may activate Akt independently of Pten [28,29].

3.7. *Levels of Pten-targeting microRNAs do not correlate with Pten protein levels in lymphomas*

Lymphomas TL3, 6 and 12 contained low levels of Pten protein, although neither a decrease in the amounts of RT-PCR product nor genetic aberrations were observed. Recent reports have revealed the involvement of microRNAs in downregulation of Pten expression [13,14,30–33]. We therefore examined whether certain microRNAs contributed to post-transcriptional downregulation of the expression of Pten. For all lymphomas with sufficient available RNA, expression levels of the well-studied *Pten*-targeting microRNAs miR-19a and miR-21 were analyzed using real-time RT-PCR (Supplementary Fig. 3). Expression of miR-19a was almost constant among all lymphomas and was similar to the expression levels in normal thymocytes from 20-week-old mice. Some lymphomas exhibited relatively high miR-21 expression, but there was no significant inverse correlation between miR-21 expression level and Pten protein level.

4. Discussion

In our present study of radiation-induced thymic lymphomas, *Pten* inactivation occurred with a frequency of 30% (7 of 23 lymphomas) and was caused by a variety of biallelic structural abnormalities including base substitutions, long and short (1 and 3 bp) insertions, duplication, and deletions (Table 1). Epigenetic silencing was not observed.

Frequent LOH around the *PTEN* locus has been reported in human leukemias (20% frequency) and lymphomas (23% frequency) [34,35]. *PTEN* alterations, however, have only been sporadically detected in hematopoietic neoplasms [36–42]. In contrast, aberrant DNA methylation in the *PTEN* promoter region has been detected at a frequency of 20% in T-cell ALLs and at a frequency of 18% in B-cell ALLs [26], suggesting that epigenetic silencing is a dominant mechanism of *PTEN* inactivation in hematopoietic neoplasms. Santos et al. [22] has suggested that

Pten in radiation-induced mouse thymic lymphomas also undergoes epigenetic silencing in C57BL/6J and BALB/c F1 hybrid mice. In our present study, however, no aberrant DNA methylation was detected in CpG islands located in the 5′ (upstream) region of *Pten*. Thus, epigenetic silencing does not play a major role in *Pten* loss in radiation-induced thymic lymphomas in B6C3F1 mice. Mao et al. [61] also reported that *Pten* does not undergo methylation-mediated transcriptional silencing in $p53^{+/-}$and $p53^{-/-}$mice. The reason for these discrepancies is not clear, although different mice strains may exhibit distinct patterns of CpG island methylation. Indeed, it has been reported that patterns of CpG island methylation in T-cell lymphomas are driven by the genetic configuration of tumor cells [43]. Santos et al. [22] reported that intragenic mutations do not occur in any genes on chromosome 19, including *Pten*, in C57BL/6J and BALB/c F1 hybrid mice. In contrast, our data indicated frequent intragenic mutations of *Pten* in C57BL/6J and C3H/HeJ F1 hybrid mice, as was suggestive of the influence of the host genetic background on the mutation spectrum. We previously demonstrated strain-based differences between C57BL/6 and C3H mice with respect to genomic alterations of *Kras* and genome-wide copy numbers in thymic lymphomas [44,45].

We have shown that LOH of *Pten* occurred at a frequency of about 30% (7 of 23 lymphomas) and that 8 lymphomas contained complex *Pten* structural abnormalities. Most of the available information regarding the mutation spectrum of *PTEN* has been derived mainly from epithelial cancers such as endometrial carcinomas, glioblastomas, prostate carcinomas, and others [6,7]. In these cases missense, nonsense and frameshift mutations predominate; missense mutations have been found to cluster in exons 5 and 6, encoding the PTEN phosphatase domain, and nonsense and frameshift mutations cluster in the poly $(A)_6$ stretches and in exons 7 and 8, encoding the C2 domain [6]. Comparison of mutations has shown that *PTEN* mutations in tumors depend on tissue type. Endometrial carcinomas predominantly contain frameshift mutations (>60% frequency), whereas glioblastomas contain fewer frameshift mutations but more missense mutations (61% frequency). Recently, *PTEN* frameshift mutations were found to cluster exclusively in exon 7 in human T-cell ALLs [29]. On the other hand, hemizygous *PTEN* deletions have been reported in 39% of prostatic adenocarcinomas, and homozygous *PTEN* deletion has been observed in 5% of prostate tumors [46]. Of note, hemizygous *PTEN* deletion is usually accompanied by an interstitial microdeletion.

In this study, we detected focal homozygous deletions at *Pten* in 13% of lymphomas (3/23; TL8, 11 and 20), all of which resulted in dearth of Pten protein. In two lymphomas (TL8 and 11), homozygous deletions were positioned within hemizygous deletions, possibly resulting from independent deletions occurring in both alleles in these lymphomas. On the other hand, TL20 had a homozygous deletion without adjacent hemizygous deletions. The observation that TL20 contained distally extending continuous LOH on chromosome 19 suggests that this homozygous deletion might have been caused by sequential events, in which one small deletion within the *Pten* locus was followed by subsequent mitotic recombination. Notably, microdeletions have been reported in several key tumor-related genes, such as *IKZF1*, *PAX5* and *CDKN2A*, in human ALLs [47–51] and in *Nocth1* and *Bcl11b* in murine thymic lymphomas [52,53]. The sequence at the deletion breakpoint strongly suggests that these deletions were generated by illegitimate V(D)J recombination and microhomology-mediated rearrangement [49,52–54]. Homozygous *PTEN* deletions were recently identified in 4% of human primary childhood T-cell ALLs [29].

A previous study on T-cell lymphomas developed in chromosomally unstable *Terc*-, *Atm*- and *Trp53*-null mice also reported frequent homozygous deletions at the *Pten* locus [39]. In addition, recent reports on breast cancers [55] and medulloblastomas [56] demonstrated that *Pten* was selectively targeted in a background of defective homologous recombination repair of DNA double-strand breaks (DSBs). Importantly, radiation is a genotoxic stress that increases the frequency of DSBs and stimulates V(D)J rearrangements at cryptic recombination signal sequences [57]. Therefore, radiation-induced DNA DSBs may facilitate hemi- and homozygous deletions of the *Pten* locus.

The two-hit model posits that both copies of a tumor suppressor gene must be inactivated before cancer can develop [58]. Some tumor suppressor genes, however, are inactivated solely by hemizygous loss, indicating the involvement of haploinsufficiency. Several lines of evidence suggest that *Pten* is such a haploinsufficient tumor suppressor gene, at least in epithelial cancers such as prostate cancer [59,60]. A study of radiation-induced lymphomas from *Trp53* heterozygous and/or null mice suggested that *Pten* is a haploinsufficient tumor suppressor gene and that *Pten* haploinsufficiency is a common characteristic of radiation-induced lymphoma development [61]. It is unclear whether *Pten* is inactivated in a bi- or mono-allelic manner in thymic lymphomas of wild-type mice. In the present study, biallelic *Pten* inactivation occurred in seven of eight lymphomas. Although complete loss of *Pten* resulted in increased phosphorylation of Akt, lymphomas with decreased *Pten* expression or monoallelic inactivation (TL15) did not always exhibit increased Akt phosphorylation. Together with the frequent biallelic point mutations and microdeletions observed at the *Pten* locus, these findings suggest that decreased *Pten* dosage alone is not sufficient for activation of the PI3K/Akt signaling pathway during radiation-induced lymphomagenesis. Manifestation of *Pten* haploinsufficiency may require other genetic or epigenetic aberrations. It has been suggested that in tumors of defined tissue origin and genetic background, *Pten* functions in a haploinsufficient manner, but that for others both alleles need to be inactivated [62].

Conflicts of interest

The authors declare that there are no conflicts of interest.

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Appendix A. Supplementary data

Supplementary data associated with this article can be found, in the online version, at doi:10.1016/j.mrfmmm.2009.12.011.

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Review

Recent Advances in the Biology of Heavy-Ion Cancer Therapy#

Nobuyuki HAMADA[1†*], Tatsuhiko IMAOKA[2†], Shin-ichiro MASUNAGA[3], Toshiyuki OGATA[4], Ryuichi OKAYASU[5], Akihisa TAKAHASHI[6], Takamitsu KATO[5], Yasuhiko KOBAYASHI[7], Takeo OHNISHI[6], Koji ONO[3], Yoshiya SHIMADA[2] and Teruki TESHIMA[4]

Heavy ions/DNA double-strand break repair/Intratumor quiescent cell population/p53/Bcl-2/Metastasis and angiogenesis /Carcinogenesis.

Superb biological effectiveness and dose conformity represent a rationale for heavy-ion therapy, which has thus far achieved good cancer controllability while sparing critical normal organs. Immediately after irradiation, heavy ions produce dense ionization along their trajectories, cause irreparable clustered DNA damage, and alter cellular ultrastructure. These ions, as a consequence, inactivate cells more effectively with less cell-cycle and oxygen dependence than conventional photons. The modes of heavy ion–induced cell death/inactivation include apoptosis, necrosis, autophagy, premature senescence, accelerated differentiation, delayed reproductive cell death of progeny cells, and bystander cell death. This paper briefly reviews the current knowledge of the biological aspects of heavy-ion therapy, with emphasis on the authors' recent findings. The topics include (*i*) repair mechanisms of heavy ion–induced DNA damage, (*ii*) superior effects of heavy ions on radioresistant tumors/cells (intratumor quiescent cell population, *TP53*-mutated and *BCL2*-overexpressing tumors), (*iii*) novel capacity of heavy ions in suppressing cancer metastasis and neoangiogenesis, and (*iv*) potential of heavy ions to induce secondary (especially breast) cancer.

INTRODUCTION

Energetic heavy ions are defined as charged particles heavier than helium ions, and they generally have high relative biological effectiveness (RBE).[1-4] Unlike conventional photons such as X- and γ-rays, heavy ions form a sharp Bragg peak (a pronounced rise in energy deposition of radiation during its travel through matter), with a steep dose falloff downstream. Because of the primarily narrow Bragg peak, spread-out Bragg peaks (SOBP) have been devised to obtain broad and uniform dose distribution,[5] thereby enabling dose escalation to the target tumor volume without much exacerbation of normal tissue complications. Such excellent biological properties and dose conformity represent a rationale for heavy-ion cancer therapy. Ever since the first clinical experience in 1977,[6] the number of treated patients has been growing steadily and has already exceeded 6,000. So far, heavy-ion therapy has achieved good cancer controllability in short treatment times while sparing critical normal organs.[7-9] A number of new facilities are becoming operational worldwide in addition to the currently available ones, leading to a wider popularization of heavy-ion therapy.

It is well established that biological effectiveness of ionizing radiation varies with its linear energy transfer (LET), namely, the rate of energy loss along the trajectory of an ionizing particle (usually expressed in keV/μm).[10] High-LET

*Corresponding author: Phone: +81-43-206-4631,
Fax: +81-43-251-4531,
E-mail: hamada-n@nirs.go.jp

[1]Space Radiation Research Unit, International Open Laboratory, National Institute of Radiological Sciences, 4-9-1 Anagawa, Inage, Chiba 263-8555; [2]Experimental Radiobiology for Children's Health Research Group, Research Center for Radiation Protection, National Institute of Radiological Sciences, 4-9-1 Anagawa, Inage, Chiba 263-8555; [3]Particle Radiation Oncology Research Center, Research Reactor Institute, Kyoto University, 2-1010 Asashiro-nishi, Kumatori, Osaka 590-0494; [4]Department of Radiation Oncology, Osaka University Graduate School of Medicine, 2-2 Yamadaoka, Suita, Osaka 565-0871; [5]Heavy-ion Radiobiology Research Group, Research Center for Charged Particle Therapy, National Institute of Radiological Sciences, 4-9-1 Anagawa, Inage, Chiba 263-8555; [6]Department of Biology, School of Medicine, Nara Medical University, 840 Shijo-cho, Kashihara, Nara 634-8521; [7]Microbeam Radiation Biology Group, Japan Atomic Energy Agency, 1233 Watanuki-machi, Takasaki, Gunma 370-1292, Japan.

†These authors contributed equally to this work.

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heavy ions produce dense ionization along their trajectories, and cause complex and irreparable clustered DNA damage.[11,12] Heavy ions are generally more genotoxic and cytotoxic to irradiated cells than low-LET photons.[1–4] The biological effectiveness depends not merely on LET but also on ion species (or ion track structure), such that the RBE of carbon and neon ions for the clonogenic survival peaks at LET of ~100 and ~200 keV/μm, respectively.[13–15] Changes in cellular ultrastructure at the electron-microscopic level (e.g., irregular protrusions and invaginations of plasma membrane, distended sarcoplasmic reticula, and increased autophagic vacuoles) occur as early as a few minutes after heavy-ion exposure, and autophagy might be involved in removal of such disruption.[16–18] The mode of heavy ion–induced cell death/inactivation includes apoptosis, necrosis, autophagy, premature senescence, accelerated differentiation, delayed reproductive death in the descendants of irradiated cells, and bystander cell death.[1–4,19–32] Heavy ions are effective at killing cells with little cell-cycle and oxygen dependence of radiosensitivity,[1–4] and possess high potential to suppress angiogenesis, metastasis and arrhythmia.[33–35] Moreover, heavy ions may overcome tumor radioresistance caused by mutation of the tumor suppressor gene *TP53* (also known as *p53*), overexpression of the oncogene *BCL2* (also *Bcl-2*), and intratumor hypoxia.[36–40] Although heavy-ion therapy has provided favorable clinical outcome with irradiation alone, interest is increasing in combined modalities, especially with molecularly targeted approaches. In comparison with heavy ions alone, the combination with chemical agents (e.g., Bcl-2 inhibitor HA14, anticancer drug docetaxel, and halogenated pyrimidine analogue 5-iodo-2'-deoxyuridine), hyperthermia and gene therapy enhances tumor cell killing.[41–48] Beer, its constituents (β-pseudouridine and glycine betaine), melatonin and α-lipoic acid ameliorate heavy ion–induced damage to normal cells.[49–57] Such approaches may further increase the therapeutic ratio (i.e., ratio of lethal dose to effective dose). On the other hand, the potential of heavy ions to cause adverse effects must not be overlooked. Although clinical efforts have succeeded in reducing acute reactions after treatment, late effects such as secondary cancer induction are gradually becoming a matter of concern. Absolutely no information is currently available on the secondary cancer risk from heavy ions; however, some evidence has accumulated regarding cancer induction in experimental animal models. For example, in a series of studies on mouse Harderian gland tumor, RBE increased with LET, reaching a maximum of 30–45 at 100–200 keV/μm, and did not decrease substantially thereafter up to 650 keV/μm.[58,59] Experimental evidence for cancer induction in other organs is currently being accumulated.

This paper briefly reviews the current knowledge of the biological aspects of heavy-ion therapy, focusing on the recent findings of the authors. Firstly, the basic mechanisms of DNA repair for heavy ion–induced damage are reviewed, which underlie their high biological effectiveness. Secondly, as examples of the superior biological characteristics of heavy ions in preclinical settings, the targeting and overcoming of radioresistant tumor cells are reviewed particularly in the case of intratumor quiescent cell populations and radioresistant cells due to *p53* mutation and *Bcl-2* overexpression. The antimetastatic and antiangiogenic potential of heavy ions is also reviewed as another example of their potential advantage in therapy. Finally, experimental information on the cancer-inducing potential of heavy ions is reviewed especially in regard to breast cancer induction. The companion articles by Okada *et al.* and Minohara *et al.* review the clinical and physical aspects of heavy-ion therapy, respectively.[60,61]

BIOLOGICAL EFFECTS OF HIGH-LET HEAVY-ION RADIATION FROM THE ASPECT OF DNA DOUBLE-STRAND BREAK REPAIR

It is important to explain the biological basis for the successful world-leading carbon-ion therapy at the Heavy-Ion Medical Accelerator in Chiba (HIMAC) of the National Institute of Radiological Sciences (NIRS), Japan. In this section, our focus is on the repair of DNA double-strand breaks (DSBs) induced by low- and high-LET radiation. By demonstrating the inefficient repair of DNA and chromosome damage with high-LET radiation, the crucial basis for its high biological effectiveness can be demonstrated.

Inefficient rejoining of DNA DSBs induced by high-LET heavy-ion irradiation

A substantial number of studies on DNA DSB and its repair in cells exposed to high-LET heavy ions have been reported.[62,63] In general, DSB repair is inhibited as a function of LET (up to 200 keV/μm), and the degree of rejoining correlates with cell survival. If the rejoining is inefficient, a high number of remaining DSBs persist after irradiation, leading to lower cell survival. We have also shown this tendency by experiments performed with constant-field gel electrophoresis (M. Noguchi and R. Okayasu, personal communication).

Inefficient repair of DNA DSBs with high-LET irradiation as measured by γH2AX assay

The commonly used gel-electrophoresis method as discussed above may be convenient and useful, but it usually requires the use of a high radiation dose such as 20 Gy, which is significantly higher than the dose range used for cell survival.[64,65] Thus, many researchers have recently employed another method called γH2AX focus assay, which assumes that there is a one-to-one correlation between one DSB and one γH2AX focus.[66] The sensitivity of this assay is also higher than the gel method.[66,67] An example of data using this assay is shown in Fig. 1 for γH2AX appearance and disappearance kinetics after irradiation of cultured